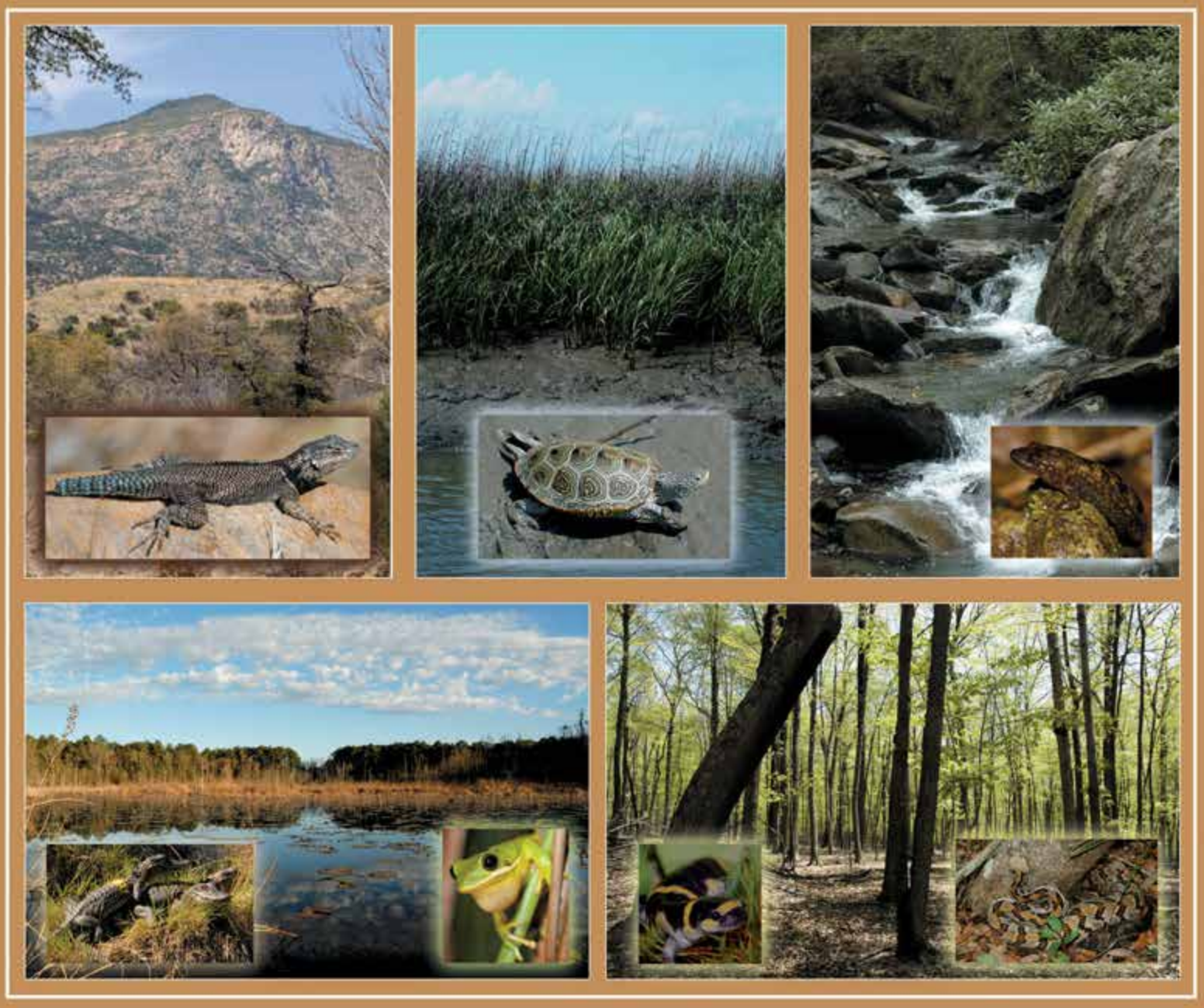
PARTNERS IN AMPHIBIAN AND REPTILE CONSERVATION

INVENTORY AND MONITORING:

RECOMMENDED TECHNIQUES FOR REPTILES AND AMPHIBIANS

With Application to the United States and Canada



PARC Technical Publication IM-1

PARTNERS IN AMPHIBIAN AND REPTILE CONSERVATION

Gabrielle J. Graeter | Kurt A. Buhlmann | Lucas R. Wilkinson | J. Whitfield Gibbons

PARC INVENTORY AND MONITORING INITIATIVE

This inventory and monitoring initiative has been established through partnerships among Partners in Amphibian and Reptile Conservation (PARC), the United States Forest Service (USFS), the University of Georgia's Savannah River Ecology Laboratory (UGA-SREL) and the Department of Defense. The objective of this handbook is to provide land managers and landowners with information about reptiles and amphibians and the qualitative and quantitative techniques that can be used to obtain information on diversity, distribution, and abundance of these animals. It is not intended to establish any specific sampling or monitoring protocols. This handbook is user-friendly, addresses both reptiles and amphibians, and provides specific recommendations on techniques that may be used for each species.

The PARC mission is "to conserve amphibians, reptiles, and their habitats as integral parts of our ecosystem and culture through proactive and coordinated public-private partnerships." PARC is not a funding organization or a policy maker but instead has been created to increase communication among diverse public and private groups and individuals interested in amphibian and reptile conservation. The diversity of participants makes PARC the most comprehensive amphibian *and* reptile conservation effort ever undertaken. To find out more about PARC, please visit our website at: http://www.parcplace.org

OBJECTIVES

The objective of this publication is to provide a user-friendly book to guide land managers, private land owners, and biologists in conducting amphibian and reptile inventory and monitoring programs. We aim to accomplish this not by designing new sampling techniques or protocols, but by providing a comprehensive summary of the techniques available, how to get started, and appropriate references to consult.

For more detail on particular techniques or sampling protocols, please refer to those cited throughout this document and listed in Appendix XI and these key references:

Dodd, C. K., Jr. 2003. Monitoring amphibians in Great Smoky Mountains National Park. Circular 1258, US Geological Survey, Tallahassee, Florida.

Heyer, W. R., M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, M. S. Foster, editors. 1994. Measuring and monitoring biological diversity: Standard methods for amphibians. Smithsonian Institution Press, Washington, DC, USA.

McDiarmid, R., M. Foster, C. Guyer, J. W. Gibbons, and N. Chernoff. 2012 Reptile Biodiversity - Standard Methods for Inventory and Monitoring. University of California Press. Berkeley.

Mitchell, J. C. 2000. Amphibian Monitoring Methods & Field Guide. Smithsonian National Zoological Park, Conservation Research Center, Front Royal, VA. 56 pp.

In addition to the information we have provided and which is available in references cited throughout the document, it is essential to consult and involve your regional herpetologist, particularly during the planning stages of an inventory or monitoring program. They may have critical insight into the timing, location, or study species for the program you are planning.

Suggested citation: Graeter, G. J., K. A. Buhlmann, L. R. Wilkinson, and J. W. Gibbons (Eds.). 2013. *Inventory and Monitoring: Recommended Techniques for Reptiles and Amphibians.* Partners in Amphibian and Reptile Conservation Technical Publication IM-1, Birmingham, Alabama

TABLE OF CONTENTS

Contributing Authors iii

Chapter 1. Introduction 1

A guide for the United States and Canada 1

Who should use this guide 2

How to use this guide 2

Threats to amphibians and reptiles 2

Inventory versus monitoring 11

Inventory and monitoring programs and conservation 13

Chapter 2. Amphibians and Reptiles of the United States 15

Importance of natural history data to inventory and monitoring 15

Nomenclature for native species 16

Exotic herpetofauna of the United States 20

Habitats in the United States 28

Regional Resources on the amphibians and reptiles of the United States 29

Chapter 3. Essentials of Sampling Design 35

Scope 35

Basic concepts 35

Defining project goals 35

Hitting the target 36

Sampling units 37

Inventory 37

Creating species lists 37

Determining species presence/absence 38

Monitoring 38

Site occupancy 38

Relative abundance 39

Population estimation 39

Conclusions 41

Chapter 4. Study Planning and Data Collection 42

Introduction 42

Capture data 43

Climate and seasonality 43

Habitat and microhabitat 46

Locality 47

Maps 47

Photos 48

Geotechnologies 49

Automated data acquisition 50

Dataloggers 51

Environmental Sensors 52

Automated Monitoring of Animals 55

Standards and data management 60

Permits 60

Chapter 5. Standard Techniques for Inventory and Monitoring 61

Introduction 61

Selection of techniques 62

Techniques with a design component 63

Time-constrained searches 63

Area-constrained searches 65

Quadrat sampling 66

Transect surveys 68

Active sampling 71

Hand collecting 71

Visual encounter surveys 73

Dipnetting and sweep samples 75

Stovepipe sampling 76

Kick sampling 77

TABLE OF CONTENTS

Egg mass and nest counts 80
Snorkeling surveys 83
Electroshocking 87
Auditory surveys 89
Basking surveys and basking traps 90
Sign and tracking 92
Road cruising 94
Aerial surveys 99
Easy passive sampling 100
Artificial cover 100
PVC pipe surveys 103
Leaf-litterbag surveys 105
Automated recording systems 107
Intensive passive sampling 109
Aquatic and terrestrial funnel trapping 109
Terrestrial drift fences and pitfall traps 114
Sampling at snake hibernacula 117
Tracking techniques applicable to monitoring 120
Sampling techniques for exotic amphibians and reptiles 123
Seek advice and further training 125
Table 5-1: Species X Techniques Table 127
Chapter 6. Analysis of Inventory and Monitoring Data 203
Introduction 203
Rapid assessment inventory 203
Comprehensive survey inventory 203
Presence/absence monitoring 204
Population status monitoring 204
Estimating species richness 204
Species accumulation rates 204
Quadrat sampling 204
Multiple sampling 205
Coefficients of association 205
Estimating population size 205
Mark-recapture 206
Population estimators 206
Removal sampling 209
Diversity indices 209
Occupancy models 210
Chapter 7. Conclusions and Recommendations .. 212
The importance of inventory data 212
The value of monitoring 213
Modification of techniques 213
Dissemination of results 213
Epilogue 214
Appendix I. Handling Live Amphibians and Reptiles 214
Appendix II. Determination of Age, Sex, and Reproductive Condition 219
Appendix III. Marking Amphibians and Reptiles 229
Appendix IV. Collecting Tissue for Biochemical Analysis 234
Appendix V. Preparing Scientific Specimens 238
Appendix VI. Decontamination of Field Gear 242
Appendix VII. Photo-Vouchering 244
Appendix VIII. Sample Datasheets 277
Appendix IX. Equipment Lists 289
Appendix X. Conservation Status of Amphibians and Reptiles 291
Appendix XI. Sources for Further Information 294
Appendix XII. Manufacturers and Suppliers 299
Literature Cited 301
Acknowledgements 320

CONTRIBUTING AUTHORS

Kimberly M. Andrews – University of Georgia's Savannah River Ecology Laboratory

James B. Bettaso – U.S. Fish and Wildlife Service

Kurt A. Buhlmann – University of Georgia's Savannah River Ecology Laboratory

Erin Clark – U.S. Fish and Wildlife Service

Dean A. Croshaw – University of New Orleans

Michael E. Dorcas – Davidson College

Luke A. Fedewa – U.S. Army

J. Whitfield Gibbons – University of Georgia's Savannah River Ecology Laboratory

Xavier A. Glaudas – University of Nevada

Gabrielle J. Graeter – NC Wildlife Resources Commission

Aaliyah D. Green – University of Georgia's Savannah River Ecology Laboratory

Judith L. Greene – University of Georgia's Savannah River Ecology Laboratory

W. Jeffrey Humphries – NC Wildlife Resources Commission

John B. Jensen – Georgia Department of Natural Resources

Jason L. Jones – Idaho State University

Mary Beth Kolozsvary – Siena College, NY

Thomas M. Luhring – University of Georgia's Savannah River Ecology Laboratory

Peri Mason – Wesleyan University

Walter E. Meshaka, Jr. – State Museum of Pennsylvania

Brian S. Metts – Southeastern Natural Sciences Academy

Tony M. Mills – Low Country Institute

Charles R. Peterson – Idaho State University

Robert N. Reed – USGS Fort Collins Science Center

Brian D. Todd – University of Georgia's Savannah River Ecology Laboratory

Ria N. Tsaliagos – University of Georgia's Savannah River Ecology Laboratory

Tracey D. Tuberville – University of Georgia's Savannah River Ecology Laboratory

Anton D. Tucker – Mote Marine Laboratory, Sarasota, FL

Lucas R. Wilkinson – Savannah River Ecology Laboratory

John D. Willson – University of Georgia's Savannah River Ecology Laboratory

Lawrence A. Wilson — Fernbank Science Center

Christopher T. Winne – University of Georgia's Savannah River Ecology Laboratory

Cameron A. Young – University of Georgia's Savannah River Ecology Laboratory

Robert T. Zappalorti – Herpetological Associates, Inc.

Kurt Buhlmann

CHAPTER 1: INTRODUCTION

Lucas R. Wilkinson and J. Whitfield Gibbons

Biological diversity, the variety of all living things on earth, is under serious threat on a worldwide scale. Habitat loss, invasive species, unsustainable use, and other factors have led to declines in populations and extinctions of species that continue at a rapid rate. As a result, there is great demand for inventory and monitoring programs that can quantify baseline levels of biodiversity and identify at-risk populations so that conservation efforts can be better directed. Amphibians and reptiles, often maligned or ignored, are integral parts of natural ecosystems. Increasing awareness of amphibians and reptiles has led to the recognition that, like other forms of life, herpetofaunal biodiversity is imperiled; the growing list of declining populations suggests a worldwide crisis. Still, the status and plight of most populations remains largely unstudied and unknown. Inventory and Monitoring: Recommended Techniques for Amphibians and Reptiles with Application to the United States and Canada is intended to provide a standardized framework for developing and implementing inventory and monitoring programs in North America. By studying this guide, users will learn to identify inventory or monitoring goals, implement an appropriate sampling regime, and analyze and interpret collected data to estimate amphibian and reptile diversity and abundance.

Jeff Alvarez

Figure 1-1. PARC regions

A GUIDE FOR THE UNITED STATES AND CANADA

This inventory and monitoring guide by PARC, Partners in Amphibian and Reptile Conservation, is designed to provide species- and region-specific standards for inventory and monitoring projects. The guide draws on the expertise of herpetologists working in

each region of the country and focuses on methods and techniques suited to the species and habitats occurring there. The guide addresses techniques and species that are specific to all five regions of the country, as designated by PARC (see Fig. 1-1), but also has application to Canada. The PARC Southeast region includes Alabama, Arkansas, Florida, Georgia, Kentucky, Louisiana, Mississippi, North Carolina, South Carolina, and Tennessee. The Northeast includes Connecticut, Delaware, Maine, Maryland, Massachusetts, New Hampshire, New Jersey, New York, Pennsylvania, Rhode Island, Vermont, Virginia, and West Virginia, and the Midwest region is made up of Illinois, Indiana, Iowa, Kansas, Michigan, Minnesota, Missouri, Nebraska, North Dakota, Ohio, South Dakota, and Wisconsin. The Southwest region is comprised of the states of Arizona, California, Colorado, New Mexico, Nevada, Oklahoma, Texas, and Utah. Lastly, the Northwest region includes Alaska, Hawaii, Idaho, Montana, Oregon, Washington, and Wyoming.

WHO SHOULD USE THIS GUIDE?

This guide is intended to broaden the scale and scope of amphibian and reptile conservation in the United States by allowing new stakeholders to take part in inventory and monitoring. Governmental biologists at all levels, non-governmental biologists, private consultants, non-governmental organizations, private landowners, and concerned citizens, even those without specific training in herpetological research, are encouraged to use this guide to assist in developing new programs that will facilitate amphibian and reptile conservation throughout the United States and throughout the world.

HOW TO USE THIS GUIDE

1. Learn the basics. Chapters 1 and 2 provide basic information about the amphibians and reptiles of the United States, their habitats, and the threats they face that make inventory and monitoring imperative.

2. Establish goals and make a plan. Chapters 3 and 4 will help in identifying the goals and design of a program and assist in planning a study.

3. Discover the techniques. Chapter 5 outlines the standard techniques used for an inventory or monitoring program and provides insight into selection and development of the associated sampling designs.

4. Determine how to analyze data and apply the results. Chapter 6 outlines the methods available for analyzing inventory and monitoring data and provides sources for more detailed information.

5. Make a plan for dissemination of results. Chapter 7 provides general recommendations and a discussion of the dissemination of results.

6. Consult with authorities and experts. Appendices X and XI provide literature sources and contact information for organizations and governing bodies that can help you obtain permits, learn the current legal status of amphibian and reptile species, and receive additional help.

7. Implement a program. Having developed a foundation in amphibian and reptile biology, clear project goals, a well defined study design, and the skills required to execute it, users of this manual can implement a rigorous inventory or monitoring project that will make a significant contribution to amphibian and reptile conservation.

THREATS TO AMPHIBIANS AND REPTILES

Kimberly M. Andrews, Brian D. Todd, and Luke A. Fedewa

Threats are defined here as occurrences that have the potential to negatively affect amphibian and reptile populations. At an organizational meeting in Atlanta, Georgia in 1998, Partners in Amphibian and Reptile Conservation (PARC, www.parcplace.org) outlined six primary threats as contributing factors of global conservation concern to amphibians and reptiles, meaning that they are substantial factors in already noted declines, or they are factors that could affect future status. Although amphibian population declines are widely discussed in the scientific literature (e.g., Blaustein 1994; Dodd 1997; Alford and Richards 1999; Young et al. 2001, Stuart et al. 2004), herpetologists are just beginning to realize the extent of amphibian declines in the United States, and that reptile declines are also occurring (Gibbons et al. 2000; Winne et al. 2007). This section will follow the PARC list of concerns for amphibians and reptiles: habitat loss and fragmentation, environmental contaminants, invasive species, unsustainable use, disease and parasitism, and global climate change. Due to the wide breadth of impacts and the diversity of amphibian and reptile taxa, this section serves as a broad overview of threats to herpetofauna of the United States with

examples of particular effects and species (see Semlitsch 2003 [amphibians] and Gibbons et al. 2000 [reptiles] for a more extensive presentation of threats and references).

Threats can be challenging to identify and confirm, particularly with amphibians and reptiles where many species exhibit covert behaviors (Lovich and Gibbons 1997) that result in a scarce understanding of some fundamentals of organismal biology. This lack of data complicates protection initiatives. While a proactive approach must be used to ultimately conserve some of the more sensitive herpetofaunal species, herpetologists must practice caution in making recommendations or implementing practices for preservation of species that are still lacking information regarding basic ecology. However, if we delay acting until we have perfect knowledge of every issue, anthropogenic alteration may already have caused irreversible damage.

HABITAT LOSS AND FRAGMENTATION

Effects

Habitat loss and fragmentation are often considered the most pervasive and serious threats to amphibians and reptiles (Mittermeier et al. 1992) and are catalysts for many species receiving federal listing (Wilcove et al. 1998). Habitat loss occurs when land-use changes take place, converting intact ecosystems into forms usable by humans, such as agricultural, commercial, or residential developments. Fragmentation is a form of habitat loss in which a portion of intact habitat is divided into smaller patches, disrupting the spatial functioning of local population dynamics (rates of birth, migration, and mortality) for many species. Although habitat loss occurs naturally from ecological disasters (fires, hurricanes, tornadoes), here, we specifically refer to anthropogenic forms of disturbance.

Habitat loss and fragmentation can be detrimental to animal populations both directly and indirectly, depending on the scale of the development and subsequent level of impact. Mortality can occur immediately as habitats are converted and developed, such as with forest clearcuts, residential neighborhoods, and transportation infrastructure. Habitat loss can also occur indirectly when remaining habitats are affected by human presence, as seen with diamondback terrapins (*Malaclemys terrapin*) and incidental mortality resulting from recreational crab trapping in the Kiawah Island waterways, South Carolina (J.W.G., personal communication). Habitat loss can also occur indirectly from secondary effects such as hydrological changes, pollution, changes to microhabitat or vegetation composition, demographic barriers to movements, and

Michael Marchand

Habitat loss through forest clearing is a major threat to herpetofauna

Michael Marchand

Development for new homes is a major cause of habitat loss in many areas of the country

disruption of predator-prey relationships that render a habitat inadequate. For example, in one study, Todd and Rothermel (2006) documented lower growth and survival of recently metamorphosed southern toads (*Bufo terrestris*) following forest clearcutting.

An estimated 2.2 million acres of habitat are converted annually in the United States, most of which are on non-federal forest land (USDA-NRCS 2001). Amphibians and reptiles reside in many habitats that are ecologically sensitive (e.g., wetlands, Gibbs 2000; marine, Gray 1997) or are prime for development (e.g., coastal sandhills, Frost et al. 1986). For example, a diversity of herpetofauna [inhabiting wetlands] (e.g., anurans, Gibbons et al. 2006; salamanders, Semlitsch 1998; crocodilians, Lang 1987; snakes, Glaudas et al. 2007; turtles, Buhlmann and Gibbons 2001) rely on a complex dynamic involving both aquatic and terrestrial components (Gibbons 2003). Between 1986 and 1997, urbanization accounted for 30% of wetland conversion, agriculture 26%, silviculture 23%, and rural development 21% in the United States (Mac et al. 1998). In the period of 1992-1997, 101,000 acres of wetlands per year were converted. Through the “no net loss” pro-

gram 69,000 acres were gained (USDA-NRCS 2001). Unfortunately, this did not include wetland protection so much as wetland construction, which is a less desirable conservation measure for some herpetofauna (Dahl 2000). Longleaf pine (*Pinus palustris*) and wiregrass (*Aristida* spp.) habitats, host to highly endemic and diverse herpetofaunal communities, once covered 28.3 million hectares in the Southeast, but due to habitat loss, only 2% remains (Mac et al. 1998). Additionally, the loss of prairie habitat in the western United States is estimated at up to 99.9% (Samson and Knopf 1994). One third of the species listed as endangered by the Committee on the Endangered Wildlife in Canada are grassland species (WWFC 1988) and the United States also has a large number of grassland species listed (55) or candidates for listing (728; Samson and Knopf 1994).

Habitat loss and fragmentation can also occur when the actual loss of habitat is very small but barrier and edge effects from the developed area permeate into the adjacent unaltered habitat. The creation of barriers and edges which inhibit animal movement reduces the habitat available to animals for the fulfillment of ecological needs. Roads are a prominent example of such a barrier (e.g., Andrews 1990; Reed et al. 1996; Andrews and Gibbons 2005; Andrews et al. 2006). The road system of the United States contains over 6.2 million km of roads comprising 1-2% of the land area; however, ecological impacts can extend up to 100 m from the road, affecting an estimated 20% of the land area (Forman 2000). Further, Rudolph and colleagues (1999) estimated greater than 50% declines of snake populations within 450 m of roads. Therefore, habitat loss from roads can occur directly from construction and indirectly from effects extending into surrounding areas. Likewise, fragmentation of stream habitat often occurs at road-stream crossings and can result in decreased movement of individuals between the upstream and downstream sides of the road crossing (Jackson 2003; Lowe 2003). Secondary forms of habitat loss that increase landscape-level fragmentation will likely have greater cumulative effects than direct sources alone, and can involve time lags before full effects become apparent (Findlay and Bourdages 2000).

Research challenges

Habitat loss and fragmentation are difficult phenomena to study and conclusively pinpoint, particularly if they take place over large ecological scales (spatial, temporal, or varying in diversity and intensity). Although many effects can be measured at the individual level, habitat loss and fragmentation often result in population-level impacts. Habitat loss is considered one of the most widespread ecological impacts, particularly in areas with high population growth. For instance, Florida's population increased by 834 people/day in the 1990's (FSP 2005). Presumably no major group of animals or plants is completely exempt from the threats of habitat loss and fragmentation. Amphibians and reptiles, which constitute an estimated 25% of the world's vertebrates (Mittermeier et al. 1992), are certainly no exception. Further, due to the intricate habitat requirements of these animals, herpetofauna will continue to be directly and indirectly affected by this constant and increasing trend of land conversion.

Potential solutions

Human expansion across the globe is inevitable, followed by the conversion of wildlife habitat. The most effective means of preserving herpetofaunal populations amidst a growing populace's needs is to protect areas containing sensitive habitats, species, or migratory routes (Dodd and Smith 2003). Effective buffer zones around core habitats, such as wetlands, will more sufficiently protect the area that is being used by the animals of concern (Burke and Gibbons 1995; Buhlmann and Gibbons 2001; Semlitsch and Bodie 2003). Herpetologists need to be more involved in design processes for wetland mitigation and other restorative activities, along with management regimes that support the persistence of wildlife populations. Lastly, more biological research is needed to clarify ecological characteristics essential for the development of feasible conservation measures for various habitats.

ENVIRONMENTAL CONTAMINANTS

Effects

Environmental contaminant effects on amphibian and reptile species are as diverse and understudied as they are prolific, being found nationwide in freshwater (e.g., Mason 1996), marine (e.g., Kennish 1996), and terrestrial (e.g., Novotny and Chesters 1981) habitats. Ecotoxicology is a topic generating increasing interest in its applications to amphibians and reptiles (e.g., Sparling et al. 2000). Gates and colleagues (1985) documented an absence of eastern hellbenders (*Cryptobranchus alleganiensis*) in streams containing acidic drainage from mines, industrial effluent, or improperly treated sewage waste. Acidification results in sodium depletion in terrestrial amphibians, an element essential for cellular homeostasis (Wyman 2003). Many contaminants also have sub-lethal effects on amphibian development and metamorphosis. For example, exposure to the heavy metal mercury has been shown to affect the timing of metamorphosis and to cause developmental abnormalities among larvae and metamorphosing animals (Unrine and Jagoe 2004; Unrine et al. 2004;

Unrine et al. 2005), possibly reducing the subsequent survival and fitness of terrestrial animals. Additionally, road de-icing salt has been shown to negatively affect developing wood frog larvae and lengthen their time to metamorphosis (Sanzo and Hecnar 2006). Likewise, exposure to insecticides has also been shown to directly and indirectly affect survival, development, and metamorphosis of amphibian larvae (Boone and Semlitsch 2001; Hayes et al. 2002; Metts et al. 2005). Various pesticides and environmental pollutants have a variety of endocrine-disrupting effects (e.g., Guillette et al. 2000) that have resulted in many developmental and reproductive problems in alligators (*Alligator mississippiensis*, Guillette and Gunderson 2001). Banded watersnakes (*Nerodia fasciata*) collected from contaminated sites have been shown to accumulate toxic substances such as selenium, cadmium, and arsenic within their livers (Hopkins et al. 1999). Hopkins et al. (1999) also demonstrated an increased metabolic rate in snakes with high contaminant loads which could lead to deleterious effects on snake fitness. Lastly, studies on environmental contamination in turtles have found that direct effects are detectable in both eggs and hatchlings (e.g., Bishop et al. 1998). Due to the physiological sensitivities of many amphibians and reptiles, herpetofauna are promoted as ideal indicator species for environmental contamination investigations (e.g., Lamb et al. 1995).

Research challenges

USDA

Environmental contaminants come from many different sources, including chemicals applied to crops and industrial pollution

Although some forms of environmental pollution result in immediate mortality, many have chronic, sublethal effects that compromise fitness in the form of malformations and reduced survivorship and breeding ability (e.g., Hayes et al. 2002). Ecotoxicological research often requires long-term physiological studies to determine the true extent and severity of contaminants. This necessity complicates adequate data collection for the Food and Drug Administration (FDA) and Environmental Protection Agency (EPA) substance approval processes.

iStockphoto

Industrial pollution is another major source of environmental contamination.

Potential solutions

The most obvious, and likely the most effective, solution for reducing environmental pollution impacts on amphibians and reptiles is to regulate the release and disposal of toxic substances. Habitat contamination is extremely difficult to correct retroactively due to the persistence and easy dispersal of many pollutants. The retention of buffer zones surrounding critical habitat can assist in habitat protection by diluting or preventing pollution in areas of high ecological sensitivity (e.g., Gibbons 2003). Legislation needs to assume a landscape-level perspective, include air and water policies, be enforceable, and support research and monitoring. In addition to non-release or release-reduction strategies, ceasing production of certain chemicals, particularly endocrine-disrupting agents, should be beneficial not only to the health of herpetofauna, but to humans as well (Colburn et al. 1997).

INVASIVE SPECIES

Effects

Michael Marchand

Herpetofauna can also become problematic when they become established outside their native range.

Because species expansions occur via natural mechanisms, we define invasive species as nonindigenous organisms whose presence is attributed to introduction by humans. Invasive species range from plants to invertebrates and vertebrates, and can pose a risk to entire ecological communities because a single species can disrupt an entire food web by depredating or outcompeting native species (Gibbons et al. 2000; Semlitsch 2003) or altering evolutionary pathways (Mooney and Cleland 2001). More than 120,000 invasive microbes, plants, and animals have been identified in the United States, resulting in both positive and negative economic impacts. However, invasive species are ultimately estimated to cost the United States over $314 billion per year in damages (Pimentel et al. 2001). Invasive plants can alter the structure of the vegetation community and subsequently affect ecosystem dynamics such as predator-prey interactions. In some instances, invasive plant species have been implicated as a contributor in the decline of amphibians (Maerz et al. 2005) and reptiles (Stewart et al. 1993).

Invasive invertebrates can cause problems for herpetofauna in subtle, indirect, or direct, and sometimes dramatic, ways. One of the primary invasive invertebrate species of concern in the Southeast is the red fire ant (*Solenopsis invicta*), whose range is currently expanding in northerly and westerly directions. Fire ants spread rapidly due to their ability to readily colonize disturbed habitats, such as roads and powerlines (Stiles and Jones 1998) or forest clearcuts (Todd et al. 2008). Fire ants have proven problematic in depredating neonatal alligators (Allen et al. 1997), salamanders (Todd et al. 2008), and both egg-laying marine (Moulis 1997) and terrestrial (Buhlmann and Coffman 2001) turtles. Although undocumented, fire ants probably affect egg-laying snakes as well (Tuberville et al. 2000).

Exotic earthworms are also a topic of increasing interest (Hendrix and Bohlen 2002), but effects on amphibians and reptiles are not yet fully understood. However, they can be a supplementary prey source for generalist amphibians (Maerz et al. 2005). Herpetofauna can also serve as dispersers for invasive species as seen with the 29 tick species that have entered the United States on imported reptiles since 1962 (Burridge and Simmons 2003).

Scott Bauer, USDA

Invasive species, such as fire ants, also pose a threat to reptiles and amphibians

Invasive vertebrates, including herpetofauna themselves, can have negative effects on amphibians and reptiles. For example, fish stocking has had a major impact on amphibians (Pilliod and Peterson 2000, 2001). Likewise, some invasive amphibians (Cuban tree frog, *Osteopilus septentrionalis*; cane toads, *Bufo marinus*) can out compete native anuran species (Dodd 1997). Invasive species can also become a direct problem for humans, including Nile monitors (*Varanus niloticus*; Enge et al. 2004b) and Burmese pythons (*Python molurus bivittatus*; Reed 2005) in Florida. Some species native to the United States have been introduced outside their native range and have become problem species. For example, bullfrogs (*Rana catesbeiana*) have expanded into the western United States where they are implicated in reducing abundances of Californian yellow-legged frogs (*Rana boylii*; Kupferberg 1997). For a more comprehensive discussion of the exotic herpetofauna of the United States, please see Chapter 2.

Research challenges

A major challenge of the issue of invasive species and their effects on herpetofauna is that introduced species have become geographically widespread and environmentally pervasive, rapidly becoming a worsening problem. Secondary effects can further complicate matters. Thus, an invasive invertebrate can have effects on the survival or abundance of a par-

ticular plant species, altering the habitat of a reptile or amphibian. For example, the hemlock wooly adelgid weakens and kills the eastern hemlock tree, thereby drastically altering the microclimate (moisture, shade, and temperature) along small streams in the Appalachian mountain range, which could have repercussions for salamanders living in these habitats (Brooks 2001). The rate of change in the status of invasive species is so rapid in some systems that meeting new research demands can be difficult. Furthermore, it can be a lengthy process to determine suitable study methods when the species or problem is novel to a particular habitat or region, or when certain conditions or factors necessitate the use of alternative methods.

Potential solutions

Education of the public and officials about the importance of minimizing the spread of species to new areas is a key step in reducing the threat of invasive species. One approach is more stringent regulations and enforcement to reduce the influx of exotic species. This includes a more thorough and careful inspection at our ports and along all transportation routes (planes, trucks, boats, other vehicles). Likewise, widespread monitoring of high-risk areas (i.e., those with high flow of goods from other countries) could help in alerting scientists to problematic exotic species before they become irreversibly established. Further, the distribution or identification of sources for invasive species could assist land managers. Additionally, many invasive plant species are spread via the horticulture and landscaping industry, a sector that should be targeted for outreach and information distribution. Finally, regional or statewide restrictions on ownership of known invasives such as bullfrogs, African clawed frogs, or red-eared sliders may help reduce the spread of many invasive animals.

UNSUSTAINABLE USE

Effects

Sustainable use is the harvest of individuals from wild populations at levels that support their persistence into the unforeseeable future. Inherent in this definition is the assumption that harvest levels do not alter natural population dynamics. Herpetofauna are harvested for many different reasons, including food, skin, sport, pets, or curios such as jewelry. Many cultures rely on herpetofauna to feed themselves or to generate income. However, problems develop when too many people are taking too many animals, causing harvest levels to exceed that which can be supported by natural reptile or amphibian populations.

Amphibians are frequently harvested for the pet trade, food, biological and medicinal supplies, and for bait (Jensen and Camp 2003). Both anurans and salamanders are popular pets; however, some of the greatest levels of wild take for the pet trade occur in other countries with groups such as the brightly colored poison dart frogs (*Dendrobates* and *Phyllobates*; Jensen and Camp 2003). There was also a substantial frogging industry in Florida prior to World War II (Enge 1993). Currently, there are farms that produce captive-bred bullfrogs (*Rana catesbeiana*) to supply restaurants with frog legs (Lutz and Avery 1999). These commercial farms also produce animals for biological and medicinal outlets (Gibbs et al. 1971), but many bait shops still obtain wild-caught salamanders from local collectors (Jensen and Waters 1999).

There has been an extensive amount of reptile harvest, much of which has been unsustainable (Gibbons et al. 2000). Crocodilians have been harvested primarily for their meat and skin, but ornamentals, such as teeth, have also been sold in the tourist industry. Harvest for the leather trade reached levels that nearly drove several crocodilian species to extinction (Brazaitis 1989). Legal protection was designated in addition to the establishment of alligator and crocodile (*Crocodylus acutus*) farms and regulated take of wild populations in Florida. For instance, alligators over 1.2 m (David et al. 1996) or eggs and hatchlings are taken due to the low impact on recruitment by removal at these life stages (Rice et al. 1999). As a result, many of these populations are now considered stable (King 1989). Crocodilians are an example of how sustainability is achievable if programs are designed and implemented in a manner appropriate to the natural history of the animal.

Snakes in the United States are harvested for the pet, meat, and skin trades. Based on the LEMIS (Law Enforcement Management Information System) database managed by USFWS, an estimated 184,000 snakes representing 77 taxa were exported from the United States for the pet trade during 1996-2000; 56% of these exports were declared wild-caught snakes (Reed et al. 2004). Rattlesnakes, primarily of the genus *Crotalus*, are harvested in rattlesnake round-ups for the meat and skin trade. Although snakes can be collected by means such as gassing gopher tortoise burrows, the vast majority of captures are incidental collections of both dead and live specimens found on the road (Berish 1998; Fitzgerald and Painter 2000). Although the effect of snake harvest on some local populations should be taken seriously, the numbers are not nearly as staggering as the number of snakes lost to road mortality (Reed et al. 2004).

Turtles are unlikely candidates for sustainable harvest due to the fact that they are long-lived animals with low juvenile survivorship. While turtle harvest occurs throughout the United States, the situation is most pronounced in the southeastern United States where the highest turtle diversity occurs nationwide. Many turtle species have been incorporated into the pet trade, and documented declines stemming from the pet trade have been reported for the box turtle (*Terrapene carolina*, Lieberman 1994) and the bog turtle (*Glyptemys muhlenbergii*, Copeyon 1997). Turtle harvest for food is applicable for only a few species in the United States, but is widespread globally, particularly in Asian cultures. However, in the late 1800s to early 1900s, diamondback terrapins (*Malaclemys terrapin*) were harvested as a gourmet food item (Carr 1952); in some parts of the Northeast, many diamondback terrapin populations are still exploited for the international food trade. After suffering severe reductions, harvest of alligator snapping turtles (*Macrochelys temminckii*) has only recently been effectively outlawed. Lastly, sea turtles harvest is prevalent globally, but the main issue in the United States is incidental bycatch of sea turtles by the fishing industry, an activity which has been identified as contributing to population declines (Hayes et al. 2003). The shrimp industry has been an area of substantial concern and Turtle Excluder Devices (TEDs) are now a requirement for shrimp vessels (Murray et al. 1992).

Kurt Weiskotten

Research challenges

When developing effective sustainable take models, life-history characteristics of the proposed species must be incorporated. Long-lived, late maturing animals with low fecundity (i.e., reproductive output) cannot be harvested at the same annual rate as high-turnover species (species with high reproductive output and shorter lives) simply because they cannot recover as readily. It is challenging to determine what level of take is sustainable when data elucidating basic biology of many species are still missing. Furthermore, the majority of harvests go undocumented, making proper enforcement difficult. Much of what is documented is not reported at the species level and completely lacks locality information, making impact assessment difficult (Schlaepfer et al. 2005). Lastly, the issue of sustainable harvest is incredibly political as it is often rooted in long-term social or cultural traditions. Therefore, political issues must be taken into account when developing regulations or laws. Fortunately, in the United States, very little harvest of reptiles or amphibians is conducted out of necessity for feeding oneself or family.

Potential solutions

While it is necessary to implement legislative measures to establish a legal framework, laws are difficult to enforce and frequently are not respected by some collectors or others. Farms that produce captive individuals to sell have provided an excellent alternative for species that have high reproductive rates and are suitable to captivity. The concept of sustainable harvesting can best be achieved through education and increased communication with resource users; the basic goal of both scientists and harvesters is the same, albeit for different reasons - to promote the persistence of the species of interest.

DISEASE AND PARASITISM

Effects

Disease is a phenomenon that poses a threat to biodiversity (Daszak and Cunningham 2000) and has become a topic of concern for herpetofauna across the globe. Animals that rely on metapopulation dynamics (e.g., amphibians) or those with extensive social behaviors (e.g., gopher tortoises [*Gopherus polyphemus*]), are particularly vulnerable as they have high levels of interaction among individuals (Hess 1996). Furthermore, a diversity of helminthian parasites can be found on both amphibians and reptiles, so many that new species have been described after being found on host herpetofauna (Pérez-Ponce de León

et al. 2002). Due to the diversity of habitats occupied and ecological strategies represented by amphibians and reptiles, herpetofauna serve as unique models in studies of host-parasite relationships (Aho 1990).

Forrest Brem

A frog infected with chytid fungus.

Amphibian disease is justly causing much alarm due to the pathogenic chytrid fungus *Batrachochytrium dendrobatidis* (*Bd*) which can quickly kill otherwise healthy individuals (Daszak et al. 1999). Although many population die-offs have occurred in Central America and Australia (e.g., Berger et al. 1998), Bd has not generally been attributed to any specific extinctions in the United States, but has been documented extensively in many North American amphibians (Daszak et al. 2005). Another source of massive die-offs of amphibians is an iridovirus (*Ranavirus* sp.) to which larval, metamorphosing, and recently metamorphosed amphibians are susceptible (e.g., Jancovich et al. 1997). These epizootics generally occur in disturbed habitats and have resulted in local population declines, but not extinctions (Carey et al. 2003). The mechanisms of overland dispersal for both pathogens are not clear; however, they appear to be capable of spreading rapidly across large areas (e.g., Whiles et al. 2006). Human transport on clothing or gear is one possibility (Carey et al. 1999), in addition to the introduction of invasive fish species (Carey et al. 2003). Limb deformities and reduced survivorship in amphibians have also been noted as a result of trematode infection (Pacific treefrogs, *Hyla regilla*, Johnson et al. 1999).

While amphibians are currently receiving much attention due to acute die-offs, many reptile diseases are also emerging and are of utmost importance. Gopher tortoises are at risk for contracting the readily contagious and lethal Upper Respiratory Tract Disease (URTD) caused by the bacterium *Mycoplasma agassizii* (Smith et al. 1998), a disease that surfaced in their sister species, the desert tortoise (*Gopherus agassizi*) (Peterson 1994). Lovich and colleagues (1996) have noted shell lesions on yellow-bellied sliders (*Trachemys scripta*) and river cooters (*Pseudemys concinna*), apparently an unrelated affliction of unknown origin. The authors also documented shell sloughing in Barbour's map turtle (*Graptemys barbouri*), but no shell disease was observed as in other species. In another study by Dodd (1988), flattened musk turtles (*Sternotherus depressus*) were found suffering from bacterial septicemia that affected shell health. The population declined dramatically that year and high incidence of infection seemed to be the primary cause. Additionally, green sea turtles (*Chelonia mydas*) have experienced severely reduced fitness and functionality due to viral fibropapillomatosis that occur as growths and limit their functionality (Herbst 1994). Snakes are hosts for many parasites (Ali et al. 1984; Pérez-Ponce de León et al. 2001) and the extent of parasite loads in snakes appears to be grossly underestimated (Cameron A. Young, personal communication). Fitness and mortality implications of parasites on snakes are not well understood.

Research challenges

One of the most challenging aspects about protecting wild herpetofauna from disease is the spatial scale at which the problem occurs, even for a single pathogen. Some diseases appear in remote locations that are difficult to access for basic ecological studies. The temporal scale is also problematic with diseases, such as the chytrid fungus, that act on their host organisms with a rapidity that complicates scientific study. Additionally, monitoring disease is challenging because some reptile species are innately difficult to detect due to covert behaviors which may be amplified when they become ill. Disease outbreaks can occur naturally and may not be remedied completely; however, it is possible that the central source contributing to global epidemics is human activity. An increase in air and water pollution compromises wildlife health and increases vulnerability to disease. Additionally, travel across the globe enables a reliable and rapid medium for disease dispersal.

Potential solutions

The first step in understanding and solving the global problem of disease outbreaks is to recognize that there is no single solution. Like wildfires, some disease occurrences are natural and may be necessary in maintaining natural ecological fluctuations. Increasing awareness and information sharing between public citizens and international scientists is critical

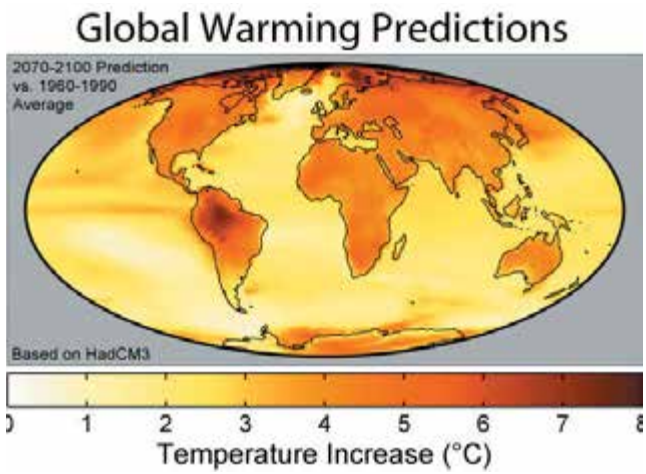
to assessing and combating a global problem. Online information sources and reporting programs or education in local schools and community gatherings are examples of activities that can increase community initiative. Secondly, some outbreaks can be prevented by avoiding the release of potentially infected animals from captivity or from other geographical locations. Monitoring both international and domestic wildlife trade for diseased animals would also reduce pathogenic transfers (e.g., Karesh et al. 2005). In these latter instances, disease issues and invasive species problems become one and the same. Efforts by field researchers to minimize the spread of disease between study sites, including sanitization of boots and equipment, are critical. Lastly, maintaining strong environmental standards and pollution reduction legislation will provide a cleaner, healthier environment that can increase the quality of life for both wildlife and humans.

GLOBAL CLIMATE CHANGE

Effects

Global climate change is a contentious issue that is often politicized, frequently debated, and commonly surrounded with uncertainty. Nevertheless, global climate change has historically been the rule rather than the exception (Carey and Alexander 2003). Climatological records based on proxy data such as growth rings in trees and coral colonies, cores of glacial ice and coral reefs, and sediment samples from many regions suggest that periods of climatic stability have frequently been punctuated by abrupt changes (Alley et al. 1997; Lang et al. 1999; Taylor 1999; Birks and Ammann 2000). It is generally safe to assume that the global climate has continually changed throughout the evolution of all living organisms. However, there is general concern that the recent changes in global climate are occurring at an unprecedented rate (Schneider and Root 1998) and that many species may have difficulty adapting to such rapid change, particularly in light of the many other human-induced threats that they face. Global climate change is generally used to describe the ongoing shifts in regional climate that result in changes in temperatures and rainfall but also encompasses the topic of ozone depletion. In the past century, the average global surface temperature has increased by 0.5 ºC, although some regions have experienced cooling rather than warming (Easterling et al. 2000). Importantly, a recent study has summarized the overwhelming scientific support that the climate is generally warming globally (Oreskes 2004).

The effects of climate change on herpetofaunal populations are generally unknown, although they may be classified into two categories: direct and indirect effects. Climatic factors may directly affect herpetofaunal populations through changes in the time available for development or activity. Temperatures or moisture patterns do not necessarily have to exceed lethal limits to affect a population. Although species declines have been correlated to climatic factors (Daszak et al. 2005), few studies have documented whether temperatures or moisture levels have directly killed individuals. Of the herpetofaunal taxa, amphibians may be most likely to be influenced by changing climatic factors due to their high levels of evaporative water loss, facilitated by high temperatures. Work by Blaustein et al. (1994) demonstrated that increased UV-B radiation significantly reduced survival in larval salamanders. Similarly, Baud and Beck (2005) showed that tadpoles of the southeastern species, spring peepers (*Pseudacris crucifer*), were negatively affected by UV-B. However, the ability of UV-B to penetrate pond water and damage developing embryos and larvae remains uncertain.



Robert Rohde

Predicted distribution of temperature change due to global warming from the Hadley Centre HadCM3 climate model.

Indirect effects from global climate change will arguably have a greater impact on reptiles and amphibians than direct effects. Species distributions and community compositions may shift as climate change alters habitat and prey distributions. Droughts and increased temperatures brought about by changes in regional climates may increase the number and severity of wildfires, thereby eliminating much habitat for herpetofauna. Reductions in the amount of regional rainfall may reduce wetland acreage, resulting in declines of aquatic and semi-aquatic species, as well as amphibians that rely on wetlands for larval habitat. Furthermore, the effects of global climate change may inter-

act and become magnified by habitat fragmentation. Fragmented landscapes may impede the ability of species to respond to climate-induced habitat changes (Halpin 1997). For example, migratory success of individuals will likely be diminished as they encounter increasingly fragmented landscapes.

Indirect effects of climate change on herpetofaunal reproduction pose other important concerns. Yearly reproductive migrations of pond-breeding amphibians correlate with seasonal changes in temperature and rainfall and several studies have examined the influence of climate change on the arrival times of breeding adults (summarized in Carey and Alexander 2003). Some of these studies have found conflicting results, but several have demonstrated that spring-breeding anurans and salamanders have begun reproducing earlier during recent, warmer years (Beebee 1995; Gibbs and Briesch 2001). Besides affecting the phenology of migrations, climate change that alters rainfall patterns may differentially affect some pond-breeding amphibians that rely heavily on adequate rainfall to make their overland migrations (Todd and Winne 2006), possibly affecting competitive larval interactions. Among reptiles, global climate change may have the greatest impact on species with temperature-dependent sex determination (Janzen 1994). Unless animals exhibit behavioral changes in nesting strategies or pivotal temperatures follow changes in climate, sex ratios may become skewed, thereby affecting population demographics and persistence (Gibbons et al. 2000).

Climate change also indirectly affects herpetofauna by providing favorable conditions for the introduction and spread of invasive species or emergent diseases. Although the mechanisms are unclear, changes in habitat and turnover in community composition may provide non-native species with a chance to invade new habitats. Similarly, alteration in regional climates is believed to have contributed to declines of many amphibian species by creating favorable conditions for pathogen growth or otherwise facilitating the spread of epidemic diseases, specifically the chytrid fungus (Pounds and Crump 1994; Pounds et al. 1999; Burrowes et al. 2004; Pounds et al. 2006).

Research challenges

Limited research has been conducted on the effects of climate change on amphibians and reptiles of the United States and virtually all lines of inquiry still demand intensive study. We need to determine the temperature and moisture tolerances of amphibian and reptile species, as well as their means for behavioral compensation to extreme conditions. Studies of herpetofauna in altered habitats may serve as proxies toward this end. Additionally, long-term studies on the reproductive phenology of amphibians based on call surveys or drift fence studies could shed light on the effects of climate change. Carefully designed inventory and monitoring studies can examine possible changes in the distributional status of herpetofaunal species as climate patterns change. Lastly, pivotal temperatures and nesting strategies of reptiles with temperature-dependent sex determination need to be carefully studied for changes in nesting behaviors and nest-site locations.

Potential solutions

There is general concern that the highly elevated atmospheric concentrations of CO_2 and other greenhouse gases may be driving the rapid climate change we are currently experiencing. Current atmospheric greenhouse gas levels are at all time highs (based on 450,000 years of data), presumably from anthropogenic sources such as the burning of fossil fuels. Although reductions in greenhouse gas emissions have been suggested as a way to reduce the potential magnitude and speed of climate change, the uncertainty surrounding future climate predictions and the feedback from anthropogenic causes has led many people to postpone action in this arena. While inaction on this front may have disastrous consequences for global biodiversity and human health, there is a real, short-term financial cost to switching to alternative fuels and reducing pollutant emissions that must be considered when making policy decisions. There are additional actions that may be more immediately implemented to help reduce the effects of climate change. In particular, preservation of intact landscapes and habitat corridors surrounding natural areas, state parks, national forests, and national parks may promote the ability of herpetofauna to adapt to climate change. Lastly, preventing the introduction and spread of non-native animals and emerging diseases that may act synergistically with climate change is of utmost importance.

INVENTORY VERSUS MONITORING

Kurt A. Buhlmann

Distinguishing the goals of an inventory program from a monitoring program is important and sometimes under-appreciated. Inventory and monitoring are two separate and distinct approaches for assessing biodiversity of an area at the species level. By definition, resource managers cannot monitor species without

knowing what species they have. Therefore, inventory always precedes monitoring. In this document, we distinguish between two different levels of inventory, a rapid (short term) assesment and a comprehensive project, as well as two levels of monitoring, a detected/non-detected and a population-level analysis. The purpose of the section below is to aid resource managers in asking: what type and level of information is needed, what time frame may be required to collect the desired field information, and which techniques are the most effective and appropriate.

A quick summary of techniques used and recommended by herpetologists is found in the Species x Techniques Table in Chapter 5. All amphibian and reptile species are listed, as are the two categories for inventory, and the two categories for monitoring.

Inventory

The goal of an inventory is to develop a list of species known to occur in a prescribed managed area or habitat. A species inventory can be accomplished by direct field surveys, compiling records from previous surveys, canvassing museum collections, and obtaining incidental records from reliable sources. Ideally, an inventory can establish which species are present and which ones are presumably absent, although declaring that a rare or secretive animal is not present must be done with caution. A thorough inventory for reptiles and amphibians often involves sampling in different seasons and/or years and the use of many survey techniques.

Depending on the knowledge needed and the time frame available, a field inventory for amphibians and reptiles may be cursory or thorough. Some basic inventories may be conducted over the course of a few site visits, while a comprehensive survey may extend over several seasons and even multiple years. A Rapid Assessment is often conducted during a short window of time. Many consultant surveys for developers involve short term searching of the habitat or site, coupled with an expert opinion of the species that may occur there. Note that a Rapid Assessment (presence/absence) for some species is nearly impossible, even if it's in the middle of the season for peak activity. For example, in Virginia, the timber rattlesnake [Coastal Plain population (Canebrake)] is virtually impossible to find without an extensive survey and even then a false-negative result is likely to occur. For these species, presence/absence should be based on a combination of historical records and a Habitat Assessment. Knowledge of the target species' known general distributions, preferred habitats (i.e., soil type, wetland type, forest stand age), and the geographical region (ecoregion or physiographic province) is required. Such short-term surveys are most often incomplete for amphibians and reptiles because of the highly seasonal patterns of activity displayed by most species. For example, an inventory conducted to document presence of adults of ambystomatid salamanders is likely to be unfruitful if conducted outside of the winter breeding period, when these otherwise highly fossorial species migrate across the surface to the breeding ponds. For the remainder of the year, they remain buried in leaf litter, or within a network of small mammal tunnels. A Rapid Assessment may detect the most common species or the ones that are most generalist in their choice of habitat. However, the goal of many inventories, especially those conducted on natural areas, or lands proposed for development, often require knowledge of the rare species present. In remote, tropical areas of the world, Rapid Assessments are often the types of inventory that are feasible, given the difficulties accessing terrain, and lack of established field stations. Given the secretive nature of many amphibians and reptiles, their limited seasonal detectability, and annual variation related to weather patterns, multi-year Comprehensive Inventory projects are necessary to develop complete lists of species occurring in a given area (see species accumulation curve in Chapter 6).

Monitoring

The purpose of monitoring is to evaluate the persistence, trends, and demography of target species' populations in a defined area. Depending on the scope of the project, monitoring may be designed to confirm continued presence of a species throughout its previously identified range. Alternatively, a monitoring program may be designed to detect changes in numbers, age classes, and health of specific populations in a discreet area, such as a national park or forest, or other natural area. By definition, resource managers cannot monitor species without knowing what species they have. Therefore, inventory always precedes monitoring. In some situations, the simple detection of a species on a defined area during an inventory does not necessarily mean that a population exists to monitor. Inventories often detect individuals of species that are migrating between habitats, and the habitats they require may not even occur on the management area. Thus, evaluation of the species list produced from an inventory must be done in conjunction with an evaluation of the habitats present and the prognosis for viable populations to occur in the area.

Once target species are identified as occurring on specific defined areas, monitoring may provide information on such population features as periods of seasonal activity, population density and abundance, and movement patterns in response to environmental variables such as temperature and rainfall. Long-term monitoring programs allow annual comparisons within a species and may reveal that particular species are declining or increasing in abundance in an area.

The goals of a monitoring program will vary with the needs of the resource manager. Detecting the continued presence of a species on an annual or multi-year program may be sufficient, especially for perceived common species. Monitoring the trends in population size, recruitment, and health for species of conservation concern may require more intensive, time-consuming techniques. The Species x Techniques Table in Chapter 5 lists techniques recommended for both the detection of a species' continued presence and statistical analysis of population trends.

In recent years there has been a shift within the scientific community as to how monitoring is defined and conducted. Many monitoring programs for herpetofauna and other wildlife begin as a single inventory event and are expanded into monitoring by conducting inventories at regular time intervals. Many successful monitoring programs begin without clear objectives and study plans, but there are limitations associated with this. For example, extraneous data may be collected and it may be impossible to collect some data due to the sampling scheme. There is also the issue that wildlife, especially amphibians and reptiles, may go undetected and that detectability varies due to a variety of factors, including site differences and environmental conditions. Researchers have been developing methods for dealing with the limitations of traditional monitoring programs (MacKenzie et al. 2002, 2003, 2004, 2006). These methods have become more well known and are being adopted increasingly by ecologists and biologists.

Monitoring becomes a more powerful tool when it involves clearly determining objectives, making a priori hypotheses, and incorporating detection probabilities. A monitoring study will yield much more accurate and useful data if a detailed plan has been made in advance that takes detectability issues into account. Often habitat data combined with utilization can focus monitoring efforts in specific habitat areas, thus increasing the detectability of rare or cryptic species (MacKenzie et al. 2006). Models such as PRESENCE have been developed to assist in this process (http://www.mbr-pwrc.usgs.gov/software/presence.html). The United States Geological Survey (USGS) and Amphibian Research and Monitoring Initiative (ARMI) websites have additional information on this topic (http://armi.usgs.gov; http://fresc.usgs.gov/products/papers/1443_adams.pdf).

Long-term monitoring of habitat availability and condition is often as important as monitoring species presence. Monitoring of habitat condition and evaluating trends in habitat condition lead to the most important step, which is actual habitat or population management. Inventory and monitoring are often identified as goals of a conservation program. However, simply knowing what is present and what is declining or increasing does not equate to conservation. One needs to identify the actions and funding needed. Of course, a further application of a monitoring program might also be to identify gaps in understanding of species biology and ecology, thus leading to basic research questions and projects. Thus, a complete conservation program for amphibians and reptiles involves inventory, monitoring, management, and research. Feedback loops should function between these categories.

INVENTORY AND MONITORING PROGRAMS AND CONSERVATION

Kurt A. Buhlmann

The observed declines in amphibians worldwide and in the United States and Canada have been well-documented in the literature over the past two decades (Phillips 1994; Wake 1991; Green 1997; Lannoo 1998). Appropriately, efforts to collect baseline data about the occurrence and distribution (inventories), as well as the status of populations (monitoring), have been initiated for amphibians. Surprisingly, fewer comprehensive projects have been initiated for reptiles, yet the recent literature indicates that reptiles, specifically the turtles and crocodilians, are as threatened as the frogs (Gibbons et al. 2000). Data on the declines of snakes and lizards are difficult to evaluate, often because simply detecting their presence through inventory is difficult and actually monitoring trends in populations has been even more so.

In the United States, a rather comprehensive knowledge base exists regarding the distribution of the nation's amphibians and reptiles. Museums around the country and elsewhere contain specimens that indicate not only where species occur, but also where

they occurred historically. State Natural Heritage Programs have diligently collected information for the past several decades on the occurrences of rare species (element occurrences) and have assembled those databases for conservation planning (see www.natureserve.org). Most states have published field guides illustrating the distribution and ecology of amphibians and reptiles within their political boundaries. More recently, the PARC program has produced publications for landowners that provide guidance about the management of habitats for amphibians and reptiles (Bailey et al. 2006; Mitchell et al. 2006; Pilliod and Wind 2008). Thus, resource managers in the United States have a wealth of information available to them to manage amphibians and reptiles.

Around the world, the knowledge base regarding the distribution and ecology of amphibians and reptiles is much more limited. Non-governmental organizations (Conservation International, World Wildlife Fund, Wildlife Conservation Society, The Nature Conservancy, and others) are expending funds and establishing programs to collect basic information about amphibians and reptiles and their status. University scientists are conducting research that also leads to increased knowledge of amphibians and reptiles. A network of field stations, especially within the world hotspots of biodiversity (Mittermeier et al. 1999), are being established in many tropical areas around the world in order to establish monitoring programs that include amphibians and reptiles (http://www.teamnetwork.org).

The USGS Amphibian Research and Monitoring Initiative (ARMI) (http://armi.usgs.gov) has produced standardized methodology to help resource managers develop programs and monitor amphibian populations, as well as serve as a national center to detect and evaluate trends in amphibians. ARMI is the national research and monitoring program initiated by the Department of Interior under the directive of the President and Congress to respond to indications of worldwide declines in amphibian populations. The USGS, the science and research bureau for the Department of Interior, was given lead responsibility for planning and organizing the program in cooperation with the National Park Service, U.S. Fish and Wildlife Service, and Bureau of Land Management. The ARMI program is also investigating incidences of malformations in amphibians. In conjunction with ARMI, the U.S. Fish and Wildlife Service has initiated a nationwide survey for malformed amphibians on national wildlife refuges. In conjunction with the USGS National Wildlife Health Center in Madison, Wisconsin, examinations of amphibian specimens are conducted to determine the causes of the malformations. The National Park Service has begun an extensive program to inventory and monitor natural resources within national parks. The intent of the monitoring data is to provide better information for decisions on management of park resources. Many national parks are including amphibians on their lists of species of concern for monitoring. The North American Amphibian Monitoring Program works with the ARMI program and many partner organizations to manage a long-term roadside calling survey for frogs. An opportunity for resource managers to involve their areas exists (www.pwrc.usgs.gov/naamp).

Finally, the PARC Program is a partnership dedicated to the conservation of both amphibians and reptiles primarily through conservation of their habitats. PARC participants include state and federal agencies, industry, conservation organizations, research laboratories, museums, and the academic community. PARC focuses not only on endangered and threatened species, but also works toward the objective of "keeping common native species common." To this end, this PARC inventory and monitoring guide is designed to elaborate on techniques and serve as additional supplemental information, as it represents the experiences of many herpetologists within the PARC community. It also attempts to integrate knowledge about inventory and monitoring techniques for reptiles, which has not been a focus of other nationally based programs to date. However, this guide is not proposing new protocols for monitoring amphibians, and for that information we direct the reader of this document to the USGS ARMI website and the programs described above (http://armi.usgs.gov).

Northern Pacific rattlesnake (*Crotalus viridis oreganus*)

Tracy Brehm

CHAPTER 2: AMPHIBIANS AND REPTILES OF THE UNITED STATES

J. Whitfield Gibbons

The diversity of amphibians and reptiles in the United States rivals the herpetofaunal species diversity of most countries. However, despite the widespread distribution and abundance of this fascinating array of animals in many regions, herpetofauna are among the most poorly represented vertebrates in most state and federal wildlife programs. The reasons pertain to the historical application of wildlife programs to game and commercial species for practical purposes. Nonetheless, with the increasing awareness among land managers and conservation biologists that all components of natural habitats are important in maintaining sustainable ecosystems, mounting emphasis is being placed at local, regional, and national levels on programs that offer insights into, and increase knowledge of, the ecology and life histories of native herpetofauna. Determining basic ecology as well as the population status and trends for target species are critical for proper and prudent land management.

IMPORTANCE OF NATURAL HISTORY DATA TO INVENTORY AND MONITORING

Knowing the geographic range and the natural history (i.e., general ecology and life history) of a species are essential first steps in selecting or developing proper inventory and monitoring techniques. Thus, basic seasonal activity patterns, such as whether an amphibian species lives on land most of its life and moves to isolated wetlands for breeding during rainy nights in the fall (e.g., marbled salamanders), or remains terrestrial for its entire life (e.g., slimy salamanders), can be vital information when setting up a sampling program. Knowing daily activity patterns, such as whether a species is active only at night (e.g., scarlet snake) or only during the day (e.g., racers) can also be important. And of course the geographical range distribution must be known in order to determine whether a particular species can be expected to be absent or likely to be present at a particular site.

It is not the intent of this document to provide all of the background information currently available on natural histories for the wide diversity of herpetofaunal species native to each region of the United States. Such information is best acquired from a combination of regional, state, or local field guides (see “Regional resources” at end of chapter) and from the experience of herpetologists who are familiar with an area of interest. Herpetological state atlases have been conducted in or are currently underway in many states and can be extremely useful in identifying particular counties or areas of a state where selected species have been documented. State departments of natural resources should be able to provide the most up-to-date distribution records for amphibians and reptiles within the state.

NOMENCLATURE FOR NATIVE SPECIES

The herpetofauna of the United States comprise approximately 460 species, of which about 200 are amphibians and 260 are reptiles. The Southeast has the highest species diversity with approximately 245 species, of which 128 are amphibians and 117 are reptiles. The next most diverse region for herpetofauna, with a total of approximately 150 species, is the Southwest, with about 117 reptiles but only 33 amphibians.

J.D. Wilson

Copperhead (*Agkistrodon contortrix*), juvenile

J.D. Wilson

Green salamander *(Aneides aeneus)*

The specific numbers given above will change from year to year based on research findings, but the basic proportions among major taxonomic groups and regions should not shift appreciably. The exact number of species will remain a dynamic one primarily because of a changing scientific nomenclature that results from research that leads to reinterpretations of the ancestral relationships of different species, the reclassification

Matthew Kull

Gray treefrog *(Hyla versicolor)*

Chauncey Leahy

Northern brown snake *(Storeria dekayi)*

of species based on new genetic findings, and taxonomic changes resulting from reevaluations based on the rules of scientific nomenclature. Occasionally, the discovery of a formerly unknown species will increase the number of species documented from the region, and unfortunately the disappearance of a species can reduce the number believed to be present. Nonetheless, although the accepted numbers of species in a taxonomic category may change, the higher level relationships within the amphibians and reptiles are relatively consistent. The following provides an overview of North American herpetofauna based on the traditional nomenclatural classification scheme of ranking classes, orders, families, and genera (sing. genus). The names below each family name represent the single genus or two or more genera in the family. Species names within each genus are given in Table 5-1 (Species x Techniques Table) at the end of Chapter 5.

The use of DNA in genetic research has resulted in reinterpretations of phylogenetic relationships among many animal species, including major groups of amphibians and reptiles. Many of these determinations have been accompanied by recommendations for taxonomic changes that have included genus and family names. Although the intent of investigators is well meaning, not all such interpretations of relationships are accepted by the scientific community, nor are the newly proposed names that reflect the perceived phylogenetic relationships. For purposes of identification of and reference to

species of herpetofauna, we adhere to the policy that "no particular name is more correct than another as long as it is known what species is being referred to" (Gibbons et al. 2008). Therefore, to simplify the dynamic process of what to call different species, we use species names that reflect those in usage and commonly accepted by herpetologists in general field guides and herpetological textbooks of the early 21st century and to which subsequent nomenclatural changes that do become accepted can be readily associated.

Changes to some species, genus, and family names can be expected to be accepted by most of the herpetological community over time, although exactly what will become accepted by the herpetological community can never be assessed with certainty. One of the most sweeping set of recent changes based on reinterpretations and understandings of the evolutionary relationships among all amphibians was led by Darrel Frost of the American Museum of Natural History. Our expectation is that the amphibian taxonomy based on the project known as "Amphibian Species of the World" by him and several colleagues will become the standard for nomenclature of frogs and salamanders of the United States. To assess the status of any family, genus, or species listed in the current guide go online to http://research.amnh.org/vz/herpetology/amphibia/ to determine the taxonomic recommendations.

John White

Speckled kingsnake *(Lampropeltis getula holbrooki)*

John White

Three-toed box turtle (*Terrapene carolina triunguis*)

Jamie Bettaso

Pacific treefrog *(Pseudacris regilla)*

Jamie Bettaso

Rubber boa (*Charina bottae*)

Jamie Bettaso

Western terrestrial gartersnake *(Thamnophis elegans)*, juvenile

Jamie Bettaso

Cascades frog (*Rana cascadae*)

CLASS AMPHIBIA – AMPHIBIANS

Order Caudata - Salamanders
- Family Proteidae
  - *Necturus*
- Family Amphiumidae
  - *Amphiuma*
- Family Cryptobranchidae
  - *Cryptobranchus*
- Family Sirenidae
  - *Pseudobranchus*
  - *Siren*
- Family Ambystomatidae
  - *Ambystoma*
- Family Dicamptodontidae
  - *Dicamptodon*
- Family Rhyacotritonidae
  - *Rhyacotriton*
- Family Salamandridae
  - *Notophthalmus*
  - *Taricha*
- Family Plethodontidae
  - *Aneides*
  - *Batrachoseps*
  - *Desmognathus*
  - *Ensatina*
  - *Eurycea*
  - *Gyrinophilus*
  - *Haideotriton*
  - *Hemidactylium*
  - *Hydromantes*
  - *Phaeognathus*
  - *Plethodon*
  - *Pseudotriton*
  - *Stereochilus*

Order Anura - Frogs and toads
- Family Ascaphidae
  - *Ascaphus*
- Family Rhinophrynidae
  - *Rhinophrynus*
- Family Pelobatidae
  - *Scaphiopus*
  - *Spea*
- Family Bufonidae
  - *Bufo*
- Family Leptodactylidae
  - *Leptodactylus*
  - *Eleutherodactylus*
- Family Hylidae
  - *Acris*
  - *Hyla*
  - *Pseudacris*
  - *Smilisca*
- Family Microhylidae
  - *Gastrophryne*
  - *Hypopachus*
- Family Ranidae
  - *Rana*

CLASS REPTILIA - REPTILES

Order Crocodilia - Crocodilians
- Family Alligatoridae
  - *Alligator*
- Family Crocodylidae
  - *Crocodylus*

Order Testudines – Turtles
- Family Chelydridae
  - *Chelydra*
  - *Macrochelys*
- Family Kinosternidae
  - *Kinosternon*
  - *Sternotherus*
- Family Testudinidae
  - *Gopherus*
- Family Trionychidae
  - *Apalone*
- Family Emydidae
  - *Chrysemys*
  - *Clemmys*
  - *Deirochelys*
  - *Emydoidea*
  - *Graptemys*
  - *Malaclemys*
  - *Pseudemys*
  - *Terrapene*
  - *Trachemys*
- Family Cheloniidae
  - *Caretta*
  - *Chelonia*
  - *Eretmochelys*
  - *Lepidochelys*
- *Family Dermochelyidae*
  - *Dermochelys*

Order Squamata - Snakes and lizards
- Suborders Lacertilia and Amphisbaenia - Lizards and Worm Lizards
- Family Gekkonidae
  - *Coleonyx*
  - *Sphaerodactylus*
- Family Iguanidae
  - *Anolis*
  - *Dipsosaurus*
  - *Sauromalus*
  - *Sceloporus*
  - *Crotaphytus*
  - *Gambelia*
  - *Callisaurus*
  - *Cophosaurus*
  - *Holbrookia*
  - *Petrosaurus*
  - *Phrynosoma*
  - *Urosaurus*
  - *Uma*
  - *Uta*

- Family Xantusiidae
  - *Xantusia*
- Family Teiidae
  - *Aspidoscelis (= Cnemidophorus)*
- Family Scincidae
  - *Plestiodon (= Eumeces)*
  - *Plestiodon (= Neoseps)*
  - *Scincella*
- Family Anguidae
  - *Ophisaurus*
  - *Elgaria*
  - *Gerrhonotus*
- Family Anniellidae
  - *Anniella*
- Family Helodermatidae
  - *Heloderma*
- Family Amphisbaenidae
  - *Rhineura*

Suborder Serpentes – Snakes

- Family Leptotyphlopidae
  - *Leptotyphlops*
- Family Boidae
  - *Charina*
- Family Colubridae
  - *Arizona*
  - *Bogertophis*
  - *Carphophis*
  - *Cemophora*
  - *Chilomeniscus*
  - *Chionactus*
  - *Clonophis*
  - *Coluber*
  - *Coniophanes*
  - *Contia*
  - *Diadophis*
  - *Drymarchon*
  - *Drymobius*
  - *Elaphe*
  - *Farancia*
  - *Ficimia*
  - *Gyalopion*
  - *Heterodon*
  - *Hypsiglena*
  - *Lampropeltis*
  - *Leptodeira*
  - *Masticophis*
  - *Nerodia*
  - *Opheodrys*
  - *Oxybelis*
  - *Phyllorhynchus*
  - *Pituophis*
  - *Regina*
  - *Rhadinaea*
  - *Rhinocheilus*
  - *Salvadora*
  - *Seminatrix*
  - *Senticolis*
  - *Sonora*
  - *Stilosoma*
  - *Storeria*
  - *Tantilla*
  - *Thamnophis*
  - *Trimorphodon*
  - *Tropidoclonion*
  - *Virginia*
- Family Elapidae
  - *Micruroides*
  - *Micrurus*
- Family Hydrophiidae
  - *Pelamis*
- Family Viperidae (= Crotalidae)
  - *Agkistrodon*
  - *Crotalus*
  - *Sistrurus*

EXOTIC HERPETOFAUNA OF THE UNITED STATES

Walter E. Meshaka, Jr.

As stewards of our natural legacy, private and public landowners have a direct interest in knowing what exotic species occur on their properties and in what abundance. With the goal of familiarizing landowners with exotic species of amphibians and reptiles that they might encounter, a brief species account was assembled for each of the established exotic species in alphabetical order of their scientific names within their respective groupings of frogs and toads, lizards, snakes, turtles, and crocodilians. The accounts are intentionally direct but informal and rely heavily but not exclusively, on the included references. In the interest of ease of reading, references are grouped here: Bartareau 2004; Bartlett and Bartlett 1999; Boundy 2004; Bufalino 2004; Burke et al. 2002; Burke and Mercurio 2002; Campbell 1996; Conant and Collins 1998; Carey 1982; Crayon 2005; Dinsmore 2004; Dixon 2000; Dixon et al. 2007; Dundee and Rossman 1989; Eason and McMillan 2000; Enge and Krysko 2004; Enge et al. 2004a,b; Enge et al. 2006; Ernst et al. 1994; Ferner and Ferner 2002; Hare 2006; Jones et al. 1995; Klawinski et al. 1994; Kleopfer et al. 2006; Kraus 2005; Kraus and Duvall 2004; Krysko and Enge 2005; Krysko et al. 2003, 2004; Krysko and Sheehy 2005; Lannoo and Nanjappa 2005; Lever 2003; Lips 1991; McDowell et al. 2006; McKeown 1996; Meshaka 2000, 2001, 2005a,b, 2006; Meshaka et al. 2004a,b, 2005a, 2006a; Meshaka and Layne 2005; Meshaka and Rice 2005; Nelson and Carey 1993; Norden and Norden 1989; Punzo 2001a,b; Reed et al. 2006; Savitzky et al 2002; Skelton and Parmley 2005; Stebbins 2003; Stewart and Lannoo 2005; Thomas 1994; Trauth et al. 2004; Turnbough 2006; Vaughan et al. 1996; Walker and Deichsel 2005; Winn et al. 1999.

The parameters associated with this undertaking are presented here. Taxonomically, the species covered in this treatment are those that are known to not have occurred naturally in the United States, such as the Green Iguana (*Iguana iguana*), and those whose native status in the United States is open to question, such as the Cuban Treefrog (*Osteopilus septentrionalis*) on the Florida Keys or the Stump-toed Gecko (*Gehyra mutilata*) in Hawaii. The species accounts do not include those species that are known to be native within the boundaries of the United States but through human-mediated dispersal have been translocated elsewhere in the country. Examples in this category include species such as the Cane Toad (*Bufo marinus*), Bullfrog (*Rana catesbeiana*), and Red-eared Slider (*Trachemys scripta elegans*). This intermediate category of species is not strictly speaking exotic to the physical boundaries of this study but are at the same time human-mediated colonizers outside of their natural geographic range. No less important in their impact than exotic species but not exotic to the United States, these species warrant recognition in an intermediate category of exotic populations of native species. A formal study of this topic would be very useful. Among the species considered to be exotic to the United States, those not included are species or colonies whose establishment is not confirmed as per the criteria of Meshaka et al. (2004a). Also not included are those exotic species or colonies considered extinct, such as the Italian Wall lizard (*Podarcis sicula*) in Pennsylvania.

J.D. Wilson

Bull frog (*Rana catesbeiana*)

Criteria as per Meshaka et al. (2004a) formed the basis to determine establishment in a state, and a literature search through December 2006, including those studies known to be In Press in 2006, was used to provide a list of states, but not all counties within those states, from which exotic species are known to occur. Sixty-three exotic species of amphibians and reptiles are established in the United States. Taxonomically, most of the species are lizards (n = 52), followed by frogs and toads (n = 6), snakes (n = 2), turtles (n = 2), and crocodilians (n = 1). Thirty-seven of these species are from the Old World and twenty-six species are from the New World. In the United States, most of these species are found in Florida and Hawaii. However, some species occur as far westward on the continent as California or as far northward as Massachusetts, and one species, the Mediterranean Gecko (*Hemidactylus turcicus*), is found astonishingly in 19 states. Most of these species have a close association with humans with respect to habitat and interest as pets or food, and are easily transported unknowingly by humans. Also,

most of these species are generally active at night, not particularly large in body size, and often blend in easily with their surroundings and so they can be overlooked easily. Together, these traits help explain the rapidly growing numbers of exotic amphibian and reptile species and populations. These species do not colonize without actual and potential ecological consequences that may even involve one another (Fig. 2-1). The Species Accounts below provide a first step in becoming aware of the exotic species that might be on managed lands, with examples of how they may interact with various components of those systems.

Walter Meshaka

Figure 2-1. Knight anole *(Anolis equestris)* catching a Cuban Treefrog *(Osteopilus septentrionalis)* in a state park in Southern Florida

Species Accounts

Frogs and Toads

1. *Dendrobates auratus*- The Green and Black Poison Frog is a New World species. Adults can reach about 3.0 cm snout-vent length (SVL). In the U.S., the Green and Black Poison Frog occurs in Hawaii. The species is typically associated with sheltered valley areas. Eggs are laid on land, and tadpoles are carried to the water. The Green and Black Poison Frog is an insectivore whose ecological impacts in Hawaii are unknown.

2. *Eleutherodactylus coqui*- The Puerto Rican Coqui is a New World species. Adults can reach about 5.8 cm SVL. In the U.S., the Puerto Rican Coqui occurs in Florida and in Hawaii. In Florida, the Puerto Rican Coqui is strongly associated with greenhouses, whereas it is an aggressive colonizer in Hawaii. Adults lay eggs in moist retreats, and the young undergo direct development in the egg, thereby emerging from the egg as miniature versions of the adult stage. The Puerto Rican Coqui is an insectivore whose ecological impacts in Florida and Hawaii are unknown.

3. *Eleutherodactylus planirostris*- The Greenhouse Frog is a New World species. Adults can reach about 3.2 cm SVL. In the U.S., the Greenhouse Frog occurs in Alabama, Florida, Georgia, Hawaii, Louisiana, Mississippi, and Texas. In Florida, the Greenhouse Frog is found in natural, generally mesic habitats, disturbed habitats, and in residential gardens. It fares poorly in frequently burned sandy uplands and avoids very wet habitats. Like the Puerto Rican Coqui, the Greenhouse Frog lays its eggs in moist retreats, and the young hatch as miniature versions of the adult stage. An insectivore, the effects this species has on small, native, semifossorial and fossorial insectivorous species with which it occurs remains unknown. However, its attraction to infrequently burned sandy uplands in Florida is another reason for maintaining the integrity of sandy, upland habitat with the adherence to a rigorous fire management plan.

4. *Osteopilus septentrionalis*- The Cuban Treefrog is a New World species. Adults can reach about 12.5 cm SVL. In the U.S., the Cuban Treefrog occurs in Georgia and Florida. There, the Cuban Treefrog is found in most native habitats, avoiding sandy uplands and open marsh. It is also abundant in many kinds of disturbed habitat. Eggs are laid in shallow fishless systems or occasionally in fishless, weedy, shallow pockets of permanent systems. The Cuban Treefrog eats many of the same prey as its potential native competitors, especially the Green Treefrog (*Hyla cinerea*). It also eats its potential competitors to the extent that its depredations negatively impact the Green Treefrog and the Squirrel Treefrog (*H. squirella*). The Cuban Treefrog also eats other vertebrates, both native (e.g., Eastern Narrowmouth Toad, *Gastrophryne carolinensis*) and exotic (e.g., Indo-Pacific Gecko, *Hemidactylus garnotii*, and Tropical house Gecko, *H. mabouia*), and is thus a threat to a wide range of small native vertebrates in a wide range of places. In Florida, this species is eaten by the Knight Anole (*Anolis equestris*).

John Jensen

Cuban treefrog (*Osteopilus septentrionalis*)

5. *Rana rugosa*- The Wrinkled Frog is an Old World species. Adults can reach about 4.4 cm SVL. In the U.S., the Wrinkled Frog occurs in Hawaii, where it is associated with stream habitats of low and mid-elevations. Eggs are laid in the water. The Wrinkled Frog is an insectivore whose impacts in Hawaii are unknown.

6. *Xenopus laevis*- The African Clawed Frog is an Old World species. Adult females can exceed 14.0 cm SVL. In the U.S., the African Clawed Frog occurs in Arizona and California, where it is highly aquatic in its habits. Males reportedly call underwater, and eggs are laid in the water. Adults consume aquatic invertebrates, fish, and other amphibians. Concern regarding negative impacts by the African Clawed Frog on native frogs and toads is warranted.

Lizards

7. *Agama agama*- The Common Agama is an Old World species. Adults can reach about 30.5 cm total length (TL). In the U.S., the Common Agama occurs in Florida, where it is found on buildings and trees of disturbed habitat. An omnivore, this species can potentially compete with and be a predator of native species, such as the Green Anole (*Anolis carolinensis*).

8. *Ameiva ameiva*- The Giant Ameiva is a New World species. Adults can reach about 61 cm TL. In the U.S., the Giant Ameiva occurs in Florida, where it is found in open, disturbed habitat. The Giant Ameiva is a terrestrial species that actively forages on invertebrates and vertebrates during the heat of the day. Its diet in Florida includes eggs of the Brown Anole (*Anolis sagrei*). In Florida, the Giant Ameiva is ecologically most similar to the Six-lined Racerunner (*Aspidoscelis sexlineata*, formerly known as *Cnemidophorus sexlineatus sexlineatus*), with which it could potentially compete. Likewise, its predatory habits on native species that are capable of using disturbed habitat warrant concern.

9. *Anolis chlorocyanus*- The Hispaniolan Green Anole is a New World species. Adults can reach about 21.6 cm TL. In the U.S., the Hispaniolan Green Anole occurs in Florida, where it favors large trees to climb in disturbed habitat. The Hispaniolan Green Anole is an insectivore, but inclusion of small vertebrates such as other lizards cannot be ruled out. Consequently, the Hispaniolan Green Anole should be viewed as a potential threat to the native Green Anole.

10. *Anolis cristatellus*- The Puerto Rican Crested Anole is a New World species. Adults can reach about 19.1 cm TL. In the U.S., the Puerto Rican Crested Anole occurs in Florida. The Puerto Rican Crested Anole is a ground-trunk lizard of somewhat shady, disturbed habitat, such as tree-lined streets and disturbed, tropical hardwood hammock. This species is an omnivore, feeding heavily on beetles and ants and is a potential predator of the native Green Anole.

11. *Anolis cybotes*- The Large-headed Anole is a New World species. Adults can reach about 22.9 cm TL. In the U.S., the Large-headed Anole occurs in Florida, where it is found on trunks of trees and on manmade structures of disturbed habitat. In Hispaniola, the Large-headed Anole eats invertebrates and small vertebrates and is thought to do the same in Florida. As such, the Large-headed Anole could be a threat to the native Green Anole.

12. *Anolis distichus*- The Bark Anole is a New World species. Adults can reach about 12.7 cm TL. In the U.S., the Bark Anole occurs in Florida, where it is found on the trunks of smooth-barked trees of disturbed habitat. The Bark Anole is an ant-eater whose dietary overlap with the native Green Anole in southern Florida is low. In Florida, the Bark Anole is eaten by the Cuban Green Anole (*Anolis porcatus*).

13. *Anolis equestris*- The Knight Anole is a New World species. Adults can reach about 45.7 cm TL. In the U.S., the Knight Anole occurs in Florida and in Hawaii. In Florida, this species prefers the semi-canopied habitats of disturbed areas, where it dominates generally smooth-barked trees from ground to canopy level. It is an omnivore that both sits and waits for its food and actively forages and is a predator of some native (e.g., Black and White Warbler, *Mniotilta varia*) and exotic (e.g., Cuban Treefrog and Brown Anole) vertebrates alike. In disturbed habitat in Florida, the native Green Anole is often more abundant in the presence of the Knight Anole than when it is alone with the Brown Anole.

14. *Anolis garmani*- The Jamaican Giant Anole is a New World species. Adults can reach about 30.5 cm TL. In the U.S., the Jamaican Giant Anole occurs in Florida, where it is found in disturbed habitat dominated by large shade trees. There, like the Knight Anole, the Jamaican Giant Anole occupies a wide range of heights. It is an omnivore that could potentially impact the native Green Anole.

15. *Anolis porcatus*- The Cuban Green Anole is a New World species. Adults can reach about 21.6 cm TL. In the U.S., the Cuban Green Anole occurs in Florida, where it is found using disturbed habitat in much the same way as its close relative, the native Green Anole. It is an omnivore that can eat surprisingly large lizards. The potential for competition and hybridization with, and predation on, the native Green Anole in disturbed habitats elevates the threat by this species to a level of concern that should not be underestimated. In Florida, the Cuban Green Anole is a predator of the Bark Anole.

16. *Anolis sagrei*- The Brown Anole is a New World species. Adults can reach about 20.3 cm TL. In the U.S, the Brown Anole occurs in Alabama, Florida, Georgia, Hawaii, Louisiana, South Carolina, and Texas. This is an ecologically versatile species that is found in a wide range of disturbed habitats with open, sunny areas. Individuals are generally found close to the ground, although males will occasionally perch higher in the trees. Primarily an insectivore, the Brown Anole also eats the native Green Anole. Interestingly, in Florida, when also in the presence of the Knight Anole, the Green Anole fares better than when alone with the Brown Anole. In Florida, the Brown Anole is eaten by the Knight Anole.

John Jensen

Brown anole (*Anolis sagrei*)

17. *Aspidoscelis motaguae*- The Giant Whiptail is a New World species. Adults grow to about 33.0 cm TL. In the U.S., the Giant Whiptail occurs in Florida, where it is found in open, sandy, disturbed habitat. It is ecologically similar to the Six-lined Racerunner with which it could be a potential competitor.

18. *Basiliscus vittatus*- The Brown Basilisk is a New World species. Adults can reach about 61.0 cm TL. In the U.S., the Brown Basilisk occurs in Florida, where it can be very abundant along canals and rockpits that are intermittently bordered with shrubs and trees onto which it will climb. It is an insectivore that will also eat small vertebrates; however, its ecological impacts on the native vertebrate fauna in Florida are unknown.

19. *Calotes mystaceus*- The Indochinese Bloodsucker is an Old World species. Adults can reach about 38.1 cm TL. In the U.S., the Indochinese Bloodsucker occurs in Florida in habitat that has been disturbed. The Indochinese Bloodsucker is arboreal and an insectivore whose impacts on the native vertebrate fauna are unknown.

20. *Calotes versicolor*- The Variable Bloodsucker is an Old World species. Adults can reach about 43.5 cm TL. In the U.S., the Variable Bloodsucker occurs in Florida in habitat that has been disturbed. The Variable Bloodsucker is arboreal, consumes invertebrates and vertebrates, and could prove a threat to some segments of the native vertebrate fauna with which it occurs.

21. *Chameleo calyptratus*- The Veiled Chameleon is an Old World species. Adults can reach about 62 cm TL. In the U.S., the Veiled Chameleon occurs in Florida and Hawaii. In Florida, it is found in disturbed habitat. Highly arboreal, the Veiled Chameleon is primarily, but not exclusively, an insectivore and can eat small vertebrates, thereby increasing the likelihood of negatively impacting some segment of the native Florida vertebrate fauna.

22. *Chameleo jacksonii*- Jackson's Chameleon is an Old World species. Adults can reach about 15.0 cm SVL. In the U.S., Jackson's Chameleon occurs in Hawaii, where it is found in secondary forests and urban situations, but also in drier habitat. Its diet is comprised primarily of invertebrates. Its ecological impacts in Hawaii are unknown.

23. *Cnemidophorus lemniscatus*- The Rainbow Whiptail is a New World species. Adults of this all-female species can reach about 25.4 cm TL. In the U.S., the Rainbow Whiptail occurs in Florida, where it is found in relatively open, disturbed habitat. Its diet is comprised primarily of invertebrates. In Florida, the Rainbow Whiptail is ecologically similar to the native Six-lined Racerunner, with which it could be a potential competitor.

24. *Cosymbotus platyurus*- The Asian House Gecko is an Old World species. Adults can reach about 8.9 cm TL. In the U.S., the Asian House Gecko occurs in Florida, where it is restricted to manmade structures. Within a few years a colony of the Asian House Gecko was displaced by increasing numbers of the House Gecko (*Hemidactylus frenatus*) at the same site where the Indo-Pacific Gecko and Ringed Wall Gecko (*Tarentola annularis*) were also present. The Asian House Gecko is an insectivore. Its ecological impact in Florida is unknown and may remain so if eventually it is completely displaced in Florida.

25. *Cryptoblepharus poecilopleurus*- The Snake-eyed Skink is an Old World species. Adults can reach about 12.7 cm TL. In the U.S., the Snake-eyed Skink occurs in Hawaii, where it is very common in rocky, coastline habitat of small islands. However, it tends not to be found with other skink species. The Snake-eyed Skink is an insectivore. In Hawaii, the loss of beachfront habitat to development is thought to be responsible for its decline.

26. *Ctenosaura pectinata*- The Mexican Spinytail Iguana is a New World species. Adults can reach about 121.9 cm TL. In the U.S., the Mexican Spinytail Iguana occurs in Florida and Texas, where it is found in open, disturbed habitat. The Mexican Spinytail Iguana is primarily an herbivore whose ecological impacts in Florida are unknown.

27. *Ctenosaura similis*- The Black Spinytail Iguana is a New World species. Adults can reach about 121.9 cm TL. In the U.S., the Black Spinytail Iguana occurs in Florida, where it is found in open disturbed habitat. It is primarily an herbivore and has been observed defecating seeds of the highly invasive exotic Brazilian Pepper (*Schinus terebinthifolius*).

28. *Cyrtopodion scabrum*- The Roughtail Gecko is an Old World species. Adults can reach about 11.7 cm TL. In the U.S., the Roughtail Gecko occurs in Texas, where it is closely associated with buildings and other manmade structures. An insectivore, the Roughtail Gecko quickly replaces the Mediterranean Gecko; however, its ecological impacts on native vertebrates in Texas are unknown.

29. *Emoia cyanura*- The Coppertail Skink is an Old World species. Adults can reach about 13.3 cm TL. In the U.S., the Coppertail Skink occurs in Hawaii. Genetics is thought to hold the key to answering the question of whether or not this species is native to Hawaii. This insectivore is rare in Hawaii and is negatively impacted by the Cattle Egret (*Bubulcus ibis*) through predation.

30. *Emoia impar*- The Azuretail Skink is an Old World species. Adults can reach about 13.2 cm TL. In the U.S., the Azuretail Skink occurs in Hawaii, where it is found in dry, lowland areas and in mid-elevation moist forests. The Azuretail Skink is an insectivore. In Hawaii the species is negatively impacted by the Mongoose (*Herpestes auropunctatus*) and Indian Mynah (*Acridotheres tristis*) through predation. What role, if any, the Metallic Skink (*Lampropholis delicata*) has played in the decline of the Azuretail Skink has yet to be determined.

31. *Gehyra mutilata*- The Stump-toed Gecko is an Old World species. Adults can reach about 6.4 cm TL. In the U.S., the Stump-toed Gecko occurs in Hawaii, where it is found in uninhabited areas and in association with humans. The Stump-toed Gecko is an insectivore and in urban areas of Hawaii is displaced by the House Gecko (*Hemidactylus frenatus*).

32. *Gekko gecko*- The Tokay Gecko is an Old World species. Adults can reach about 36.0 cm TL. In the U.S., the Tokay Gecko occurs in Florida and Hawaii. In Florida, it lives on buildings and trees of disturbed habitat, including tropical hardwood hammock. In Hawaii, individuals are found on large trees, such as Ficus trees, and sometimes in houses, especially in attics. The Tokay Gecko eats invertebrates and vertebrates and is a threat to small, native vertebrates living in disturbed, mesophytic forests in Florida. In Hawaii, the Tokay Gecko is a predator of the Tree Gecko (*Hemiphyllodactylus typus*).

33. *Gonatodes albogularis*- The Yellowhead Gecko is a New World species. Adults can reach about 8.3 cm TL. In the U.S., the Yellowhead Gecko occurs in Florida, where it is found in disturbed habitat. This insectivore, once common, is now rare. It is unknown to what extent, if any, its demise is associated with the recent colonization of other exotic geckos.

34. *Hemidactylus frenatus*- The House Gecko is an Old World species. Adults can reach about 11.4 cm TL. In the U.S., the House Gecko occurs in Florida, Hawaii, and Texas. In Florida, it is found in vegetation and buildings of disturbed habitat. In Hawaii, it thrives around human dwellings, but is also is found in secondary forests. The House Gecko is one of several insectivorous *Hemidactylus* species that is incapable of stable coexistence with one another in Florida. For example, the House Gecko in Florida replaces the Indo-Pacific Gecko and the Asian House Gecko, and appears to be replaced by the Tropical House Gecko. In urban areas of Hawaii, the House Gecko negatively impacts the Indo-Pacific Gecko, Stump-toed Gecko, Tree Gecko, and Mourning Gecko (*Lepidodactylus lugubris*).

35. *Hemidactylus garnotii*- The Indo-Pacific Gecko is an Old World species. Adults of this all-female species can reach about 12.7 cm TL. In the U.S., the Indo-Pacific Gecko occurs in Florida, Georgia, Hawaii, and Texas. In Florida, the Indo-Pacific Gecko is found on vegetation and buildings of disturbed habitat. In Hawaii, it is found in urban areas and forest, and in Texas, it is found at a zoo. An insectivore, the Indo-Pacific Gecko is replaced by the House Gecko and the Tropical House Gecko, and in turn it replaces the Mediterranean Gecko in Florida. The diet of the Indo-Pacific Gecko is very similar to that of the Tropical House Gecko and overlaps with that of the Green Treefrog, and Squirrel Treefrog where they co-occur in southern Florida. The Cuban Treefrog is a predator of this species in Florida, and in urban areas of Hawaii, the Indo-Pacific Gecko is negatively affected by the House Gecko.

36. *Hemidactylus mabouia*- The Tropical House Gecko, a.k.a. “Wood Slave,” is an Old World species. Adults can reach about 12.7 cm TL. In the U.S., the Tropical

John Jensen

Indo-Pacific gecko (*Hemidactylus garnotii*)

House Gecko occurs in Florida. It is found on vegetation and buildings of disturbed habitat, where this insectivore quickly replaces the Indo-Pacific Gecko, the Mediterranean Gecko, and Ashy Gecko (*Sphaerodactylus elegans*). It appears to also replace the House Gecko. The diet of the Tropical House Gecko is very similar to that of the Indo-Pacific Gecko, and overlaps that of the Cuban Treefrog, Green Treefrog, and the Squirrel Treefrog where they co-occur in southern Florida. Its ability to prey on small geckos in Florida should be considered and may prove to be another mechanism of displacement of other species. In Florida, the Cuban Treefrog is a predator of the Tropical House Gecko.

37. *Hemidactylus turcicus*- The Mediterranean Gecko is an Old World species. Adults can reach about 12.7 cm TL. In the U.S., the Mediterranean Gecko occurs in Alabama, Arizona, Arkansas, California, Florida, Georgia, Illinois, Kansas, Louisiana, Maryland, Mississippi, Missouri, Nevada, New Mexico, Oklahoma, South Carolina, Texas, Utah, and Virginia. Across the U.S., the Mediterranean Gecko is strongly associated with dimly lit buildings, especially those with rough surfaces. The Mediterranean Gecko is an insectivore and in Florida it decreases the abundances of its nocturnal spider prey. In Florida the Mediterranean Gecko is replaced by the Indo-Pacific Gecko and Tropical House Gecko, and in southeastern Texas it is replaced by the Roughtail Gecko. Although localized, the House Gecko and Indo-Pacific Gecko are present in Texas, where they could interact with the Mediterranean Gecko. The ecology of the Mediterranean Gecko is becoming better known in Florida even as it steadily vanishes from the Florida landscape. It faces the same fate elsewhere if its thermal tolerances do not exceed those of its superior competitors in Florida and Texas.

38. *Hemiphyllodactylus typus*- The Tree Gecko is an Old World Gecko. Adults of this all-female species can reach about 8.9 cm TL. In the U.S., the Tree Gecko occurs in Hawaii, where it is found more often in forested areas than on buildings. It is an insectivore and is thought to be impacted by habitat alteration, competition, and potential predation by the House Gecko and the Tokay Gecko.

39. *Iguana iguana*- The Green Iguana is a New World species. Adults can reach about 201.0 cm TL. In the U.S., the Green Iguana occurs in Florida and Hawaii. In Florida, individuals can be abundant around vegetated canals and rockpits. The Green Iguana is primarily an herbivore, foraging in the trees and on the ground. Juveniles are eaten by the endangered Florida Burrowing Owl (*Athene cunicularia floridana*) in southern Florida; however, tracks of adults have also been observed entering these burrows, which through disturbance alone could have negative effects on the nests of this owl.

40. *Lacerta bilineata*- The Western Green Lacerta is an Old World species. Adults can reach about 30 cm TL. In the U.S., the Western Green Lacerta occurs in Kansas, where it is found in trees and on the ground of a city. It is primarily an insectivore but in Topeka, it also eats smaller lizards like the Italian Wall Lizard (*Podarcis sicula*).

41. *Lampropholis delicata*- The Metallic Skink is an Old World species. Adults can reach about 12.7 cm TL. In the U.S., the Metallic Skink occurs in Hawaii, where it is found in human-modified areas and ecotones between secondary forest and open habitat. The Metallic Skink is an insectivore. The role, if any, by the Metallic Skink in the decline of Azuretail Skink and Moth Skink (*Lipinia noctua*) in Hawaii remains to be determined.

42. *Leiocephalus carinatus*- The Northern Curlytail Lizard is a New World species. Adults can reach about 27.9 cm TL. In the U.S., the Northern Curlytail Lizard occurs in Florida where it is found in disturbed, open, rocky terrain, often in the form of sidewalks and parking lots. This species eats insects and lizards and poses a threat to sandy, upland species that even marginally exist along the coast.

43. *Leiocephalus schreibersii*- The Red-sided Curlytail Lizard is a New World species. Adults can reach about 25.4 cm TL. In the U.S., the Red-sided Curlytail Lizard occurs in Florida, where it inhabits disturbed sites. The Red-sided Curlytail Lizard is primarily an insectivore whose ecological impacts in Florida are unknown.

44. *Leiolepis belliana*- The Butterfly Lizard is an Old World species. Adults can reach about 30.0 cm TL. In the U.S., the Butterfly Lizard occurs in Florida, where it is found in disturbed, open, grassy areas. The Butterfly Lizard is an omnivore whose ecological impacts in Florida are unknown.

45. *Lepidodactylus lugubris*- The Mourning Gecko is an Old World species. Adults of this all-female species can reach about 9.5 cm TL. In the U.S., the Mourning Gecko occurs in Hawaii, where it is found in residential neighborhoods and uninhabited areas. This species is primarily, but not exclusively, insectivorous. In Hawaii, the Mourning Gecko is displaced by the House Gecko in urban areas and has been negatively affected by the Red-vented Bulbul (*Pyconotus cafer*) through predation.

46. *Lipinia noctua*- The Moth Skink is an Old World species. Adults can reach about 10.8 cm TL. In the U.S., the Moth Skink occurs in Hawaii, where it is found on rock walls, the leaf-litter of yards, and around and on the root systems of large trees. This species is an insectivore that is thought to be negatively affected by mammals and birds through predation. What role, if any, the Metallic Skink has played in the decline of the Moth Skink has yet to be determined.

47. *Mabuya multifasciata*- The Brown Mabuya is an Old World species. Adults can reach about 13.0 cm TL. In the U.S. the Brown Mabuya occurs in Florida, where it is found in lush but disturbed habitat. The Brown Mabuya is an omnivore and consequently a potential threat to some of the south Florida vertebrate fauna.

48. *Pachydactylus bibronii*- Bibron's Thick-toed Gecko is an Old World species. Adults can reach about 14.0 cm TL. In the U.S., Bibron's Thick-toed Gecko occurs in Florida, where it is found on buildings. Bibron's Thick-toed Gecko is an insectivore whose ecological impacts in Florida are unknown.

49. *Phelsuma guimbeaui*- The Orange-spotted Day Gecko is an Old World species. Adults can reach about 17.8 cm TL. In the U.S., the Orange-spotted Day Gecko occurs in Hawaii where it is found in vegetation of suburban neighborhoods. The Orange-spotted Day Gecko is primarily insectivorous and its ecological impacts in Hawaii are unknown.

50. *Phelsuma laticauda*- The Gold Dust Day Gecko is an Old World species. Adults can reach about 14.0 cm TL. In the U.S., the Gold Dust Day Gecko occurs in Hawaii, where it is found in and around human dwellings. The Gold Dust Day Gecko is primarily an insectivore that negatively impacts exotic, injurious invertebrates in Hawaii.

51. *Phelsuma madagascariensis*- The Madagascar Day Gecko is an Old World species. Adults can reach about 27.9 cm TL. In the U.S., the Madagascar Day Gecko occurs in Florida and Hawaii and is primarily an insectivore. In Florida, it is found in disturbed habitat. The ecological impacts of this species are unknown.

52. *Podarcis muralis*- The Common Wall Lizard is an Old World species. Adults can reach about 20.5 cm TL. In the U.S., the Common Wall Lizard occurs in Indiana, Kentucky, and Ohio, where it is found in urban settings. The Common Wall Lizard is primarily an insectivore whose ecological impacts in the three states are unknown.

53. *Podarcis sicula*- The Italian Wall Lizard is an Old World species. Adults reach about 24 cm TL. In the U.S., the Italian Wall Lizard occurs in Kansas and New York. The Italian Wall Lizard is found on debris piles and nearby vegetation of urban areas, where it feeds on invertebrates. Sufficiently deep hibernacula are a limiting factor in their northward expansion. Its ecological impacts in Kansas and New York are unknown.

54. *Sphaerodactylus argus*- The Ocellated Gecko is a New World species. Adults can reach 6.4 cm TL. In the U.S., the Ocellated Gecko occurs in Florida, where it is found in disturbed habitat. The Ocellated Gecko is an insectivore whose ecological impacts in Florida are unknown.

55. *Sphaerodactylus elegans*- The Ashy Gecko is a New World species. Adults can reach about 7.0 cm TL. In the U.S., the Ashy Gecko occurs in Florida, where it is found on buildings and in vegetation of disturbed habitats. The Ashy Gecko is an insectivore that is rapidly replaced by the Tropical House Gecko. Its ecological impacts in Florida are unknown.

56. *Tarentola annularis*- The Ringed Wall Gecko is an Old World species. Adults can reach about 15.2 cm TL. In the U.S., the Ringed Wall Gecko occurs in Florida, where it is found on buildings in disturbed areas. The Ringed Wall Gecko is primarily an insectivore but also eats other geckos. Consequently, although the ecological impacts of the Ringed Wall Gecko in Florida are unknown, it may pose a predatory threat to small, native, nocturnal vertebrates in Florida.

57. *Tupinambis merianae*- The Argentine Giant Tegu is a New World species. Adults can reach about 132.0 cm TL. In the U.S., the Argentine Black and White Tegu occurs in Florida, where it is found in oak scrub and reclaimed phosphate lands. The Argentine Black and White Tegu is primarily a frugivore as an adult and individuals occupy burrows of the Gopher Tortoise (*Gopherus polyphemus*).

58. *Varanus niloticus*- The Nile Monitor is an Old World species. Adults reach about 243.0 cm TL. In the U.S., the Nile Monitor occurs in Florida, where it is found primarily, but apparently not exclusively, in disturbed habitat near water. The Nile Monitor is thought also to

inhabit mangrove islands. The Nile Monitor is a large carnivore that can chase and excavate vertebrate prey and consequently poses a severe threat to a wide range of vertebrates, including threatened and endangered species.

Snakes

59. *Python molurus*- The Indian Python is an Old World species. Adults can reach about 777.2 cm TL. In the U.S., the Indian Python occurs in Florida, where it is found in wetland and upland habitats of the Everglades as well as in disturbed habitat. Large clutches of eggs are tended by the female, increasing the likelihood of hatchling survival. This large constrictor can eat and is eaten by the American Alligator (*Alligator mississippiensis*) and has the potential to become the top predator of the southern Everglades system. Presumably, it can eat or be eaten by the Spectacled Caiman (*Caiman crocodilus*). This species poses a threat to a wide range of vertebrates, including humans.

60. *Ramphotyphlops braminus*- The Brahminy Blind Snake is an Old World species. Adults of this all-female species can reach about 17.3 cm TL. In the U.S., the Brahminy Blind Snake occurs in Georgia, Florida, Hawaii, Louisiana, Massachusetts, Texas, and Virginia. In Florida, the Brahminy Blind Snake appears to be restricted to mesic, disturbed systems. In Hawaii, individuals are found in gardens and in moist valleys. In Louisiana, Massachusetts, and Virginia, individuals are associated with human disturbance, and in the latter two states, colonies are associated with buildings. Fossorial in habits and an insectivore, its ecological impacts on ecologically similar species remain unknown.

John Jensen

Brahminy blind snake (*Ramphotyphlops braminus*)

Turtles

61. *Palea steindachneri*- The Wattleneck Softshell is an Old World species. Adults can reach about 41 cm CL. In the U.S., the Wattleneck Softshell occurs in Hawaii. Strongly aquatic in habits, this species is found in a wide range of aquatic systems. The Wattleneck Softshell is opportunistic and primarily carnivorous. In Hawaii, the Wattleneck Softshell is more abundant than the Chinese Softshell (*Pelodiscus sinensis*).

62. *Pelodiscus sinensis*- The Chinese Softshell is an Old World species. Adults can reach about 25 cm CL. In the U.S., the Chinese Softshell occurs in Hawaii. Strongly aquatic in habits, this species is found in a wide range of aquatic systems. The Chinese Softshell is primarily carnivorous. In Hawaii, the Chinese Softshell is less abundant than the Wattleneck Softshell.

Crocodilians

63. *Caiman crocodilus*- The Spectacled Caiman is a New World species. Adults can reach about 182.9 cm TL. In the U.S., the Spectacled Caiman occurs in Florida, where it appears to be restricted to canals and rockpits. As a potential competitor of the native American Alligator, the Spectacled Caiman is a potential problem even if thus far restricted in introduced habitat and geographic range. This species is presumably capable of eating and being eaten by the Indian Python.

Postscript- Snow et al. (2007) confirm the establishment of the Boa Constrictor (*Boa constrictor*) in Florida. Also, The Chinese Softshell (*Pelodiscus sinensis*) has recently been discovered in the Potomac River, Virginia (Mitchell et al. 2007).

John White

The Chinese Softshell *(Pelodiscus sinensis)*

HABITATS IN THE UNITED STATES

Kurt A. Buhlmann

Partners in Amphibian and Reptile Conservation (PARC) is a habitat-focused conservation organization. In the design of inventory projects for amphibians and reptiles, resource managers must become familiar with the habitats in which target species are most likely to occur. As companion documents to this Inventory and Monitoring Techniques Manual (I&M), PARC has published five regional Habitat Management Guidelines for Amphibians and Reptiles (HMGs). Within each guide, the major habitats of each region (Southeast, Northeast, Midwest, Northwest, Southwest) are described. Appendices in the back of each guide associate each species with its habitats of occurrence and assign a ranking of either optimal, suitable, or marginal (O-S-M system) to every habitat. For each species, its political state of occurrence, as well as its current rarity (Natural Heritage rank) and protected status, is given. The species lists in the Habitat Management Guidelines are consistent with the species list in the Species x Techniques Table of this book (Table 5.1).

Habitats and their condition are also likely targets for monitoring. In fact, monitoring the condition of the habitats of occurrence for target species goes hand in hand with monitoring the populations of the species within them. The goals of the Habitat Management Guidelines are to suggest management strategies that will benefit, or at least minimize impacts to, habitats used by amphibians and reptiles. Therefore, monitoring the effects of habitat management activities will be an important part of PARC's overall amphibian and reptile conservation strategy.

Valorie Titus

Wetland in Northeast United States.

Jamie Bettaso

The Trinity River in California is one example of habitat in Northwest region.

REGIONAL RESOURCES ON THE AMPHIBIANS AND REPTILES OF THE UNITED STATES

Southeast

Ashton, R. E., Jr., and P. S. Ashton. 1981. Handbook of Reptiles and Amphibians of Florida. Part I: The Snakes. Windward Publishing, Miami, FL.

Ashton, R. E., Jr., and P. S. Ashton. 1988. Handbook of Reptiles and Amphibians of Florida. Part III. The Amphibians. Windward Publishing Co., Miami, FL.

Bailey, M. A., J. N. Holmes, K. A. Buhlmann, and J. C. Mitchell. 2006. Habitat Management Guidelines for Amphibians and Reptiles of the Southeastern United States. Partners in Amphibian and Reptile Conservation Technical Publication HMG-2, Montgomery, Alabama. 88pp.

Bartlett, R. D., and P. Bartlett. 2003. Florida's Snakes: A Guide to Their Identification and Habits. University of Florida Press, Gainesville, FL.

Beane, J. C., A. L. Braswell, J. C. Mitchell, W. M. Palmer, and J. R. Harrison III. 2010. Amphibians and Reptiles of the Carolinas and Virginia. University of North Carolina Press, Chapel Hill.

Bjorndal, K. A., Ed. 1981. Biology and Conservation of Sea Turtles. Smithsonian Institution Press, Washington, D.C.

Boundy, J. 1997. Snakes of Louisiana. Louisiana Department of Wildlife and Fisheries, Baton Rouge, LA.

Buhlmann, K. A., T. D. Tuberville, and J. W. Gibbons. 2008. Turtles of the Southeast. University of Georgia Press. Athens, GA.

Carmichael, P., and W. Williams. 2001. Florida's Fabulous Reptiles and Amphibians. World Publications, Tampa, FL.

Dorcas, M. E. 2004. A Guide to the Snakes of North Carolina. Davidson College Herpetology Laboratory, Davidson, NC.

Dorcas, M. E., and J. W. Gibbons. 2008. Frogs and Toads of the Southeast. University of Georgia Press. Athens, GA.

Dorcas, M. E., and J. W. Gibbons. 2011. Frogs: The Animal Answer Guide. Johns Hopkins University Press, Baltimore.

Dundee, H. A., and D. A. Rossman. 1989. The Amphibians and Reptiles of Louisiana. Louisiana State University Press, Baton Rouge.

Gibbons, J. W., and M. E. Dorcas. 2004 . North American Watersnakes: A Natural History. University of Oklahoma Press, Norman, OK.

Gibbons, J. W., and M. E. Dorcas. 2005. Snakes of the Southeast. University of Georgia Press. Athens, GA.

Gibbons, J. W., and J. L. Greene. 2009. Turtles: The Animal Answer Guide. Johns Hopkins University Press, Baltimore.

Gibbons, J. W., J. L. Greene and T. Mills. 2009. Lizards and Crocodilians of the Southeast. University of Georgia Press. Athens, GA.

Gregoire, D. R. 2005. Tadpoles of the Southeastern United States Coastal Plain. United States Geological Survey Report. Florida Integrated Science Center: 60.

Jensen, J. B., C. D. Camp, J. W. Gibbons, and M. J. Elliott. 2008. Amphibians and Reptiles of Georgia. University of Georgia Press, Athens, GA.

Lohoefener, R., and R. Altig. 1983. Mississippi Herpetology. Mississippi State University Research Center, NSTL Station, MS.

Meshaka, W. E., Jr., B. P. Butterfield, and J. B. Hauge. 2004. The Exotic Amphibians and Reptiles of Florida. Krieger Publishing Co., Malabar, FL.

Mitchell, J., and J. W. Gibbons. 2010. Salamanders of the Southeast. University of Georgia Press. Athens, GA.

Mount, R. H. 1975. The Reptiles and Amphibians of Alabama. Auburn University Agricultural Experiment Station, Auburn, AL.

Palmer, W. M., and A. L. Braswell. 1995. Reptiles of North Carolina. University of North Carolina Press, Chapel Hill.

Redmond, W. H., and A. F. Scott. 1996. Atlas of Amphibians in Tennessee. Austin Peay State University, Clarksville, TN.

Ruckdeschel, C., and C. R. Shoop. 2006. Sea Turtles of the Atlantic and Gulf Coasts of the United States. The University of Georgia Press, Athens, GA.

Sinclair, R. M., Hon, W., & Ferguson, R. B., 1970. Amphibians and reptiles in Tennessee. rev.ed. State Game and Fish Commission, Nashville. 30 pp.

Wright, A. H. 2002. Life-Histories of the Frogs of Okefenokee Swamp, Georgia - North American Salientia (Anura) No. 2. Comstock Publishing Associates, Cornell University Press, Ithaca and London.

Northeast

Degraaf, R. M. 1983. Amphibians and Reptiles of New England: Habitats and Natural History. University of Massachusetts Press, 1st Edition. 96pp.

Eliot, L. 2004. The Calls of Frogs and Toads. Stackpole Books, Mechanicsburg, Pennsylvania.

Gibbs, J. P., A. R. Breisch, P. K. Ducey, G. Johnson, the late J. Behler, and R. Bothner. 2007. The Amphibians and Reptiles of New York State: Identification, Natural History, and Conservation. Oxford University Press, USA, 504pp.

Green, N. B., and T. K. Pauley. 1987. Amphibians and Reptiles in West Virginia. University of Pittsburgh Press. 284pp.

Hulse, A. C., C. J. McCoy, and E. Censky. 2001. Amphibians and Reptiles of Pennsylvania and the Northeast, Comstock Publishing Associates, Cornell University.

Hunter, M. L., A. J. K. Calhoun, and M. McCollough. 1999. Maine Amphibians and Reptiles. University of Maine Press. 252pp.

Klemens, M. K. 1993. Amphibians and Reptiles of Connecticut and Adjacent Regions. State Geological and Natural History Survey of Connecticut, Bulletin No. 112. 318 pp.

Krulikowski, L. 2007. Snakes of New England: a Photographic and Natural History Study. Self-published. 320 pp.

Mitchell, J. C. 1994. The Reptiles of Virginia. Smithsonian Institution Press, Washington, D.C.

Mitchell, J. C., and K. K. Reay. 1999. Atlas of Amphibians and Reptiles in Virginia. Virginia Department of Game and Inland Fisheries, Richmond, VA.

Pinder, M. J., and J. C. Mitchell. 2001. A Guide to the Snakes of Virginia. Wildlife Diversity Special Publication 2. Virginia Department of Game and Inland Fisheries, Richmond, VA.

Tyning, T. F. 1990. A Guide to Amphibians and Reptiles. Little, Brown and Company, Boston, Massachusetts. 400 pp.

Midwest

Breckenridge, W. J. 1958. Reptiles and Amphibians. The University of Minnesota Press, Minneapolis, MN.

Casper, G. S. 1996. Geographic Distributions of the Amphibians and Reptiles of Wisconsin. Milwaukee Publ. Museum, Inc., Milwaukee, WI.

Christiansen, J. L., and R. M. Bailey. 1989. The Snakes of Iowa. Iowa Conservation Commission. Des Moines, IA.

Collins, J. T., S. L. Collins, and T. W. Taggart. 2010. Amphibians, Reptiles, and Turtles in Kansas. Eagle Mountain Publishing. 312pp.

Fogell, D. 2010. A Field Guide to the Amphibians and Reptiles of Nebraska. Conservation & Survey Division, School of Natural Resources.

Harding, J. H. 1997. Amphibians and Reptiles of the Great Lakes Region. The University of Michigan Press, Ann Arbor, Michigan. 378 pp.

Harding, J. H., and J. A. Holman. 1990. Michigan Turtles and Lizards. Michigan State University Museum.

Holman, J. A., J. H. Harding, M. M. Hensley, and G. R. Dudderar. 1998. Michigan Snakes: A Field Guide and Pocket Reference. Michigan State University Cooperative Extension Service, East Lansing.

Johnson, T. R. 2000. Amphibians and Reptiles of Missouri. Missouri Department of Conservation, Jefferson City, MO.

Johnson, T. R. 2000. Amphibians and Reptiles of Missouri. Missouri Department of Conservation. 400pp.

MacGowan, B. J., B. A. Kingsbury, and R. N. Williams. 2005. Turtles of Indiana. Purdue University, West Lafayette, IN.

Minton, S. A., Jr. 2001. Amphibians and Reptiles of Indiana. Academy of Science, Indianapolis, Indiana.

Moriarty, J. J., and D. Jones, Eds. 1997. Minnesota's Amphibians and Reptiles - Their Conservation and Status. Serpent's Tale Natural History Book Distributors, Lanesboro, MN.

Phillips, C. A., R. A. Brandon, and E. O. Moll. 1999. Field Guide to Amphibians and Reptiles of Illinois. Illinois Natural History Survey, Champaign, IL.

VanDeWalle, T., and S. L. Collins. 2010. Snakes and Lizards in Your Pocket: A Guide to Reptiles of the Upper Midwest. University Of Iowa Press.

Southwest

Bartlett, R. D., and P. Bartlett. 2009. Guide and Reference to the Snakes of Western North America (North of Mexico) and Hawaii. University Press of Florida. 304pp.

Bartlett, R. D., and P. Bartlett. 2009. Guide and Reference to the Amphibians of Western North America (North of Mexico) and Hawaii. University Press of Florida. 288pp.

Bartlett, R. D., and P. Bartlett. 2009. Guide and Reference to the Turtles and Lizards of Western North America (North of Mexico) and Hawaii University Press of Florida. 322pp.

Berry, K. H. 1986. Desert tortoise (*Gopherus agassizii*) research in California, 1976–1985. Herpetologica 42:62–67.

Brennan, T. C., and A. T. Holycross. 2006. A Field Guide to Amphibians and Reptiles in Arizona. Arizona Game and Fish, 150pp.

Burkett, D. W. 2006. Amphibians and Reptiles of White Sands Missile Range - Field Guide 2006. White Sands Technical Services, LLC., White Sands Missile Range, NM.

Jennings, M. R., and M. P. Hayes. 1994. Amphibian and reptile species of special concern in California. California Department of Fish and Game Final Report. 255 pp.

Jones, L. L. C., and R. E. Lovich. 2009. Lizards of the American Southwest: A Photographic Field Guide. Rio Nuevo Publishers. 568pp.

Lowe, C. H., C. R. Schwalbe, and T. B. Johnson. 1986. The Venomous Reptiles of Arizona. Arizona Game and Fish Department.

Tennant, A. 1984. The Snakes of Texas. Texas Monthly Press.

Van Devender, T. R. (ed.). 2002. The Sonoran Desert Tortoise: Natural History, Biology and Conservation. University of Arizona Press. 388pp.

Werler, J. E., and J. R. Dixon. 2000. Texas Snakes - Identification, Distribution, and Natural History. University of Texas Press, Austin, TX.

Williamson, M. A., P. W. Hyder, and J. S. Applegarth. 1994. Snakes, Lizards, Turtles, Frogs, Toads and Salamanders of New Mexico. Sunstone Press, Santa Fe, NM.

Young, M. T., L. J. Livo, and S. Wilcox. 2011. The Guide to Colorado Reptiles and Amphibians. Fulcrum Publishing. 176pp.

Northwest

Bury, R. B. 1994. Vertebrates in the Pacific Northwest: Species richness, endemism and dependency on old-growth forests. In: S. K. Majumdar, D. J. Brenner, J. E. Lovich, and J. F. Schalles (eds.) Biological Diversity: Problems and Challenges. Pennsylvania Academy of Science, Easton, PA. p392-404.

Clayton, D. R., and D. H. Olson. 2009. Conservation assessment for the Oregon Slender Salamander (*Batrachoseps wrighti*). USDA Forest Service Region 6 and USDI Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml

Corkran, C. C., and C. Thoms. 2006. Amphibians of Oregon, Washington, and British Columbia: A Field Identification Guide. Revised edition. Lone Pine Publishing. Edmonton, Alberta. 176 pp.

Crisafulli, C. M., D. R. Clayton, and D. H. Olson. 2008. Conservation assessment for the Larch Mountain Salamander (*Plethodon larselli*). USDA Forest Service Region 6 and USDI Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml

Cushman, K. A., and C. A. Pearl. 2007. A conservation assessment for the Oregon Spotted Frog (*Rana pretiosa*).USDA Forest Service Region 6 and USDI Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml

Davis, R., and D. Olson. 2007. Field validation of a landscape habitat model for the foothill yellow-legged frog (*Rana boylii*) in Oregon. USDA Forest Service Region 6 and USDI Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/documents/inventories/inv-rpt-ha-rabo-hab-model-field-valid-2008.pdf

Fichter, E., and A. D. Linder. 1964. The Amphibians of Idaho. The Idaho State University Museum, Pocatello, ID.

Howell, B. L., and C. R. Roberts. 2008 A conservation assessment for the Olympic torrent salamander (*Rhyacotriton olympicus*). USDA Forest Service Region 6 and USDI Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml

Jones, L. L. C., W. P Leonard, and D. H. Olson (eds.). 2005. Amphibians of the Pacific Northwest. Seattle Audubon Society. Seattle, WA. 227 pp.

Jones, L., W. P. Leonard and Olson, D. H. 2005. Amphibians of the Pacific Northwest. Seattle Audubon Society, Seattle, WA.

Myers, C. H., L. Eigner, J. A. Harris, R. Hilman, M. D. Johnson, R. Kalinowski, J. J. Muir, M. Reyes, and L. E. Tucci. 2007. A comparison of ground-based and tree-based polyvinyl chloride pipe refugia for capturing *Pseudacris regilla* in northwestern California. Northwestern Naturalist 88:147-154.

Nauman, R. S., and D. H. Olson. 2004. Surveys for terrestrial amphibians in Shasta County, California with notes on the distribution of Shasta salamanders (*Hydromantes shastae*). Northwestern Naturalist 85:29-32.

Olson, D. H. (ed.) 1999. Survey protocols for amphibians under the Survey and Manage provision of the Northwest Forest Plan. Version 3.0. Interagency publication of the Regional Ecosystem Office, Portland, OR. Bureau of Land Management Publication BLM/OR/WA/PT-00/033+1792; U.S. Government Printing Office: 2000--589-124/04022 Region No. 10. 310 p.

Olson, D. H. 2008. Conservation assessment for the Black Salamander (*Aneides flavipunctatus*) in Oregon. USDA Forest Service Region 6 and USDI Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml

Olson, D. H. 2008. Conservation assessment for the California Slender Salamander (*Batrachoseps attenuatus*) in Oregon. USDA Forest Service Region 6 and USDI Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml

Olson, D. H., and G. Weaver. 2007. Vertebrate assemblages associated with headwater hydrology in western Oregon managed forests. Forest Science 53(2):343-355.

Olson, D. H., and R. Davis. 2007. Conservation assessment for the Foothill Yellow-Legged Frog (*Rana boylii*) in Oregon. USDA Forest Service Region 6 and USDI Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml

Olson, D. H., D. Clayton, E. C. Reilly, R. S. Nauman, B. Devlin, and H. H. Welsh, Jr. 2007. Conservation strategy for the Siskiyou Mountains Salamander (*Plethodon stormi*), northern portion of the range. USDA Forest Service Region 6 and USDI Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml

Olson, D. H., W. H. Leonard, and R. B. Bury (eds). 1997. Sampling amphibians in lentic habitats. Northwest Fauna 4:1-134.

Richter, K. O. 1995. A simple aquatic funnel trap and its application to wetland amphibian monitoring. Herpetological Review 26:90-9 1.

St. John, A. S. 2002. Reptiles of the Northwest: British Columbia to California. Lone Pine Publishing. Edmonton, Alberta. 272 pp.

Storm, R. M., W. P. Leonard, H. A. Brown, R. B. Bury, D. M. Darda, L. V. Diller, and C. R. Peterson. 1995. Reptiles of Washington and Oregon. Seattle Audubon Society, Washington. 176 pp.

Tait, C. 2007. Conservation assessment of the Great Basin Population of the Columbia Spotted Frog (*Rana luteiventris*). USDA Forest Service Region 6 and USDI Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml

Thoms, C., C. C. Corkran, and D. H. Olson. 1997. Basic amphibian survey for inventory and monitoring in lentic habitats. In D. H. Olson, W. P. Leonard, and R. B. Bury, eds. Sampling Amphibians in Lentic Habitats: Methods and Approaches for the Pacific Northwest. Society for NW Vertebrate Biology. Olympia, WA. Northwest Fauna 4:35-46.

Wheeler, G. C., and J. Wheeler. 1966. The Amphibians and Reptiles of North Dakota. The University of North Dakota Press, Grand Forks, ND.

Multi-Regional, National, and International Resources

Behler, J. L., and F. W. King. 1979. The Audubon Society Field Guide to North American Reptiles and Amphibians. Alfred A. Knopf, New York, NY.

Bonin, F., B. Devaux, and A. Dupre. 2006. Turtles of the World. The Johns Hopkins University Press, Baltimore, MD.

Bury, R. B. (Ed.). 1982. North American Tortoises: Conservation and Ecology. U.S. Fish and Wildlife Service, Wildlife Research Report 12. 126 pp.

Bury, R. B., and D. Germano (eds.). 1994. Biology of North American Tortoises. USDI, Fish and Wildlife Research 13. 204 pp

Campbell, J. A., and W. W. Lamar. 2004. The Venomous Reptiles of the Western Hemisphere. 2 vols. Comstock Books in Herpetology. Cornell University Press, Ithaca, NY.

Carr, A. 1952. Handbook of Turtles - The Turtles of the United States, Canada, and Baja California. Cornell University Press, Ithaca, NY.

Conant, R., and J. T. Collins. 1998. A Field Guide to Reptiles and Amphibians of Eastern and Central North America. Third expanded edition. Houghton Mifflin, Boston.

Dodd, C. K., Jr. 2001. North American Box Turtles - A Natural History. University of Oklahoma Press, Norman, OK.

Ernst, C. H. 1992. Venomous Reptiles of North America. Smithsonian Institution Press, Washington and London.

Ernst, C. H., and E. M. Ernst. 2003. Snakes of United States and Canada. Smithsonian Books, Washington, D.C.

Ernst, C. H., J. Lovich, and R. W. Barbour. 1994. Turtles of the United States and Canada. Smithsonian Institution Press, Washington and London.

Gibbons, J. W., and M. E. Dorcas. 2004. North American Watersnakes: A Natural History. University of Oklahoma Press, Norman.

Greene, H. W. 1997. Snakes: The Evolution of Mystery in Nature. University of California Press, Berkeley.

Kendell, K. 2002. Survey protocol for the northern leopard frog. Alberta Sustainable Resource Development, Fish and Wildlife Division, Alberta Species at Risk Report No. 43, Edmonton, AB. 30 pp.

Matsuda, B. M., D. M. Green, and P. T. Gregory. Amphibians and Reptiles of British Columbia. Royal BC Museum. Victoria, BC. 266 p.

Petranka, J. W. 1998. Salamanders of the United States and Canada. Smithsonian Institution Press, Washington and London.

Pretzlaw, T., M. Huynh, L. Takats, and L. Wilkinson. 2002. Protocol for monitoring long-toed salamander (*Ambystoma macrodactylum*) populations in Alberta. Alberta Sustainable Resource Development, Fish and Wildlife Division, Alberta Species at Risk Report No. 35. Edmonton, Alberta. 17 pp

Resources Information Standards Committee (RISC). 1998a. Inventory Methods for Pond-breeding Amphibians and Painted Turtle Standards for Components of British Columbia's Biodiversity No. 37. Prepared by Ministry of Environment, Lands and Parks Resources Inventory Branch for the Terrestrial Ecosystems Task Force Resources Inventory Committee, March 13, 1998. Version: 2.0. http://www.ilmb.gov.bc.ca/risc/pubs/tebiodiv/pond/assets/pond.pdf.

Resources Information Standards Committee (RISC). 1998b. Inventory Methods for Snakes Standards for Components of British Columbia's Biodiversity No. 38 Prepared by Ministry of Environment, Lands and Parks Resources Inventory Branch for the Terrestrial Ecosystems Task Force Resources Inventory Committee, March 12, 1998. Version 2.0 http://www.ilmb.gov.bc.ca/risc/pubs/tebiodiv/snakes/assets/snake.pdf.

Resources Information Standards Committee (RISC). 1999. Inventory Methods for Plethodontid Salamanders Standards for Components of British Columbia's Biodiversity No. 36. Prepared for: Ministry of Environment, Lands and Parks Resources Inventory Branch for the Terrestrial Ecosystems Task Force Resources Inventory Committee, March 1, 1999. Version 2.0 http://www.ilmb.gov.bc.ca/risc/pubs/tebiodiv/salamanders/assets/psalm.pdf.

Resources Information Standards Committee (RISC). 2000. Inventory Methods for Tailed Frog and Pacific Giant Salamander Standards for Components of British Columbia's Biodiversity No. 39 Prepared by Ministry of Environment, Lands and Parks Resources Inventory Branch for the Terrestrial Ecosystems Task Force Resources Inventory Committee, March 13, 2000. Version 2 http://www.ilmb.gov.bc.ca/risc/pubs/tebiodiv/frog/assets/tailedfrog.pdf

Rossman, D. A., N. B. Ford, and R. A. Seigel. 1996. The Garter Snakes: Evolution and Ecology. Animal Natural History Series, vol. 2. University of Oklahoma Press, Norman.

Rubio, M. 2010. Rattlesnakes of the United States and Canada. ECO Press. 307pp.

Smith, H. M. 1978. Amphibians of North America: A guide to field identification. Golden Press, New York. 160 pp.

Smith, H. M., and E. D. Brodie, Jr. 1982. Reptiles of North America: A guide to field identification. Golden Press, New York. 240 pp.

Stebbins, R. C. 1985. A Field Guide to Western Reptiles and Amphibians. Houghton Mifflin Company, Boston, MA.

Tennant, A., and R. D. Bartlett. 2000. Snakes of North America: Eastern and Central Regions. Gulf Publishing Company, Houston, Texas.

Tennant, A. 2003. Snakes of North America - Eastern and Central Regions. Lone Star Books, Lanham, MD.

Wright, A. H., and A. A. Wright. 1957. Handbook of Snakes of the United States and Canada - Volume I. Comstock Publishing Associates, Ithaca and London.

Wright, A. H., and A. A. Wright 1979. Handbook of Snakes of the United States and Canada, Volume III, Bibliography. Society for the Study of Amphibians and Reptiles. A. H. Wright and A. A. Wright, Ithaca, NY.

Zim, H. S., and H. M. Smith. 2001. Reptiles and Amphibians. A Golden Guide. St. Martins Press, New York, NY.

Spotted salamander (*Amybstoma maculatum*)

Michael Marchand

CHAPTER 3: ESSENTIALS OF SAMPLING DESIGN

Christopher T. Winne

This manual outlines and describes techniques for detecting reptiles and amphibians (see Chapter 5). To be valuable as a conservation tool, these techniques must be implemented using an appropriate sampling design. In essence, sampling design is the temporal and spatial structure of the data collection process. Sampling designs vary based on the goals of the project. They also determine the types of analyses and levels of interpretation that can be generated from collected data. Therefore, a clear understanding of the inventory and/or monitoring goals of any program is a prerequisite to the design of an appropriate sampling scheme. This chapter is designed as a guide to help you develop a sampling scheme that will achieve the goals of your inventory or monitoring program.

SCOPE

Sampling theory and experimental design are vast topics and have been the subject of numerous books and published articles. Moreover, new sampling designs and statistical techniques for population and community analyses are developed each year. Consequently, this chapter should be viewed as a digest of some of the essential points that should be considered when developing a sampling design, not as an encyclopedic source on sampling design. Planning a sound sampling design is one of the keys to a successful project. Therefore, project planners are encouraged to take advantage of the multitude of resources available on the subject and, when necessary, to contact researchers that have prior experience with sampling schemes for achieving specific inventory and monitoring goals.

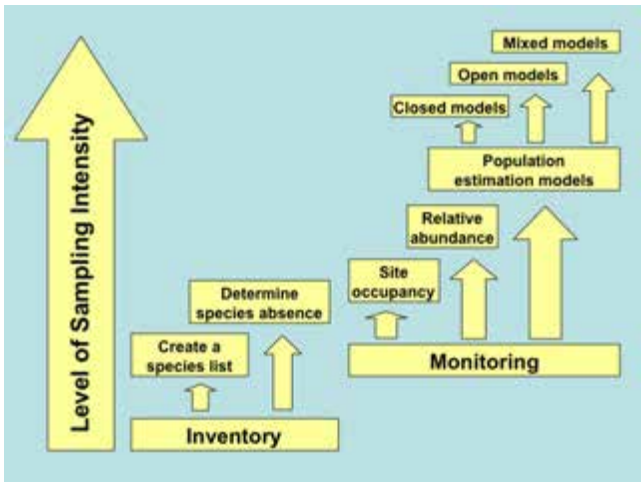
BASIC CONCEPTS

Determining the exact motivation for an inventory or monitoring program from the outset can help in determining program goals and subsequent design. Once the motivation for starting a program has been clearly identified, you can begin to define and prioritize the program goals.

Defining project goals

The first step to a successful research project is to define the goals. Two broad categories of conservation-based research goals are distinguished throughout this chapter and the remainder of the manual: inventory and monitoring. In general, the goal of an inventory project is to create a list of species known to occur in a focal area or habitat, although an inventory could be used to determine the distribution of a single species. In contrast, the purpose of monitoring is to evaluate the status and trends of populations in

an area. Monitoring usually involves assessing population size and comparing abundance or population health over time. Several levels of sampling intensity are available for surveying and monitoring populations of reptiles and amphibians (Fig. 3-1). This chapter distinguishes between two levels of inventories (creating species lists versus determining species absence) and three general metrics of abundance that can be used to monitor population trends over time: site occupancy, relative abundance, and estimated population size (Fig. 3-1).



Chris Winne

Figure 3-1. Hierarchical ranking of the sampling intensity required for each of the inventory and monitoring sampling designs described in this chapter.

Overarching objectives may include determining species presence/absence, occupancy, distribution, abundance estimates, habitat relationships, and population trends over time. For example, a biologist working for a timber company may need to know if they have any endangered amphibian or reptile species on their property. In this case, a thorough inventory would be appropriate. If the land manager of a state park is asked to assess herpetofaunal population trends over time in response to changing habitat management plans, a monitoring program will need to be designed and initiated.

Hitting the target

The target population is the population that a researcher wants to draw inferences about and is equivalent to the concept of a "sampling frame". A target population could be a biological population or it could be defined by a political boundary (e.g., county, state, or nature preserve) or habitat type. To make inferences about the target population, however, the target population will have to be sampled, perhaps by using traps or visual searches. The portion of the target population that is available for capture or observation at any time is the sample population and can be used to estimate parameters (e.g., population size, survival rate) for the sample population. The sample population must be an unbiased, representative sample of the target population, or the sampling biases must be known, to be able to draw inferences about the target population.

Whit Gibbons

Defining your target life stage is important. Juvenile and adult snapping turtles require different sampling techniques.

For example, if a researcher wanted to draw inferences about (e.g., estimate) the population size of a snake species within an isolated wetland, then the target population would be the entire population of that snake species within the isolated wetland. To make any inferences, however, the researcher will have to sample the snake population, perhaps by conducting visual searchers or by setting aquatic funnel traps along the shallow margin of the wetland. In this scenario there are several non-obvious ways for the sample population to be a biased sample of the target population. First, certain demographics of the population may be temporarily unavailable for capture, unlikely to be detected, or completely untrappable because of body size, reproductive condition, or activity levels, among other reasons. For example, neonate snakes may be less likely to be captured or observed than adults due to their small body size. Second, traps set (or observers) along the margin of the wetland may not adequately sample snakes that are located within the center of the wetland during the sampling period. Individuals that are not detectable are considered "unavailable for capture" and the sample population will only consist of the detectable or trappable portion of the population.

Issues with availability for capture can occur with other reptiles and amphibians. For example, certain demographics of a population may undergo seasonal migrations between habitats (e.g., adult pond-breeding amphibians or semi-aquatic turtles) and, therefore, they will be temporarily unavailable for capture in some habitats during certain times of the year.

Also, many reptiles hibernate below ground during winter months but are active and detectable during summer months. Consequently, it is important to consider the temporal characteristics of a sample population when designing a sampling scheme (see section on climate and seasonality in Chapter 4). In the end, a researcher is only able to estimate the size of the population that is actually catchable or detectable (both spatially and temporally).

Sampling units

In most cases the target population cannot be completely censused. As a result, a sampling scheme is required to estimate parameters for the sample population that are representative of the overall target population. One common sampling scheme is to select sampling elements (e.g., animals, quadrats) at random from the sample population, with all sampling elements having an equal probability of being selected. However, all sampling elements do not need to have an equal sampling probability provided that the probability of each being selected is known. This scheme is known as simple random sampling and is the basis for many basic statistical inference techniques. Simple random sampling does not ensure that all components (or strata) of the population are adequately represented in the sample unless there are a large number of samples relative to the size of the sample population. In addition to selecting a sampling unit and scheme, it is critical to do advance planning on sample size, so that enough individuals are detected to meet sample size requirements for the ensuing data analyses.

Systematic sampling involves selecting sampling units systematically across the entire sampling population (e.g., sampling every 10th quadrat in a linear transect). Systematic sampling increases the likelihood that all areas (or times) of a sampling population are equally represented in the sample. In most cases, systematic sampling does not yield data that can be appropriately used in statistical analyses that require entirely random sampling.

Stratified random sampling is a mixture between systematic and random sampling because it allows a researcher to capture variation that may exist among multiple strata and still ensure random sampling within a population. For example, a researcher may be interested in estimating snake abundance within a national park that contains four distinct habitat types (i.e., four strata). In this case, stratified random sampling consists of obtaining simple random samples from each of the four strata. By obtaining random samples from each of the four habitat types, the researcher gains at least three advantages over simple random sampling across the entire national park: (1) the researcher is assured of a representative sample across the entire national park, (2) the estimates should result in greater precision and lower variance than would have been possible with simple random samples collected across the national park, (3) the researcher will have habitat-specific estimates of snake abundance, which may prove more useful than completely random park-wide estimates of snake abundance (Conroy and Nichols 1996).

Spatial aspects

Many surveys do not have a spatial component, but most would benefit from accounting for the spatial aspects of a study during the planning stages. When choosing between the different sampling units, consider the spatial aspects of the sampling program, such as where the different sites are located on the landscape relative to other landscape features, such as streams, wetlands, topography, and human-made features, such as roads and buildings. Any deliberation about the sampling scheme should include a careful consideration of the biology and habitat associations of the target species, particularly when choosing where to sample in the landscape.

The spatial and temporal structure of any sampling design determines the types of statistical analyses that can be used and the kinds of questions that can be answered (see Chapter 6). It is important to take into account the tradeoff between bias and efficiency when choosing a sampling regime. For example, systematic and random sampling designs may be most efficient, but selecting the most ideal habitat in which to sample may introduce bias. Therefore, ideally the sampling scheme will minimize bias and maximize efficiency. A detailed account of all of the potential problems that can arise with sampling designs and how to properly choose a sampling design that obviates these problems is beyond the scope of this manual. Fortunately, there are several other sources that address these issues, including Hurlbert's (1984) classic paper on pseudoreplication and the design of ecological field studies.

INVENTORY

Creating species lists

The purpose of an inventory is to create a list of species known to occur in a focal area or habitat, not to assess population size or compare abundances over time (see Chapter 1). Thus, the goal of inventory should generally be to maximize the number of spe-

cies captured per unit effort. There are three ways to achieve this goal:

1. use a diversity of capture techniques

2. sample a diversity of habitat types

3. sample during peak periods of reptile and amphibian activity

If the ultimate goal is simply to create a species list, then the sampling scheme need not involve randomization, and sampling effort does not need to be standardized or repeatable, as these factors will decrease the number of species detected per unit effort (Gibbons et al. 1997).

Determining species presence/absence (i.e., detection/non-detection)

An inventory could be focused on identifying habitats or localities occupied by a single species. Many species of reptiles and amphibians occur at low densities or are cryptic and difficult to detect. If a species is not detected during a survey, the following question ultimately arises: has the species simply avoided capture or is the species truly absent from the study site? Distinguishing between these two scenarios can be critical, especially when surveying species of conservation concern. For example, if a population of a rare or cryptic species has been overlooked in inventories, then the population may not receive the protection or management it requires and local extinction could occur (Kéry 2002). To guard against this, researchers can use a simple probability model to calculate the number of unsuccessful visits to a site that are required to be statistically confident that a species is not present. Such "detectability" models have been developed by Guynn et al. (1985), McArdle (1990), and Reed (1996). Kéry (2002) provides a useful discussion of this technique and he has calculated the number of unsuccessful visits required to be 95% confident that each of three European snake species (*Vipera aspis*, *Coronella austriaca*, and *Natrix natrix*) do not inhabit a given locality. Detectability models can be applied to capture techniques for reptile and amphibian species and, when repeated over time, can be incorporated into monitoring programs. For example, detection probability models have been incorporated into site occupancy modeling for amphibians in the U.S. (Bailey et al. 2004a; Weir et al. 2005; MacKenzie et al. 2006; see below).

MONITORING

The purpose of monitoring is to evaluate the status and trends of populations in an area (see Chapter 1). Consequently, sampling designs for monitoring must be standardized and repeatable. Ideally, the sampling design should consist of systematic or random sampling across the entire population of interest (see "Hitting the target"), even at the cost of decreasing the total number of captures. Several levels of sampling intensity are available for monitoring populations of reptiles and amphibians (see Fig. 3-1). This section distinguishes between three general metrics of abundance that can be used to monitor population trends over time: site occupancy, relative abundance, and estimated population size. Additionally, this section details several commonly employed sampling schemes that have been used in conjunction with statistical techniques (see Chapter 6) to estimate population sizes and vital rates.

Site occupancy (detected/non-detected)

Site occupancy is measured as the proportion of sites or area occupied by a species. It may help to think of site occupancy as logistic regression with detectability factored in. Monitoring site occupancy is an ideal technique for documenting long-term or landscape-level changes in species occurrence, and is particularly useful for species that have large, naturally occurring population fluctuations (e.g., pond-breeding amphibians). Site occupancy is generally preferable to mark-recapture population estimators when monitoring a species over a geographically widespread area or for species with rapidly fluctuating population sizes. The rationale behind site occupancy models is that by monitoring presence/absence data for a species over large areas it may be possible to detect regional increases or declines in population persistence. As stated previously, declaring a species absent from a site can be difficult, therefore, detection probabilities must be taken into account to distinguish between absence and non-detection in site occupancy models (Bailey et al. 2004a; Weir et al. 2005).

The sampling scheme for occupancy models includes standardized, repeated observations (or trapping events) at each site. Each site should be visited a minimum of three times when detection probability is high (MacKenzie et al. 2006). During site visits the observer simply records whether or not a species was detected. Maximum likelihood techniques are then used to estimate occupancy and detectability from these capture histories (MacKenzie et al. 2002; Bailey et al. 2004a; MacKenzie et al. 2006; see Chapter 6). Whenever possible, it is preferable to determine the number of visits required by estimating detectability and occupancy in advance. A number of important factors are known to influence detectability in reptiles and amphibians, including, among other things,

habitat characteristics, observer/trapping bias, and environmental conditions (Kéry 2002; Bailey et al. 2004a; Weir et al. 2005). Therefore, it is important to carefully consider the natural history of the target species and the habitat characteristics of the sites when designing a sampling scheme (see Chapter 4). Likewise, this sampling scheme assumes that individuals are not entering or leaving a site during the sampling period. The difference between occupancy and use of a particular site must be recognized; an individual may move across and "use" that site, but not stay there. In particular, it may be necessary to measure variables that affect detectability so that they can be incorporated as covariates into occupancy models (e.g., Weir et al. 2005). Chapter 6 provides a full list of the assumptions of this technique and some of the sources for analyzing occupancy model data.

Several potential issues may arise with the use of occupancy models, including (1) meeting the closure requirement that no individuals can move in and out of a site during sampling, (2) obtaining an adequate number of visits to sites, and (3) having consistency in the number of sites available.

Relative abundance

A relatively inexpensive way to monitor the population status and abundance of a species is to determine the species' relative abundance over time (or among habitats, localities, etc.). Relative abundance is measured as the number of individuals captured per unit of sampling effort. Sampling effort can be based on visual observations (e.g., captures per man-hour), number of trap-nights (e.g., captures per trap-night), number of cover objects turned (e.g., captures per turn), or any other technique. However, some techniques (e.g., visual searches) are more sensitive to observer and sampling biases than other techniques (e.g., funnel trapping). When possible, it is important to quantify sampling biases and use those measurements as covariates in analyses (e.g., Rodda 1993). The sampling design and capture methods must be similar across the spatial and/or temporal scales that are being compared for measures of relative abundance to be valid. The reason for this necessity is two-fold: the use of relative abundance as an index of population size assumes that:

1. there is a direct linear relationship between relative abundance and population size

2. detection probability is constant across the temporal or spatial samples

To meet these assumptions it is often necessary to use standardized capture methods at similar times or weather conditions each year. Ultimately, capture data are a function of several independent factors, including:

1. population size

2. activity levels (including availability for capture)

3. detectability (likelihood an animal will be captured or observed)

In turn, activity levels, availability for capture, and detectability may be influenced by one or more of the following: age, body size, reproductive condition, season, sex, species, habitat type, and environmental conditions. Consequently, to accurately infer changes in population size based on relative abundance data, the sampling design must be carefully chosen to eliminate (or drastically reduce) all other factors that influence capture rates.

Ideally, if relative abundance is to be used on a large scale, a small-scale intensive study should be conducted to quantify the relationship between relative abundance and population size (e.g., see Rodda et al. 2005). Estimating relative abundance is a less intensive technique than estimating population size using mark-recapture or removal sampling, but it can be a useful tool for detecting large-scale population trends. An important advantage of relative abundance is that it can often be used to detect population trends for multiple species simultaneously, whereas mark-recapture studies tend to be species-specific because of the intensity of the sampling regime. Another advantage is that relative abundance data can be easily collected during long term inventory or species diversity studies. Nonetheless, confidence intervals for measures of relative abundance may be too wide to statistically detect trends in some species (e.g., Rodda et al. 2005) and mark-recapture population estimates can provide more detailed and accurate population information.

Population estimation

For some species, conservation efforts may require detailed knowledge of population sizes and/or vital rates (birth, immigration, death, and emigration rates). In most cases, it is impossible to completely census reptile and amphibian populations. Therefore, techniques such as mark-recapture and removal sampling have been developed to mathematically estimate population sizes and vital rates. All mark-recapture methods require animals to be captured and marked,

subsequently released, and recaptured or reobserved on one or more occasions. Chapter 6 details the mathematics and the assumptions behind each of these techniques. Here, I focus on the basic differences in sampling schemes required to meet the assumptions of closed-population models, open-population models, robust design models, and removal sampling models. This section is also designed as a guide to help you determine which sampling scheme and corresponding statistical technique may be the most appropriate for your monitoring project.

Closed-population models require a minimum of two sampling periods and are the least labor intensive of the population estimation techniques. The sampling scheme for closed-population analyses consists of two or more high-intensity sampling periods repeated over a short interval of time (see Fig. 3-2). Often, such sampling is performed on two consecutive days or on two consecutive sampling periods of a few days each. This scheme is used because the distinguishing feature of closed-population models is that the time between high-intensity sampling events must be 'closed' to (i.e., too short for) the possibility of births, immigration, death, or emigration. Thus, closed-population models do not provide estimates of vital rates (i.e., survivorship, population change over time). A closed-population sampling scheme should be envisioned as providing a 'snapshot' of the population size at a brief moment in time. Although it may seem intuitive, such a snapshot provides only an estimate of the population available for capture during the sampling interval (e.g., Bailey et al. 2004b). This is an important caveat because many reptile and amphibian species are prone to inactivity or use subterranean habitats that may make them temporarily unavailable for capture (e.g., an inactive, pregnant snake or an inactive, buried salamander). For such species, open-population models or robust design models may be required to estimate the size of the entire population because these models allow for temporary emigration (Bailey et al. 2004b). Animals do not need to be marked individually for closed-population analyses, but instead only require batch marks to distinguish between sampling periods. See Chapter 6 for more details on model assumptions and statistical methods for closed-population models.

Open-population models are 'open' to gains (immigration, birth) and losses (emigration, death) of animals between sampling periods. For open models, sampling intensity is usually constant over relatively long time periods. For example, a turtle study may require sampling a wetland once every month or year (see Fig. 3-2). Such a long interval between trapping events leaves the possibility of gains and losses to the population. Therefore, in addition to generating population size estimates, open models can be used to estimate survival and recruitment probabilities. However, all open models require three or more sampling periods, and thus are more resource-intensive than a closed model (see Chapter 6). In open-model designs, individuals must be given unique marks to calculate vital rates.

Robust design models are a combination of closed and open population models. The sampling regime for a robust design study involves sampling during both primary (open) and secondary (closed) sampling periods (see Fig. 3-2). The primary periods are used to estimate survival rates using an open model approach. The secondary periods are used to estimate population size and capture and recapture probabilities using a closed model approach. Information from both primary and secondary periods are combined to estimate temporary emigration rates. In reality, collecting data for a robust design model is no different than performing a series of closed-population model sampling events, with each sampling event separated by longer (open) periods of time (see Fig. 3-2). Obviously, robust design models are more labor-intensive than either closed or open population models alone, but they are preferable when the goal is to closely monitor both the population size and vital rates of one or a few focal populations (Bailey et al. 2004b; Bailey et al. 2004c). Robust design models are particularly valuable for reptile and amphibian species that exhibit temporal variation in detectability and that exhibit some form of temporary emigration as a consequence of their behavior or life history - for example, pond-breeding amphibians that immigrate to wetlands to breed and then emigrate back to terrestrial uplands during non-breeding periods, or terrestrial salamanders that migrate between subterranean and terrestrial habitats (Bailey et al. 2004b; Bailey et al. 2004c). Robust design models also increase the precision of parameter estimates such as survival and population size beyond those generated by open or closed models alone.

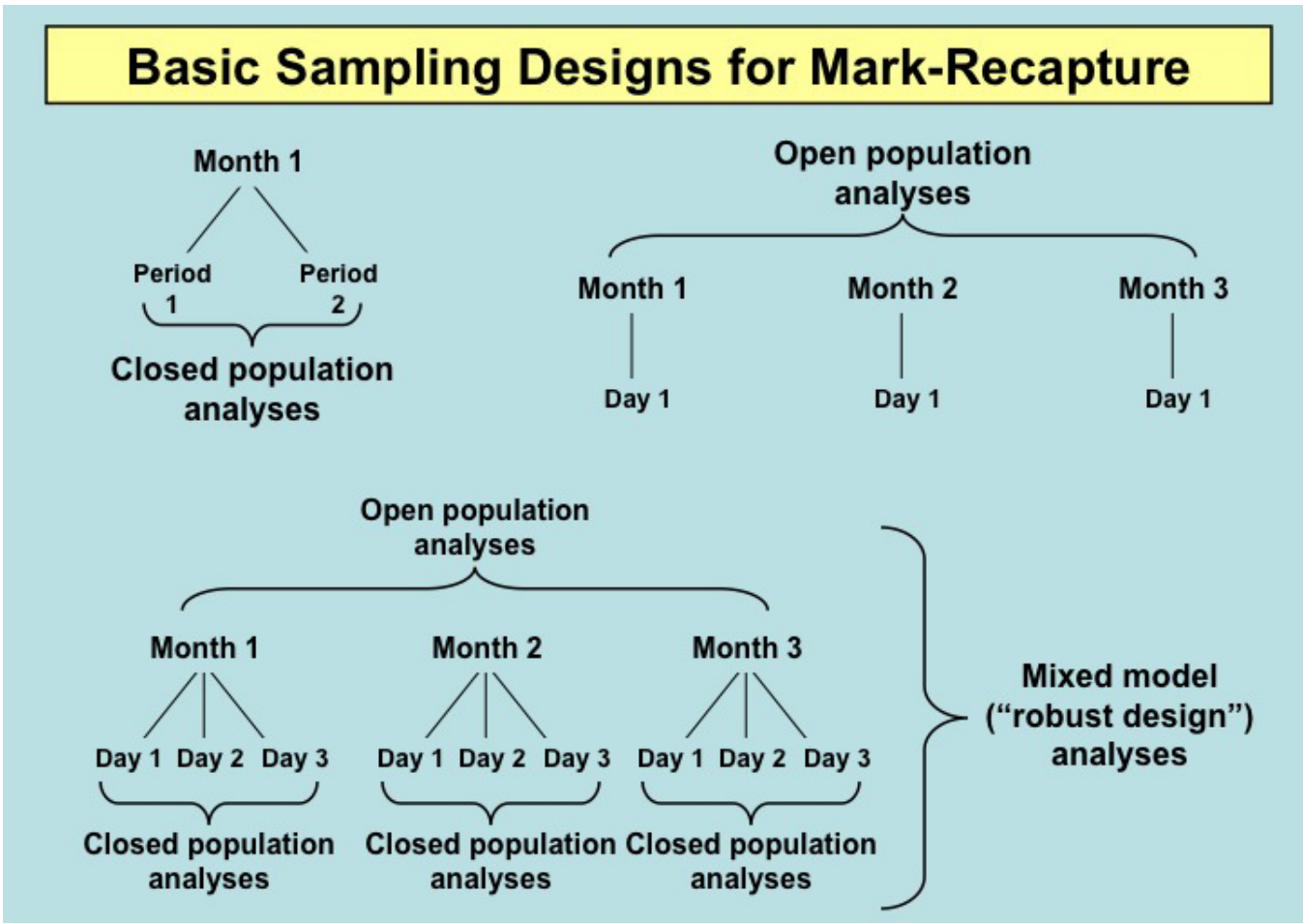
Removal sampling is a population estimation tool that requires animals to be physically removed from a population on two or more sampling occasions. Removal sampling is a useful technique for several small reptile and amphibian species (e.g., small terrestrial reptiles or amphibians), but the technique has limited applicability due to a variety of assumptions that are difficult to meet. First, the study species must be highly detectable and available for capture to allow for high removal rates. This assumption may not be true for many reptile and amphibian species due to their cryptic behavior, low activity, and/or subterranean habits. Second, sample plots must be large enough to represent the population of interest, but small enough to

facilitate removal of a large proportion of the animals. Third, sample plots must be closed (no net gains or losses) during the sampling period. In some situations this can be accomplished by erecting a barrier to prevent migration (e.g., see Bailey et al. 2004b). However, this technique does not work for species of reptiles and amphibians that cannot be confined to a small area (e.g., most large snakes, many climbing amphibians and lizards). Other assumptions include equal catchability of individuals within and across sampling periods and equal sampling effort across sampling periods.

Meeting the assumptions - For all mark-recapture and removal sampling population estimates, a researcher is only able to estimate the size of the population that is actually being sampled (see "Hitting the target"). Moreover, sampling designs that meet the assumptions of closure, openness, availability for capture, trapping effects, and effects of marking techniques on recapture rates, etc. are dependent upon the biology of the study species and are likely to vary among species. For example, the time interval for a sampling regime that is considered "closed" to mortality is probably much shorter for the little grass frog (*Pseudacris ocularis*) than for adult alligator snapping turtles (*Macrochelys temminckii*). Lastly, some species (or situations) may be too difficult to sample (e.g., species with low detectability; Bailey et al. 2004a) or generate too few recaptures for mark-recapture estimates to yield accurate population estimates. Consequently, good knowledge of the natural history of the target species is a prerequisite to properly designing sampling regimes to meet the assumptions of analyses.

CONCLUSIONS

The cryptic behavior and activity patterns of reptiles and amphibians can make them difficult to sample in many situations. In particular, reptile and amphibian species may be difficult to detect and, at times, unavailable for capture. As a result, careful consideration of sampling design, in light of the biology of the target species and the goals of a study, can go a long way towards developing a successful inventory or monitoring project and optimizing resource allocation.



Chris Winne

Figure 3-2. Basic sampling designs for mark-recapture population estimation models. The designs illustrated here are generalized examples and are not hard-and-fast rules. For example, here 'days' and 'months' are depicted to demonstrate a relative scale of time that should elapse between open and closed periods. For some species these may indeed be appropriate sampling designs, but for others, longer or shorter primary (or secondary) sampling periods may be required (see "Meeting the assumptions"). See text for further explanation of the models.


Southern alligator lizard (*Elgaria Multicarinata*)

Jamie Beltaso

CHAPTER 4. STUDY PLANNING AND DATA COLLECTION

INTRODUCTION

Michael E. Dorcas

To avoid potential complications associated with making data useful for inventory and monitoring, substantial planning is required before data collection proceeds. Specific questions that must be addressed before the first animal is captured include: 1) what types of data will be collected? 2) how will the data be collected? 3) how will the data be organized? 4) how complementary do datasets need to be? 5) how will data be analyzed to address the question(s) being posed (see Chapter 3)?

A variety of data should be taken into account during the planning stages and also collected during inventory and monitoring projects. These include data related to the animals collected (i.e., sex, size, activity), as well as climatic and seasonality data, data on the use of habitats and microhabitats, and locality data. Beyond the basics of recording the species, sex, and size of captured herpetofauna, researchers must decide which data need to be recorded for each animal. For example, are reproductive condition and mass important to the questions being asked? It is important to collect data that are relevant to the species of interest. For example, it would be very useful for a researcher to know water temperature at the time of inventorying aquatic animals because it may allow relationships to be established between an important environmental variable and animal activity. Today, simple automated data acquisition systems (i.e., dataloggers) provide relatively easy methods by which climatic data can be measured accurately and repeatedly.

Descriptions of habitats and microhabitats are important because they allow a better understanding of what is required in the physical environment of the animals. This allows for the development of improved sampling methods and ultimately, the compilation of information that is vital to the conservation of the animals. Thus, careful consideration should be taken when making decisions on habitat and microhabitat data collection.

A variety of geotechnologies have been developed that allow better collection of locality and habitat data. Inexpensive, handheld global positioning systems (GPS) allow accurate determination to within a few meters and are widely used in inventory and monitoring programs. Locality data from GPS units can then be incorporated, along with a variety of habitat layers into a geographical information system (GIS) that will allow detailed evaluation of habitat associations not previously possible.

Regardless of the types of data being collected, it is imperative that data be managed effectively so that data are collected and organized in a way that provides maximum value. Acquisition and proper application of required permits is also an important step in the planning process.

CAPTURE DATA

Walter E. Meshaka, Jr.

After determining the objectives of a proposed inventory or monitoring project, the researcher must decide which data need to be collected for each amphibian or reptile captured. The basic capture information, species, sex, and size, will likely be recorded, but more information may need to be taken.

Joe Mitchell

Supplies and tools needed for taking basic data at the time of capture. Includes a field notebook, caliper, measuring tape and ruler, Pesola mass scales, and a pencil, datasheet, and clipboard.

In an inventory, the basic information may be all that is needed, particularly if one is simply assessing presence-absence of a species. However, the goals of a monitoring program may require additional information, such as mass, reproductive condition, age, level of activity, or presence of abnormalities (i.e., malformations, skin condition, etc.), among others. For example, to monitor the reproductive success of a particular *Rana* species over multiple years, data on the reproductive condition of individuals entering and leaving a breeding site may be critical information (along with egg mass counts and subsequent counts of metamorphs emigrating from the site). Data on the marks given to each individual may also need to be recorded, for future reference. See Appendix II and III for explanations about determining an individual's age, sex, and reproductive condition, as well as marking methods for amphibians and reptiles.

Paul Crow

Weighing a hatching turtle.

CLIMATE AND SEASONALITY

Erin Clark and Gabrielle J. Graeter

Climatic factors, such as precipitation and temperature, and the variability in these factors, should be carefully considered during the planning stages of an inventory or monitoring project. By influencing the distributions of herpetofauna, as well as their feeding, migration, and reproductive activities, these climatic factors affect the outcome of sampling efforts. Climate affects the length of active seasons (Todd et al. 2007), timing of daily and reproductive activity (Todd and Winne 2006), thermoregulation strategies, and other factors of individual species behavior. Likewise, climatic factors (associated with rainfall or other meteorological events) are important for determining specific periods of species movement to breeding sites (Todd and Winne 2006), hibernacula, or nesting areas. Thus, a sampling design must address the variety of life history strategies, activity seasons, and patterns of daily behavior that different species possess.

Seasonality is crucial for determining the length of sampling periods in certain species. In combination with climatic factors, seasonal occurrence of species in particular habitat types can be predicted. For instance, some species occupy particular habitats during the winter and others during the summer months (Todd et al. 2007). Some species are only active during certain months of the year (e.g., gopher tortoise).

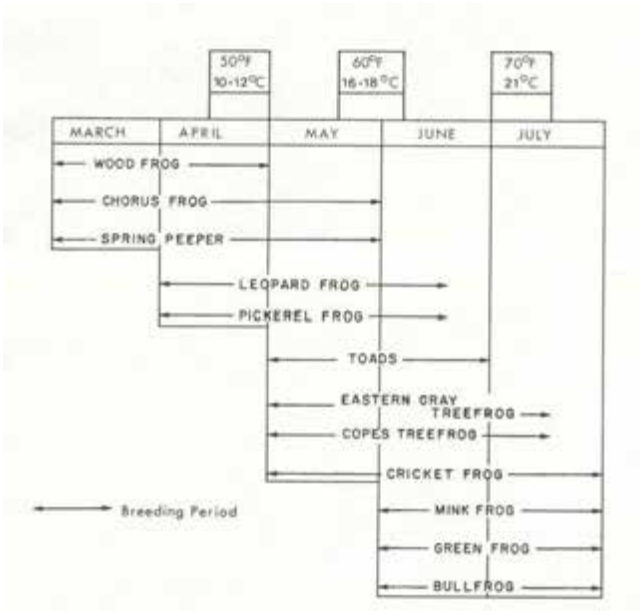


Figure 4-1. Consider the potential range for breeding of the target species.

Seasonality directly ties in with the timing of reproductive periods, such as the range of weeks or months in which certain frog species can be observed calling (Fig. 4-1). This active season or time spent in different habitats can vary throughout the range so the literature, as well as local herpetologists, should be consulted about the activity periods for a particular species in a given area. Likewise, climatic and seasonal factors can vary drastically from one year to the next, thereby influencing the outcome of a study (e.g., Fig. 4-2).

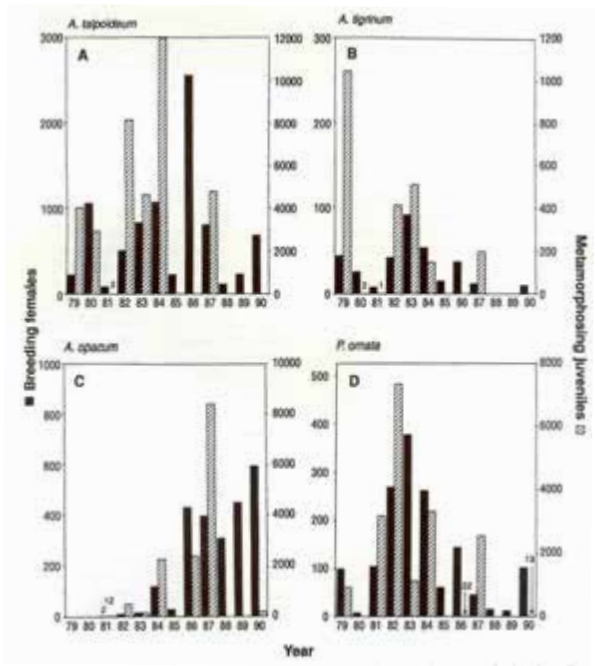


Figure 4-2. Variation among years in number of breeding females and metamorphosing juveniles of a given species can be large. Four species studied at Rainbow Bay study site on the Savannah River Site, SC demonstrate this variability.

Whit Gibbons

Seasonality of habitat within a year should be carefully considered in the designing of a study. Here is a seasonal isolated wetland in the summer, when it naturally dries completely.

Whit Gibbons

Here is the same wetland in the winter, after it has filled from rainfall.

In addition to planning according to climatic and seasonal factors, one should consider what types of climatic data to record during the inventory or monitoring program to meet program objectives. Basic weather data that should be collected in all inventory and monitoring projects include temperature (maximum and minimum, either continuously or daily, at a regular time) and precipitation (Crump 1994). There are many options for instruments that record temperature, including standard mercury thermometers, max-min thermometers, and dataloggers, among others. To measure air temperature, thermometers are often placed 2 m above the ground in a location that does not receive full sun. However, the goals of a study may necessitate a lower position to obtain temperatures closer to what the species of interest may experience. Other temperatures may need to be taken, such as water, soil, substrate, leaf litter, and even body temperature, but one should carefully consider the costs and benefits of recording each type because taking more measurements by hand can be quite time consuming (Crump 1994).

Peter Paul Van Dijk

Dry Bay in summer of 2003, a “dry” year.

Kurt Buhlmann

Annual variation in aquatic habitats should be considered in designing a study. This is “Dry Bay,” an isolated seasonal wetland in SC, in summer of 1993 (a “wet” year).

To measure daily precipitation, a rain gauge should be placed in an open area and checked once daily and emptied. However, to get an estimate of the amount of rainfall that makes it to the ground through the canopy and vegetation, the gauge should be placed within the forest. As with the equipment that measures temperature, there is a wide variety of instruments that measure precipitation, from the most basic type that is emptied manually to those that automatically record data and empty themselves (Crump 1994; Peterson and Dorcas 1994).

Other environmental data that researchers may wish to monitor are relative humidity, substrate moisture, barometric pressure, wind speed and direction, and water level and pH of breeding sites (Crump 1994). A sling psychrometer or digital hand-held thermohygrometer are two types of instruments generally used for measuring relative humidity. Relative humidity is especially important for amphibians because the amount of moisture in the air directly influences their level of activity. Similarly, the amount of moisture in the soil or leaf litter affects the activity of many amphibians and reptiles. Many types of sensors exist for measuring substrate moisture, including a tensiometer and several more elaborate types of moisture readers, including those that can be connected to dataloggers for continuous monitoring. Having data on changes in barometric pressure may also be helpful information for understanding capture data. Hand-held barometers are the simplest way to record barometric pressure, but dataloggers are also an option. Because the amount of wind can affect water loss in herpetofauna, your study objectives may necessitate recording wind speed and direction. To record wind speed, portable anemometers are generally used, but these data may also be acquired from a local weather station.

Gabrielle Graeter

A thermometer that displays the maximum and minumum temperature in a given time period can provide insights into sampling variability.

It is also a good idea to record the water level at breeding sites because the level influences the distribution and activity of some species, especially amphibians. For an inventory project, the maximum and minimum water level should be recorded, but more data may be needed for a monitoring study (e.g., changing profiles of the water depth in the water body; Crump 1994). Permanent water level gauges can be installed at each site, but other options include using a meter stick, automatic depth recorders, or dataloggers. Researchers may want to record the pH of a breeding site, particularly because of the possible link between acidification and amphibian declines (Crump 1994), and many types of pH meters are portable. The objectives of the study, as well as the availability of resources, personnel, and time, should be carefully considered before deciding which types of environmental data to record.

Gabrielle Graeter

Precipitation can be recorded with a non-automated rain gauge.

HABITAT AND MICROHABITAT

Erin Clark

Habitat types and microhabitat use are both important considerations in designing sampling efforts. Habitat is generally considered to be the environment surrounding an animal at a spatial scale of several meters to tens of meters. Microhabitats (as described by Inger 1994) are the precise places where an animal occurs within the environment. Microhabitat selection can be important in determining cover features, refugia, and local site selection for trap placement and cover boards. Herpetofauna certainly differ in their use of microhabitat sites not only by species, but within species throughout their range and across seasons. In addition, males and females may use different microhabitat features in different ways and at different times. Therefore, it is very important to record data on microhabitat use systematically and for each animal in a particular study.

The following basic microhabitat features (as adapted from Inger 1994) may need to be recorded for each animal:

1. Date and time of observation
2. Location, elevation, and vegetation type
3. Horizontal position (e.g., with reference to water bodies, vegetation, coarse woody debris or other debris, refugia, cover, breeding sites, overwintering sites)
4. Vertical position (expressed as height or depth in terrestrial and aquatic environments, respectively)
5. Substrate (e.g., logs, vegetation, soil, leaves, rocks).
6. Special information that does not fit easily into other categories (e.g., on/inside specialized habitat features)

It may also be useful to describe microhabitat in terms of the behavior associated with it; for instance, particular features are crucial for basking (e.g., turtles and lizards) or breeding. It is important to note that this list is general in nature; each of the above variables will need to be sorted into subcategories and can become much more complex, although not all field studies will choose to address microhabitat in the same detail. A fine example of the type of datasheet necessary to address microhabitat in amphibians in tropical and subtropical forests is provided by Inger (1994). It should be noted that different biomes will involve different descriptors than those he provided.

Kurt Buhlmann

A 1 m^2 quadrat made of PVC piping is commonly used for estimating the microhabitat in a study area.

WEIGHING THE COSTS AND BENEFITS OF RECORDING HABITAT DATA:

- Some information may be much more time consuming to collect.
- Researchers should carefully consider the necessity of collecting data that may cost valuable time in the field.
- Additional complexity in data collection will undoubtedly result in additional time spent in the field collection and data analysis phases of a project.
- Consider that all projects are constrained by limited funding, time, and personnel.
- We all would design perfect projects if we were not constrained by these factors, but consideration should always be given to the limitations these factors impose.

Larger-scale habitat features can be useful in determining appropriate areas for sampling. When combined with appropriate planning for seasonality and climatic factors, a consideration of habitat features can result in more effective sampling efforts. Geotechnologies now make it easier to identify generalized habitat types prior to the start of field research in many locations.

In general, habitat information should include (as adapted from McDiarmid 1994a):

1. Vegetation type (dominant forms or descriptive lists may be useful) or habitat type (if aquatic, water depth information over time is useful, as well as information on types of vegetation and their extent)
2. Climate information at each site
3. Degree of disturbance (e.g., forestry, fire, flooding, agriculture, edge effects) or duration of the habitat (if aquatic)
4. Other habitat factors (e.g., soil type, frequency of flooding, water features, nature of shorelines/borders and associated vegetation, bottom/substrate type if aquatic)

If modeling is an objective of the monitoring efforts of a particular species (e.g., predicting behavior under a particular management regime), data collected on habitat and microhabitat features will be critical to the study. Sampling efforts should be designed to allow researchers to collect habitat and microhabitat information efficiently, with minimal effort and in a standardized manner. Datasheets should make common habitat categories easy to select and include an area for comments when unusual or atypical habitat/microhabitat information is encountered.

In addition to the utility of habitat and microhabitat information for planning a sampling regime, these data are also useful for other reasons. For example, microhabitat data recorded for a single species over a relevant period of time can be used to describe the ecological distribution of that species in that particular area, and subsequently the distribution can be compared to other sites (Inger 1994). Furthermore, this information can have valuable conservation applications, such as an increased understanding of the microhabitat requirements of a threatened or endangered species or as valuable information for stream or wetland restoration programs. Although there are many advantages to recording habitat and microhabitat data for captures, the limitations need to be thoroughly considered during the planning stages (see box "weighing the costs and benefits of recording habitat data," above).

LOCALITY

Erin Clark

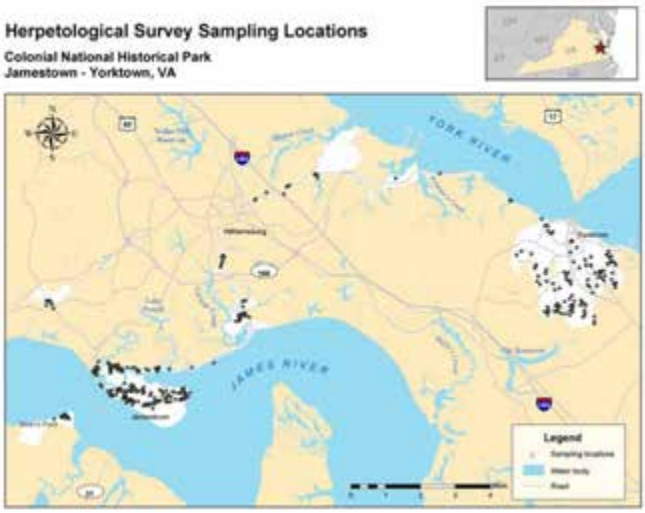
Information on locality (e.g., of a species, habitat types) is essential for the planning phase of most inventory and monitoring programs. This type of information can also be quite useful during fieldwork and data analysis. Three major types of tools – maps, photos, and geotechnologies – aid in assessing, documenting, and processing locality-related information.

Maps

There are four key features to consider regarding the use of maps during the design phase of an inventory or monitoring project. First, maps can be helpful in identifying a species' range and/or distribution. If project goals include documenting species occurrence, all species likely to be found in a given sampling area must be adequately documented. Distribution should be documented to the county level (at a minimum) in order to recognize animals as new records for an area or to make note of a species not recently documented in a given area.

Second, maps are often used in designing a sampling scheme. Differing habitat types may result in varied

opportunities for both trapping and sampling type. For example, if multiple habitat types are available, the researcher may want to use stratified random sampling to achieve equal representation of each habitat type. The use of maps to determine trap placement can save both time and energy in the setup phase of a project.



Joe Mitchell

Figure 4-3. A map of a study area can help identify species distributions and concentrations.

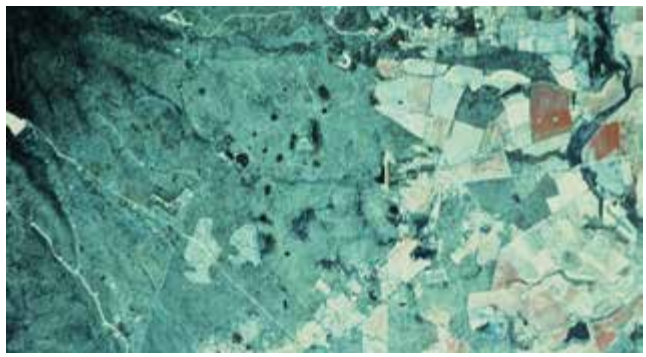
Third, maps are an essential piece of equipment for orienteering at potential field sites. The likelihood of becoming lost during field surveys should not be underestimated. Insufficient knowledge of sampling areas can decrease efficiency and result in downtime in the field. A good map of the areas to be sampled and sufficient map-reading skills will make sampling efforts easier and more effective.

Fourth, an inventory or monitoring program may require maps as an end product (see Fig. 4-3). Early in the sampling design process, the goals of the project should be used to determine the types of maps the project is likely to produce. These may include range maps, species occurrence in various habitats, areas of animal concentration, or other features. These maps can be essential to managers looking to avoid or locate high concentrations of rare species or areas likely to house them. Also, they allow managers to see areas in need of management with clear delineation. In addition, geotechnologies can make mapping easier and more efficient (see section on Geotechnologies in this chapter).

Photos

Photos of animals captured during sampling may be a valid way to document presence of the species (as long as identification of the species is possible from the photo). A number of reptiles and amphibians have characteristic features that allow voucher photos to be used. Furthermore, when a species is rare, it may be preferable to take pictures rather than collect the animals (see Appendix VII on using photographs for vouchering).

Like maps, landscape-level photos of a potential study area can be used in planning the location of study sites, determining habitat types, and anthropogenic influences, among others. Landscape photos are usually taken from an airplane or helicopter or even from satellites. They often lend an invaluable perspective on study areas due to an ability to reveal otherwise unknown patterns and to detect a variety of characteristics at different scales (see Figs. 4-4 and 4-5).



Kurt Buhlmann

Figure 4-4. Aerial photo of a sinkhole pond complex study area.

Photos serve another key purpose in inventory and monitoring efforts. They allow us to individually recognize previously seen animals of some species (e.g., *Ambystoma opacum*; see Appendix III on marking amphibians and reptiles) and document habitat and microhabitat features, animal behavior, and interactions. These photo records can be used during data analysis to recall, interpret, and supplement information in field notes and datasheets. Cameras should be standard equipment in all inventory and monitoring efforts and readily available to field personnel.

Whit Gibbons

Figure 4-5. Aerial photos of a study site can reveal important features

Geotechnologies

Geotechnologies include the following four elements:

1. Geographic Information Systems (GIS)
2. Geographic Positioning Systems (GPS)
3. Remote sensing
4. Databases

The use of geotechnologies has increased in recent years, and for good reason, because they are rapidly transforming the way that many studies are conducted. For example, these technologies have proved useful in many sectors of society, ranging as broad as city planning, anthropology, wildlife studies, and business marketing. In the ecology and wildlife management fields, the application of these technologies is vast and new techniques and uses are continually being developed.

Tracey Tuberville

A GPS unit can be useful for recording location of captured individuals

GPS technology has three basic components:

1. Satellites in space that continuously broadcast broad-spectrum radio signals.
2. A network of control stations and ground monitoring that maintain the time standard for the system and calculate exact orbital information for satellites.
3. Individually carried GPS signal receivers on the ground (Millspaugh and Marzluff 2001).

GPS receivers can obtain horizontal position estimates within 20 m of their true location 95% of the time (with increased accuracy using differential correction; Wells 1986; Millspaugh and Marzluff 2001). In fact, the newest GPS systems have sub-meter accuracy (e.g., Trimble backpack units, as of May 2008) and technology will likely continue to improve.

GIS is generally defined as a computer system which can assemble, store, manipulate, and display data which are geographically referenced. Usually, GIS is also considered to include the data that go into the system and the operating personnel. GIS can be a useful tool during the planning stages of an inventory or monitoring program. For example, habitat and preliminary species distribution maps created in ArcGIS may be helpful for assessing the different habitat types in a study area and in determining a sampling scheme for all species of interest. The information acquired through a GIS for study planning and design purposes will likely ensure greater efficiency during sampling.

Certain considerations regarding data for use in GIS should be addressed early in experimental design. Currently, when data are collected and processed by GIS software, the error for each location is determined and a correction of points is automatically conducted. However, a number of factors can improve the quality of data and should be considered. The use of data dictionaries in some GPS units (e.g., Trimble GPS units) allows researchers to collect standardized information with each data point based on pre-defined data fields. Therefore, the assignment of criteria and choices within data fields should be logically and accurately assigned before sampling begins. Identifiers (like ID fields) specific to project purposes can be easily added to data dictionaries (although if each point is saved as a separate file, a unique file name will be assigned to the point by the Trimble Unit). No matter how many fields are available in a data dictionary, it is advisable to also provide a "Comments/Notes" field. Data should be collected, where possible, in a projection or datum (e.g., WGS 84, NAD 27, NAD 83) compatible with other data sets already existing in a given area. This allows data sets to be combined and potentially analyzed together. A geographic point can be displaced by nearly 100 m if expressed in a different datum (August et al. 1996), therefore it is important that data be collected in a standardized datum. Further, clear knowledge of coverage available for sampling sites and study areas may make data collection using GIS easier to define.

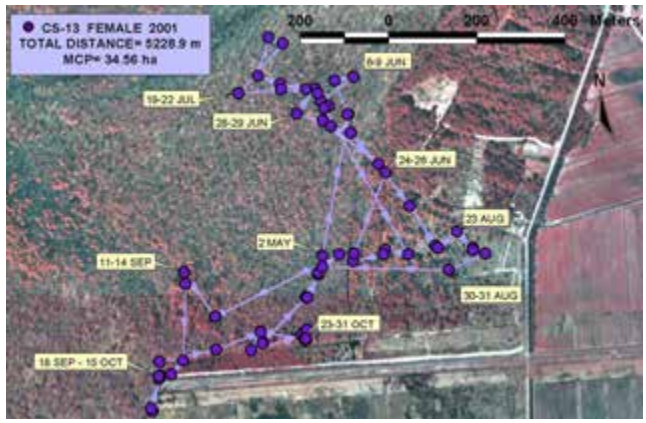
GIS also has many helpful applications and advantages beyond the planning stages of an amphibian and reptile inventory or monitoring project. This technology is valuable in many situations because it allows for greater efficacy and accuracy during the fieldwork

and data analysis phases. For example, in the field, GPS receivers can help with recording and navigating to specific locations. In most receivers there is a navigation option that provides a map or other device that can direct researchers to previously marked locations or to fixed landmarks. This can be useful during sampling both in finding sampling locations and in reducing time spent in the field recording location data.

The use of this technology provides many opportunities for analysis of field data which were in the past more difficult and less accurate. Some examples of analytical procedures found in GIS technology include overlay functions, proximity analysis, and three-dimensional data modeling (August et al. 1996). A number of overlays are widely available including drainages, climate, habitats, topography, soils, roads, and human features. Data are expressed in GIS format as points, lines, polygons, and grid cells (August et al. 1996).

Determining habitat use (particularly when combined with microhabitat data) has become easier with the use of GIS. This technology has allowed improvements in mapping of habitat, species range, movement, and study area (e.g., see Fig. 4-6). It also allows researchers (through the use of extensions) to model animal behavior through habitats, across environmental gradients, and throughout time. GIS technology has been used recently in herpetology to identify and find sampling locations for large-scale projects, create maps of habitat, to mark burrow locations for tortoises, to define home range size for a number of species, and to look at spatio-temporal movement patterns. A number of extensions exist in GIS software for the analysis of data. For specific examples of analysis programs developed within the GIS framework, see: Hooge and Eichenlaub (1997) and Rogers and Carr (1998). Although a full overview of creation of a GIS database from analog data will not be covered in detail here, sources are available for this purpose (see August et al. 1996 or August 1993).

GIS has also proved helpful in modeling the effects of land management on populations. For example, the effect of varying forest management plans on terrestrial salamander populations was modeled using GIS in order to create a predictive map of salamander abundance (Gustafson et al. 2001). Many land managers and biologists are also using GIS to evaluate current management plans and make recommendations for future plans (e.g., Willson and Dorcas 2003a). Predicting the occurrence of a species in the landscape is a major focus of many conservation efforts and much has been researched and written on this topic (Scott et al. 2002).



Chris Peterson

Figure 4-6. Map showing tracked movements of a Timber Rattlesnake.

Geotechnologies should not be used as a substitute for field work and data collection, but instead as a way to enhance the utility of data and provide more dynamic analysis of it (e.g., telemetry data). As with any technology, the limitations (e.g., expensive equipment, more time-consuming, accuracy limited in certain situations, limited GIS data and coverage in some study areas) should be carefully considered before designing and implementing a sampling program that involves these techniques.

AUTOMATED DATA ACQUISITION

Michael E. Dorcas and Charles R. Peterson

Herein we describe methods to automatically and continuously measure variation in the physical environment and activity of amphibians and reptiles. Data quantifying the relationship between environmental variation and herpetofaunal activity and physiological state (e.g., temperature) can be used to optimize sampling procedures for inventory and monitoring programs and in the interpretation of population changes (Peterson and Dorcas 1992; 1994). Because many factors vary through time, it is often important to sample regularly over hours, days, and even seasons. For example, the environmental temperatures available to an amphibian or reptile may change dramatically over just a few hours and over extended periods of time (Peterson et al. 1993). Automated sampling systems make it possible to measure continuously a wide variety of variables accurately at one or more sites.

We discuss how to automatically monitor the environments of herpetofauna using dataloggers, environmental sensors, and physical models and how to automatically monitor animal body temperatures using dataloggers. We also discuss how to automatically monitor movement and activity using automated

radiotelemetry, global positioning systems (GPS), passive integrated transponders (PIT) tags, and automated photographic and video techniques. We have included the names of various manufacturers, especially for the equipment and materials that we have used. However, our experience with different types of equipment and techniques is limited and the listing of a particular vendor does not indicate our endorsement. Furthermore, the evolution of equipment is proceeding rapidly and many of our specific comments may already be out of date.

DATALOGGERS

A datalogger is an instrument that records and stores data collected through the use of one or more sensors. Dataloggers are generally programmed by a computer, allowed to operate autonomously, and then the data are downloaded after some prescribed time period. Dataloggers measure voltages produced by sensors (i.e., transducers such as thermistors) and convert raw voltages into the unit of interest (e.g., temperature). Some dataloggers can also be used to control various instruments, such as turning on and off a piece of equipment at specified intervals. In general, dataloggers can be classified into either single- or multi-channel dataloggers. Single-channel dataloggers measure only one sensor and multi-channel dataloggers measure two or more sensors.

Multi-channel dataloggers

Since the 1970s, the task of gathering continuous data has been greatly facilitated by the development of microprocessor-based dataloggers. The distinguishing characteristics of field dataloggers include portability, programmability, battery power, and the ability to read inputs from several types of sensors at user selected intervals (Campbell 1990). Dataloggers have numerous advantages over devices such as mechanical recorders and strip chart recorders, including a wider operating temperature range, increased sensor compatibility, higher accuracy, greater data storage capacity, and much easier transfer of data to computers for analysis (Pearcy et al. 1989).

Factors to consider when selecting a multichannel datalogger include cost, reliability, the range of operating conditions (temperature and humidity), accuracy, resolution, number of channels, sensor compatibility, processing power, data storage and retrieval options, and power requirements (Tanner 1990). Costs range from approximately $60 to over $5000 US. Powerful, versatile systems, capable of measuring multiple sensors, are available for less than $1700 (including the interface and software for downloading data). Features to look for in this price range include: 12-bit or greater resolution, the ability to measure microvolts (e.g., thermocouples), switch or pulse-counting capability (for cup anemometers and tipping-bucket rain gauges), the ability to provide excitation voltages (for thermistors and electrical resistance humidity sensors), and digital outputs for controlling devices such as tape recorders and radiotelemetry systems. A more expensive datalogger, capable of resolving nanovolts, is required for measurements of some variables (e.g., soil water potentials using thermocouple psychrometers). If you cannot afford to purchase a datalogger, you may be able to borrow one or simply use data being collected by a datalogger already in use at or near your study site.

Many dataloggers need to be placed into some type of enclosure to protect them from weather conditions and vandals. Most manufacturers offer enclosures. A less expensive alternative is a small plastic toolbox. Better yet, a dry storage box used in boats provides an inexpensive waterproof enclosure suitable for many applications. In areas exposed to direct sunlight, it may be necessary to paint the enclosure white or to shade it to prevent overheating. The use of a desiccant (e.g., silica gel) may be required in humid environments to keep conditions in the enclosure within the range of operating conditions of the datalogger. To prevent vandalism, enclosures can be buried to hide them. Burial also reduces the range of temperatures to which the datalogger is exposed. In areas prone to flooding (e.g., in the floodplain of a river) dataloggers can be protected from rising water by suspending them from trees using rope.

Another problem frequently encountered is damage to sensors or sensor wires, often due to animals. Because thermocouples or thermistors often must extend a considerable distance from the datalogger, they are often vulnerable to damage. We have had problems with moose (*Alces alces*) and other large mammals incidentally damaging or pulling thermocouples loose from their attachment to the datalogger. Burying or covering the exposed parts of the wires with moderate-sized rocks or logs often helps to prevent this from occurring. Another problem has been failure of the thermistor or thermocouple sensors due to damage by rodents chewing on the wire insulation. To prevent this, thermistor wires can be enclosed in small diameter PVC pipe.

DEPLOYING AND PROGRAMMING DATALOGGERS:

Training

- Powerful & flexible systems (e.g., Campbell Scientific) take considerable time to learn
- Simpler systems (e.g., Onset Computer) are easier to learn but are less flexible
- Work with someone familiar with equipment
- Some manufacturers offer training sessions
- Most dataloggers come with user-friendly software

Case Study: Onset Computer Corporation Weather Station by M. E. Dorcas

- Relatively inexpensive
- Easier to use than Campbell systems
- Has up to 15 sensors
- Can be set up in an afternoon if user has moderate technical and computer skills
- Runs for up to a year on 4 AA batteries
- Holds 500,000 measurements in memory
- Uses "smart sensor" technology (datalogger automatically detects the sensor that is plugged in and calculates the proper units)
- An Onset weather station was used for 3 years on Davidson College Ecological Preserve in Mecklenburg County, NC with minimal problems

Single-channel dataloggers

Single-channel dataloggers can provide several advantages when compared to multi-channel systems. Single-channel dataloggers provide much simpler and less expensive alternatives to the measurement of environmental variables when only one or a few variables need to be measured. Moreover, single-channel dataloggers are ideal if environmental variables must be measured at locations some distance apart. Also, because of their small size, single-channel loggers can often be much more easily deployed, protected from environmental conditions, and hidden from potential vandals if needed. Specially designed, single-channel dataloggers are available to measure temperature, solar and lunar radiation, barometric pressure, relative humidity, precipitation, soil moisture, and wind speed.

We have experience using two different types of single-channel dataloggers. For most of our applications requiring monitoring of a single or few variables, we have used dataloggers from Onset Computer Corporation ($60 to $400 US). These single-channel dataloggers are easily programmed and downloaded through a serial or USB interface using the BoxCar or Hoboware software available from Onset. The software runs on Windows or Macintosh computers, is easy to use, automatically recognizes the type of datalogger attached, and allows downloaded data to be easily exported in text or Microsoft Excel formats for analysis.

The development by Dallas Semiconductor of the iButton Thermochron datalogger provides a considerably less expensive and smaller alternative to many of the dataloggers above when temperature measurements are required. Approximately the size of four stacked dimes, Thermochrons measure 5.9 mm in thickness, 17.4 mm in diameter, and weigh 3.12 g. Thermochrons are relatively inexpensive (approximately $15 each). Thermochrons were found to be very reliable and accurate in laboratory calibrations (Angilletta and Krochmal 2003) and are programmed through a USB computer interface. Thermochrons are somewhat limited in capacity, recording from 2048 to 8192 time and date-stamped temperature readings with 0.5°C resolution at sampling intervals selected by the user (Angilletta and Krochmal 2003; Roberts and Thompson 2003).

Both Onset Computer and Dallas Semiconductor provide software and interface cables that allow downloading and limited programming of their dataloggers using personal digital assistants (PDAs) running the Palm operating system. You can also use tablet PCs to interface with dataloggers in the field.

ENVIRONMENTAL SENSORS

The following sections describe sensors that are most often used in conjunction with dataloggers to measure important environmental variables. For weather stations, sensors are usually mounted on an instrument tripod which can be purchased from a supplier or constructed. Figure 4-7 shows examples of measurements of air and soil temperatures, radiation, relative humidity, and wind speed measurements plotted versus hour of day. We do not have extensive experience with measuring all of these variables, and thus have

had to rely on the literature or advice from engineers or other scientists to prepare some of the following material. See Flowers (1978), Fritschen and Gay (1979), WMO (1983), Marshall and Woodward (1985), Finklestein et al. (1986), Bingham and Long (1988), Pearcy et al. (1989), Skaar et al. (1989), Campbell (1990), and Tanner (1990) for more information on sensor instrumentation.

Temperature

For a variety of reasons, thermocouples are often the preferred sensors to use in many field studies requiring automated temperature measurements. They are relatively accurate, inexpensive (about $1.75 per meter), are available in a wide range of sizes, respond quickly, and can be used over long distances without affecting the signal (Pearcy et al. 1989). For temperature measurements in the range of biological interest (-70 to 100°C), Type T (copper-constantan) thermocouples are most appropriate because this combination of metals produces a relatively large voltage that changes linearly with respect to temperature (approximately 40 µv/°C). Thermocouples come in a wide range of sizes; 24 gauge (0.5 mm diameter) wire is commonly used for measuring water, soil, and air temperatures. Thermocouples can be easily made by stripping the ends of the wire, twisting the exposed ends together, and then soldering the ends together (Pearcy et al. 1989).

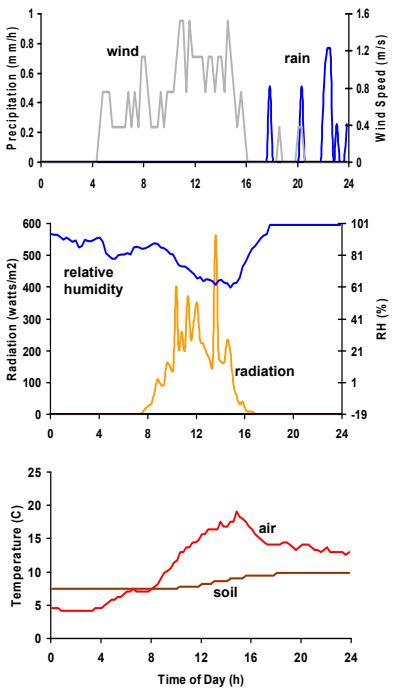


Figure 4-7. Variation in wind, precipitation, relative humidity, solar radiation, and air and 20 cm soil temperatures, sampled using a weather station (Onset Computer Corp.) every 30 minutes for an entire day 4 December 2005 on the Davidson College Ecological Preserve.

Thermistors (temperature sensitive resistors) are another commonly used device for measuring temperature. Advantages include high sensitivity and accuracy and fast response time. However, thermistors are more expensive, less rugged, and cannot easily be used over long distances. Some dataloggers can read both thermocouples and thermistors easily, whereas other dataloggers may be unable to read either thermistors or thermocouples.

Most single-channel temperature dataloggers use thermistors as the temperature sensor, but some can measure thermocouples. Some single-channel dataloggers come with the sensor built-in as part of the logger and thus temperature fluctuation of the entire datalogger (or a portion of it) is actually measured. Others provide the option of plugging in a thermistor or thermocouple for measurement some distance away from the datalogger. Many single-channel dataloggers designed to measure temperature do not require any special housing and can simply be mounted or positioned where desired. Specially designed, waterproof, single-channel dataloggers for measuring water temperature are available (e.g., Stowaway-Tidbits, Onset Computer) and can be tethered and tossed into the water. Other dataloggers may be used to monitor water temperature, but may require waterproof housing (e.g., wide-mouthed plastic bottles such as Nalgene) with the temperature sensor (e.g., thermistor) exiting the housing through a sealed port.

We usually measure air temperatures at animal height (e.g., 1 cm) and/or at 2 m (a standard reference height for meteorological stations), water temperatures on the bottom and 1 cm from the surface, and soil temperatures from at least two depths (e.g., 1 and 20 cm) if we are interested in burrow temperatures. Thermocouples used to measure air temperatures usually should be shaded. Some manufacturers provide radiation shields for temperature dataloggers and other sensors at a moderate cost. To measure shallow water temperature in aquatic environments where the water depth may fluctuate greatly over short periods of time, we attach the end of the thermistor or thermocouple to a float (e.g., a bobber used for fishing) which is attached loosely to a metal rod anchored in the bottom so that it can rise and fall with fluctuating water levels.

Physical models and operative temperatures

Temperature is one of the most important factors influencing the activity of reptiles and amphibians, and thus our ability to determine their presence and abundance (Feder and Burggren 1992; Peterson et al. 1993). For this reason, it is important to describe accu-

rately the thermal environments of herpetofauna. The thermal environment of submerged, aquatic amphibians and reptiles (e.g., tadpoles and some turtles) can be characterized relatively easily by measuring the temperature of the surrounding water. Describing the thermal environments of terrestrial herpetofauna is more complex because a variety of factors interact to determine their body temperatures. These factors include air temperature, substrate temperature, radiation, humidity, soil moisture, wind speed, and animal properties such as size, shape, rate of evaporative water loss, and reflectivity (Bakken 1992). A single-number representation of the thermal environment that incorporates these factors is the operative temperature (Bakken and Gates 1975; Bakken 1992).

A simple and relatively inexpensive approach for measuring operative temperatures involves the use of physical models which incorporate animal properties such as size, shape, and reflectivity (Bakken and Gates 1975). This approach has been applied with considerable success to dry-skinned ectothermic vertebrates (i.e., reptiles; e.g., Crawford et al. 1983; Peterson 1987; Grant and Dunham 1988) and with limited success to amphibians (O'Conner and Tracey 1992; Schwarzkopf and Alford 1996). The challenge with amphibian biophysical models is incorporating evaporative water loss. Some authors have been successful in using agar models that evaporate water at similar rates to amphibians (Schwarzkopf and Alford 1996). Although many studies have used hollow casts of actual reptiles, an easy method of constructing physical models for snakes involves using sections of hollow copper tubing of approximately the same length and diameter of a real snake. A copper model approximating the diameter and reflectance of the dorsal surface (accomplished by painting the copper tubing) of the study species usually closely matches the environmental temperature experienced by a real snake (Peterson et al. 1993). Vitt and Sartorius (1999) provide evidence that self-contained dataloggers such as Tidbits (Onset Computer Corp.) may serve as good estimators of body temperature for some small reptiles (e.g., many lizard species). Shine and Kerney (2001) provided a detailed analysis of various physical parameters that may affect physical models for reptiles.

Humidity

Electrical resistance (e.g., Phys-Chemical Research Corp.) or capacitance (e.g., Vaisala, Inc.) humidity sensors provide a convenient way of automatically measuring atmospheric water vapor (Tanner 1990). Costs range from $85 to $400. Some electrical humidity sensors may be damaged by condensation or air contaminants (Campbell 1990). Sensor elements need to be individually calibrated at least annually and may need to be replaced periodically. See Skaar et al. (1989) for a comparison of commercial hygrometers. Ventilated wet-bulb, dry-bulb psychrometers are more accurate but are more expensive, require power to run the fan, require attention to keep the water reservoir filled, and will not read accurately below freezing. Onset Computer Corporation provides moderately priced ($140-$200) dual-channel dataloggers that measure atmospheric water vapor and air temperature. The Hoboware software can calculate and output temperature, relative humidity, and dew point.

Precipitation

Precipitation can be measured automatically with a tipping bucket rain-gauge connected to a pulse counting channel on a datalogger (Tanner 1990). When a specified depth of water has collected, the bucket tips and empties. The number of tips is counted with the datalogger. Resolution in the range of 0.1 - 0.2 mm is possible (WMO 1983). Onset uses single-channel event recorders to record data from a single tipping bucket rain gauge (cost is about $400). To measure precipitation in the winter, tipping buckets can be heated so that snow will melt and the water will drain from the bucket. Weighing bucket rain gauges provide a more accurate but more expensive way to measure precipitation (Tanner 1990).

Radiation

Two types of solar radiation sensors (pyranometers) that are commonly used with dataloggers are silicon photocells (e.g., Li-Cor LI2000SB) and thermopile devices (e.g., Eppley Model PSP, Kipp and Zonen Model CM11). Silicon cells are considerably less expensive than the thermopile devices (approximately $200 vs. $1300-$3000). However, because their spectral response is limited to 400 nm to 1100 nm, silicon cells should not be used within vegetation canopies or to measure reflected radiation (Tanner 1990).

Single-channel radiometers that measure solar and lunar radiation are available from Onset Computer. Lunar radiation has been shown to affect the activity of some reptiles (Clarke et al. 1996) and amphibians (Fitzgerald and Bider 1974). We used one of these (Stowaway LI) to measure solar and lunar radiation as part of a study examining factors affecting snake activity in South Carolina (Willson et al. 2006). Light intensity is recorded on a logarithmic scale (log Lumens/m2), thus allowing easy visualization of lunar and solar radiation (Fig. 4-8).

Wind speed

Wind speed can be automatically measured with a cup anemometer and the pulse counting channel of a datalogger. Cup anemometers are omnidirectional, have linear responses, and are reasonably precise (Campbell 1990). Factors to consider when selecting an anemometer include size, the range of wind speeds over which the sensor operates (especially the starting and stopping thresholds), cost, and durability. Propeller anemometers have lower thresholds and can be used to measure wind direction but are more expensive (Campbell 1990). Anemometers are usually mounted at a height of 2 m. If more than one anemometer is available, wind profiles can be determined so that the 2 m wind speed can be used to calculate wind speeds at other heights.

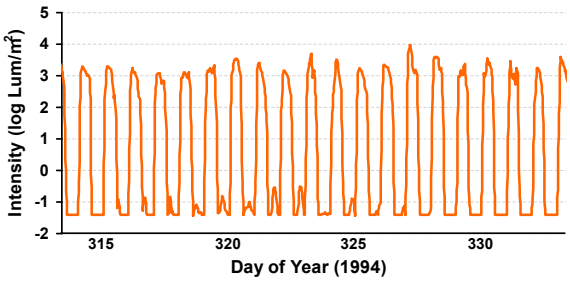


Figure 4-8. Variation in light intensity (solar and lunar radition) over a 20-day period in November 1994 at Ellenton Bay on the Savannah River Site, SC. Note the progressive delay in moon rise from days 316 until 327 and the absence of measurable lunar radiation after day 328.

AUTOMATED MONITORING OF ANIMALS

Monitoring animal activity and correlating that activity with environmental variation allows predictions of when animals are most active and thus, most easily sampled for inventory or monitoring purposes (Peterson and Dorcas 1992). Automated systems that provide information on animal locations, thus allowing analysis of habitats most used by animals, assist in further refinement of sampling efforts. Some amphibian and reptile species or groups are more easily monitored automatically than other species. For example, the use of satellite GPS may work for monitoring large chelonians such as sea turtles, but will not work for smaller species of reptiles or any amphibian due to the size of the equipment. A variety of methods have been used to automatically monitor amphibian and reptile populations and some methods originally used for monitoring other animal groups can be adapted to monitor herpetofauna.

Automated radiotelemetry

Radiotelemetry has been used extensively to monitor the movements and habitat use of many animals, including many species of reptiles and amphibians (Millspaugh and Marzluff 2001). Automated systems used in conjunction with radiotelemetric techniques can also be used to automatically, continuously monitor body temperature variation in many species of reptiles and amphibians. Accurate inferences of activity patterns and microhabitat use based on body temperature variation, especially when combined with environmental temperatures, can then be made (Fig. 4-9).

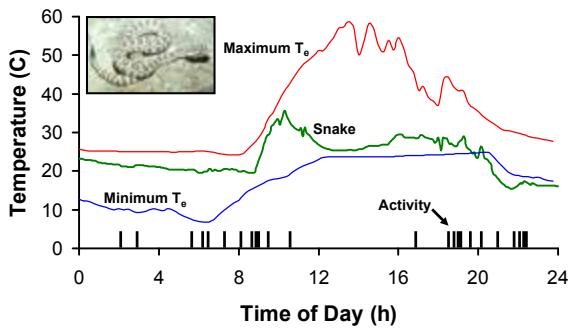


Figure 4-9. Variation in the body temperature and activity of a Great Basin rattlesnake (*Crotalus lutosus*) in southeastern Idaho over an entire day in the summer of 1991. Temperature was monitored using a thermistor within a surgically implanted radiotransmitter which varied the transmitter pulse rate. Activity was determined by toggles of a mercury switch within the transmitter. Transmitter pulse rate was monitored with an automated system described in the text. Environmental temperatures (Te) were determined with physical snake models and a Campbell datalogger (Cobb 1994).

Generally, the factor determining whether or not a species is suitable for radiotelemetric projects is size; larger animals are generally more easily outfitted with radiotransmitters. However, the continued development and refinement of telemetry equipment is making it possible to use automated telemetry techniques to monitor smaller and smaller species. For example, using an automated monitoring system, we have monitored the body temperatures of a male rubber boa weighing only 35 g for three months using a small, surgically-implanted transmitter (approximately 2 grams; BD-2, Holohil Ltd., Ontario). The snake faired well, gaining 15 g during the study period.

For snakes, most transmitters must be surgically implanted and thus, temperature sensitive transmitters allow accurate automated monitoring of body temperature (Peterson et al. 1993). Temperature-sensitive transmitters use a temperature-sensitive resistor (thermistor) which causes the pulse rate of the transmitter to vary positively with temperature. Addition of a temperature sensitive option to a typical transmitter generally adds about $50 to $75 US to the cost, but

does not substantially increase the size or mass of the transmitter. Temperature-sensitive transmitters should be calibrated in a waterbath before surgical implantation and equations developed (we typically use a second or third order polynomial) that describe the relationship between temperature and interpulse interval. Some manufacturers calibrate temperature sensitive transmitters for the researcher but we recommend verification of those calibrations. We also highly recommend that transmitters be calibrated after removal from the animal to check for a shift in the temperature sensitivity.

Temperature-sensitive radiotransmitters, either surgically implanted or externally attached, could also be automatically monitored. The movements of moderately sized salamanders (*Ambystoma sp.*) and anurans (*Bufo sp.*) have been monitored using surgically implanted or externally attached radiotransmitters (approximately 2 grams). The inclusion of a temperature option generally adds minimal weight to the transmitter.

A variety of automated systems have been developed to monitor animals outfitted with radiotransmitters. Automated systems that can monitor temperature-sensitive radiotransmitters usually are designed to record the interpulse interval rate of radiotransmitters. The simplest of these systems monitors only one animal (i.e., one frequency) and uses a recycling timer to automatically turn a radio receiver and cassette tape recorder on and off at regular intervals (Beaupre and Beaupre 1994; Cobb 1994; Lutterschmidt et al. 1996). The tape must then be manually decoded (i.e., time the pulse intervals) using a stop watch or other timer. More advanced systems can systematically scan over many frequencies (i.e., monitor multiple animals), time the interpulse intervals, and then store the data in memory. We have used a Fast Data System (Telonics, Mesa, AZ) for this purpose to study body temperature variation in free-ranging garter snakes (*Thamnophis elegans*; Peterson 1987) and in free-ranging rubber boas (*Charina bottae*; Dorcas and Peterson 1998). More advanced systems have been developed using dataloggers to turn on and off the receivers, change frequencies of the receivers, and time interpulse intervals (Grothe 1992; Cobb 1994). LoTek has developed an elaborate, but expensive (approx. $14,000 US) system which allows the user to enter calibration values so that interpulse intervals are automatically converted into body temperatures (S. Beaupre pers. comm.).

Some manufacturers can outfit their transmitters with activity switches (usually mercury switches) that allow detection of animal movements (Grothe 1992; Cobb 1994). When combined with temperature-sensitive capabilities and automatic monitoring, these units allow strong inferences to be generated regarding activity patterns and microhabitat use (Fig. 4-10). Because the amount of movement necessary to trigger the switch is dependent on the position of the sensor in the animal, we recommend only intra-individual comparisons. We also recommend calibration of these activity transmitters to determine how much movement is required to trigger the sensor.

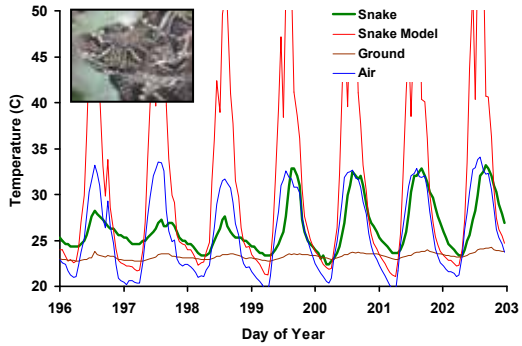


Figure 4-10. Variation in body temperature of an eastern diamondback rattlesnake (*Crotalus adamanteus*) over a one-week period in August 1999 in southeast South Carolina. Body temperature was measured using surgically-implanted dataloggers (see text). Environmental temperatures were measured using thermocouples and a multi-channel datalogger (CR-10, Campbell Scientific, Logan UT). Note that the snake apparently was in a shallow burrow the first 3 days and then remained on the surface and in the shade for the rest of the week.

Animal temperature monitoring using dataloggers

Attachment or implantation of temperature dataloggers to amphibians and reptiles can allow automated monitoring of body temperature. One of us (M.E.D.) used Onset Tidbit dataloggers to automatically monitor the body temperatures of eastern diamondback rattlesnakes (*Crotalus adamanteus*) while tracking their movements using radiotelemetry. Tidbits were programmed and coated in a polymer (Elvax, Minimitter, Sunriver, OR) before implantation into the body cavity. By examining body temperature plots combined with environmental data, we can infer activity patterns and microhabitat use (Fig. 4-10).

We have successfully used Thermochron iButtons to measure seasonal body temperature variation and infer activity patterns (e.g., basking) in painted turtles (*Chrysemys picta*) inhabiting a farm pond in Davidson, NC (Fig. 4-11; Grayson and Dorcas 2004). We glued the Thermochrons to plastic cable ties and attached them through holes drilled in the marginal scutes of the turtles (Fig. 4-12). After programming, Thermochrons were sealed using plastic tool dip (Plasti Dip International, Circle Pines, MN; a fast drying liquid

plastic traditionally used to coat the grips of hand tools). We recovered and successfully downloaded 32 of 53 Thermochrons from the turtles (3 dataloggers were lost from turtles and 17 were not recaptured).

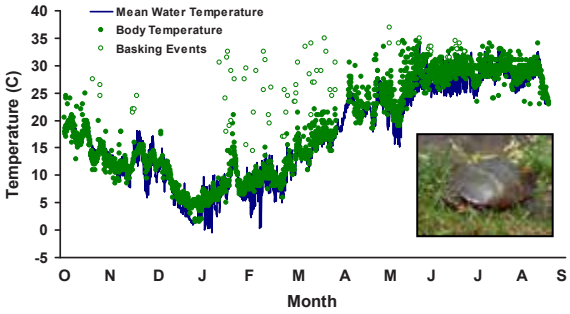


Figure 4-11. Seasonal temperature variation of one male painted turtle, *Chrysemys picta*, over 1 yr, monitored using a Thermochron iButton (Grayson and Dorcas 2004). Water temperatures were measured using Onset Tidbit dataloggers. Basking temperatures were designated as those 6 C greater than water temperature.

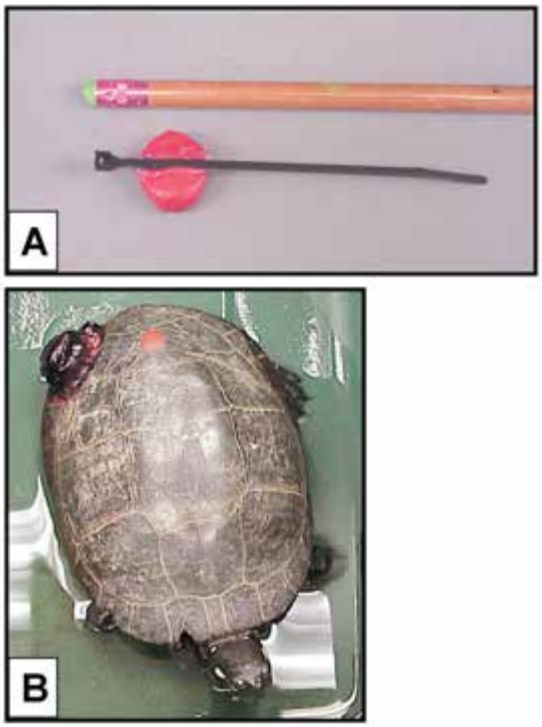


Figure 4-12. Attachment techniques used by Grayson and Dorcas (2004). A) Thermochron iButtons were programmed, dipped in plastic tool dip and then attached to a UV-resistant cable tie. B) Dataloggers were attached to the posterior carapace by drilling two small holes in the marginal scutes.

Automated monitoring of PIT tags

Passive integrated transponders (or PIT tags) have become a widely used method of individually marking a variety of animals (Gibbons and Andrews 2004; see Appendix III on marking amphibians and reptiles). Some researchers have developed or used automated systems for monitoring animals implanted with PIT tags. These include studies of fish (Prentice et al. 1990), voles (Harper and Batzli 1996), and bats (Kunz 2001). To monitor animals with implanted or attached PIT tags, a PIT tag reader must be placed in an opening or area which the animal is expected to move through. Boarman et al. (1998) have used Psion dataloggers to develop an automated system that reads PIT tags to monitor the movements of desert tortoises (*Gopherus agassizii*) when diverted under highways through culverts. Each time a tortoise passed over the reader's detecting coil, the system recorded the PIT tag identification number, time of day, date, and duration of time the tag was within reading distance of the coil. Gruber (2004) used an automated PIT tag reading system to monitor the activity of geckos (*Gehyra variegata*). Geckos with PIT tags glued to them were detected when they moved across a PIT tag reading coil encircling the base of a tree.

Other situations may provide ideal opportunities for monitoring the activities of amphibians and reptiles using certain resources. The hibernacula of some snakes, especially in northern latitudes, may have discrete openings through which the movements of snakes implanted with PIT tags could be automatically monitored relatively easily. PIT-tagged amphibians and reptiles passing through openings in drift fences (Gibbons and Semlitsch 1981) could also be monitored using automated PIT tag readers. For studies of most amphibian populations, this is impractical as a result of expense or effort. However, automated systems may prove useful in intensive studies of small populations of amphibians (e.g., *Rana sevosa*) that breed at relatively small and discrete wetlands.

Automated Global Positioning Systems

Elaborate automated systems for monitoring animals with attached global positioning systems (GPS) have been developed and used to automatically track many species of large mammals and some birds (Rogers 2001). Several types of systems have been developed for transmitting location data from the GPS to the investigator. Some systems use radio signals to transmit data to the investigator, whereas more sophisticated systems use satellite links (Argos satellite system) to retrieve data from animals, and others store data on board the GPS unit until it is retrieved. For many investigators, costs are prohibitive. Prices for units that allow transmission of data via radio signals or satellite link range from approximately $3500 to $5000 US each. Systems that store data onboard are considerably less (approximately $2000-$2500).

Historically, because of the cost and weight of current systems, only large reptiles have been suitable for

GPS monitoring. Thus, the use of automated GPS in herpetofauna has been limited primarily to studies of sea turtles. However, smaller systems designed for birds are now available that weigh less than 20 grams (TAV-2417 Argos, Telonics, Mesa, AZ) and could be used on some relatively large reptilian species. Satellite GPS systems have been successfully used to monitor movements of immature loggerhead sea turtles (*Caretta caretta*; Cardona et al. 2005). Even fine-scale movements can be monitored, allowing precise data to be collected on location and depth (Yasuda and Arai 2005). Balazs et al. (1996) provides details of how to attach satellite GPS transmitters to the carapace of turtles and similar techniques could be applied to reptilian taxa other than sea turtles. If funding is sufficient, automated GPS techniques could likely be applied to reptilian taxa such as medium to large sized tortoises and possibly even some crocodilians.

Automated recording systems

Automated systems for monitoring vocalizations of animals have been employed extensively to monitor anuran populations (see Chapter 5 – Automated Recording Systems for more on this technique). Automated recording systems (ARS) can be used in place of manual calling surveys or can be used to help optimize manual calling surveys (Peterson and Dorcas 1994). ARS can be used to survey for anuran species in places difficult to access for manual calling surveys and can be left in the field for extended periods of time, thus increasing the probability of detecting a given species. ARS minimize disturbance to calling anurans and provide a permanent sampling record that can be evaluated by multiple experts if required (Mohr and Dorcas 1999; Todd et al. 2003). When combined with information on environmental variation, data from ARS can be used to help optimize monitoring programs based on manual calling surveys (Bridges and Dorcas 2000).

Various systems have been developed and range from simple cassette recorders controlled by a recycling timer to more elaborate, digital recording systems with timing mechanisms that save sound files to a minidisc or memory card. Many systems have voice clocks that give an audible time stamp to each sampling interval. Digital models record the time of recording with each sound file, thus making the voice clock unnecessary. Some dataloggers (e.g., Campbell Scientific) have the ability to control electronic equipment and can be adapted to turn recording devices on and off while simultaneously measuring and recording environmental variables (Dorcas and Foltz 1991). ARS, commonly known as "frogloggers," can be constructed (Peterson and Dorcas 1994; Barichivich 2003) or purchased (Bedford Technical, Colleyville, TX).

We used a system in South Carolina to monitor anuran calling at a Carolina bay during June and July (Bridges and Dorcas 2000). We found that if investigators sampled during recommended sampling periods for manual calling programs, they would detect most species, but would likely miss detecting southern leopard frogs (*Rana sphenocephala*) because this species did not typically call during the summer except between midnight and 0400 hrs (Fig.4-13), a time period well after that recommended for manual sampling. We have also used an automated system to monitor the effects of anthropogenic noise (e.g., airplane) on calling activity of anurans inhabiting a wetland and found that we could effectively monitor airplane noise and calling (C. Steelman, unpublished data).

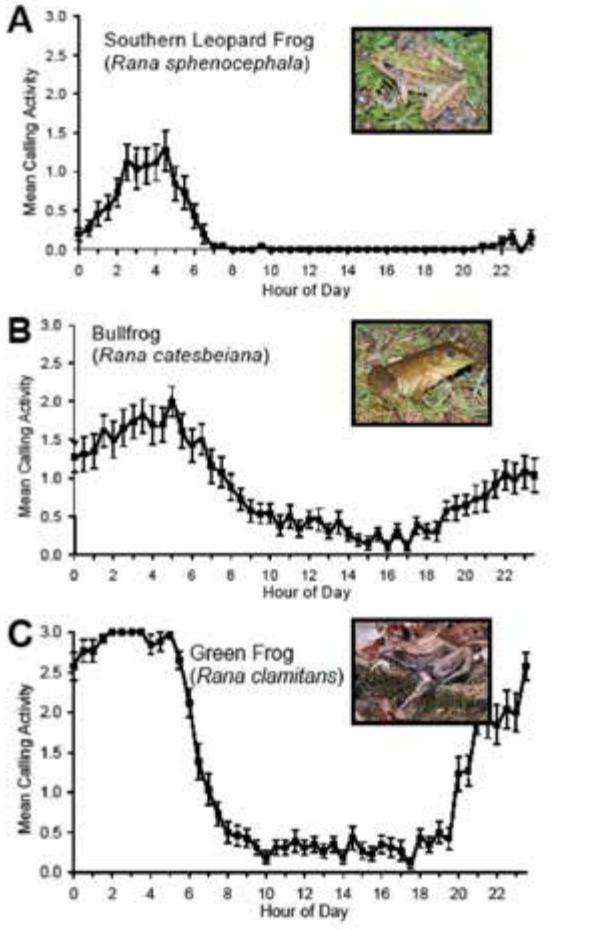


Figure 4-13. Daily calling patterns in the genus *Rana* recorded at Flamingo Bay, Savannah River Site, South Carolina, during the summer of 1997 (Bridges and Dorcas 2000). Mean calling activity was calculated by averaging the recorded calling activity levels (0-3) for each 30-min time period over all 26 days of the study. Error bars denote plus/minus one standard deviation. Note that the most intense calling occurred between midnight and dawn and that southern leopard frogs (*Rana sphenocephala*) called almost exclusively after midnight.

Photographic animal monitoring

The use of automatically controlled cameras provides a unique opportunity to remotely observe the activities of animals, thus providing information on animal activity useful for optimizing inventory and monitoring programs. Additionally, such systems may be used to document the occurrence of rare species unlikely to be detected by manual sampling. Several investigators have used cameras that take a still image when a light beam is broken or when a switch is mechanically triggered. For example, Tuberville and Burke (1994) conducted a study using 35-mm film cameras activated by the breaking of a light beam to detect potential predators of freshwater turtle nests. DeVault and Rhodes (2002) used 35-mm film cameras triggered by a mechanical switch to investigate scavengers in South Carolina. When a dead rat was moved by a scavenger, the camera switch was triggered and the camera took an image of the area. Several snakes were discovered scavenging the dead rats used as bait in the study (T. DeVault, pers. comm.). Guyer et al. (1997) describe in detail the construction of a system that uses a modified digital camera (Canon SureShot Max/Date) in conjunction with a pressure switch plate to monitor vertebrate activity. The use of a digital camera for applications such as this greatly increases the image capacity of these systems and because film developing is not required, greatly reduces costs of operation. Campbell Scientific produces a digital camera (model CC640) for use in harsh environments that can easily be triggered by timing devices or dataloggers.

Numerous investigators have used automatically controlled still and video cameras to examine bird nest predation by various predators, including snakes (e.g., Peterson et al. 2004; Renfrew and Ribic 2003). Most researchers using video have used time-lapse video (2-5 frames/sec) which allows tapes to last a relatively long time (up to 24 hours). However, triggering a video camera to record based on stimuli such as a switch or breaking of a light beam should be possible and may allow the investigator to deploy the system without maintenance for longer periods of time because tape is not used when the switch is not triggered. Furman Diversified produces a wide range of video camera systems specifically designed for use in the field.

Michael Marchand

A well camoflaged camera can prove very useful for automated monitoring of a species

STANDARDS AND DATA MANAGEMENT

Judith L. Greene

Most serious shortfalls in gathering and managing descriptive data on amphibians and reptiles can be avoided through planning and preparation prior to collecting data. The list below includes common issues and problems that need to be addressed when implementing an inventory or monitoring program.

1. Research and study goals and the specific data to be gathered must be clear to all parties involved (e.g., funding agency representatives, researchers, and technicians).

2. Data must be gathered in an organized, consistent manner. Design a datasheet that is objective and simple to use, and which includes all relevant information in sufficient detail. If funds and expertise allow it, invest in personal digital assistants (PDAs) or electronic laboratory notebooks which can be programmed with customized forms for direct data entry in the field (this can help minimize data entry and data transfer errors).

3. All personnel involved must be trained to gather data in the same manner. Attention to detail and consistency are paramount. Handwriting must be legible.

4. Store data routinely in one place until the data can be entered into a database. Keep electronic backups or photocopies of the originals in a different secure location. More than one person should be familiar with the procedure and storage locations.

5. Consider how the data will be used and then enter the data into an appropriately designed database. A spreadsheet such as Microsoft-Excel is adequate for many straightforward datasets. Microsoft-Access may be a better option if the data are a subset of a bigger relational database. Copy the data on a weekly basis at minimum to a portable storage medium and keep the files in a separate location.

6. Review the data and the data management system early in the process and then periodically on a regular basis. This will allow early detection of errors and inconsistencies, which can be identified and corrected before valuable information is lost.

7. One competent, detail-oriented person should oversee the entire process from data collection to data entry to data storage.

PERMITS

Judith L. Greene

Prior to undertaking any inventory or monitoring project, the researcher must procure the appropriate permits required by law (state and/or federal) for the animals in question.

Permitting requirements vary from state to state, and information for most, if not all, states is now available on the internet. There is usually some minimal handling fee charged. In addition, some animals have been granted special status at the state or federal level and may require special permits. Researchers should also be aware that some sites, such as parks and wildlife refuges, are restricted areas and require special access and collecting permits in addition to state collecting permits.

The time period to receive routine permits or to receive approval to work with protected species can be quite lengthy, so it behooves the researcher to determine needs and to apply for the necessary permits as soon as possible. It is imperative that the rules, regulations, and reporting requirements be adhered to strictly. Failure to do so, particularly with species of special status, may result in fines, revocation of the permit, or worse.

Gopher frog (*Rana capito*)

CHAPTER 5. STANDARD TECHNIQUES FOR INVENTORY AND MONITORING

INTRODUCTION

Gabrielle J. Graeter

This chapter serves as a source of succinct summaries of the techniques available for doing inventory or monitoring with amphibians and reptiles. We have not described or invented new techniques; rather, we discuss those that have been proven effective. We have organized the techniques into three classes - active sampling, easy passive sampling, and intensive passive sampling - which represent the level of sampling intensity (modeled after Dodd 2003). The summary for each technique provides a general description of the method and its utility, the limitations, potential biases, and suggestions for how to alleviate these issues, a distinction between the use of the technique for inventory versus monitoring programs, and some recommendations for use of the technique. The main objective of each section is to provide an accurate and realistic description of the method, while also providing helpful key references on the topic. Since the requirements and study design will differ between inventory and monitoring for a particular technique, the distinction between these two sampling modes should be helpful during the planning stages of a program.

Time- and area-constrained searches and quadrat and transect sampling can each stand alone as sampling techniques for herpetofauna, yet they are unique in that each of these techniques has a design component, thereby allowing for application to other techniques. In other words, a researcher can set up a transect survey in an area for a particular species and they can also apply the transect concept to other techniques, such as snorkeling, aerial surveys, or placement of artificial cover, as part of the study design. These four techniques are outlined first so that you can consider their application to subsequent techniques in the chapter.

The Active Sampling class includes sections on hand collecting, visual encounter surveys, dipnetting and sweep samples, stovepipe sampling, kick sampling, electroshocking, egg mass and nest counts, snorkeling surveys, auditory surveys, basking surveys, sign and tracking, road cruising, and aerial surveys. Easy Passive Sampling has sections on artificial cover, PVC pipe surveys, leaf-litterbag surveys, and automated recording devices. Aquatic and terrestrial funnel trapping, terrestrial drift fences and pitfall traps sampling at snake hibernacula are included under Intensive Passive Sampling. A few standard research techniques have some applicability to monitoring amphibians and reptiles (e.g., radio-tracking), so we

TECHNIQUES COVERED IN THIS CHAPTER

It is important to distinguish between techniques that encompass design concepts, such as time- and area-constrained searches, quadrat sampling, and transect surveys which can be applied to many other sampling techniques, and those techniques that are more stand-alone and specific. For example, a technique like time-constrained searching has a design component that can be applied to many other sampling techniques, such as dipnetting and egg mass counts. Similarly, a snorkeling survey may involve using a transect as part of the design.

Techniques with a Design Component:
Time-constrained searches
Area-constrained searches
Quadrat sampling

Additional Topics covered:
Tracking techniques applicable to monitoring
Sampling techniques for exotic amphibians and reptiles

Specific Techniques:
Visual encounter surveys
Hand collecting
Dipnetting & sweep samples
Kick sampling

Egg mass and nest counts
Snorkeling surveys
Auditory surveys
Basking surveys & basking traps
Sign and tracking
Road cruising
Aerial surveys
Artificial cover

PVC pipe surveys
Leaf-litterbag surveys
Automated recording devices
Funnel trapping
Aquatic & terrestrial drift fences & pitfall traps
Stovepipe sampling
Electroshocking
Sampling at snake hibernacula

include a brief section describing these supplementary techniques. Lastly, because most regions in the United States are dealing to some extent with issues regarding exotic herpetofauna, we provide some pointers on sampling techniques for these animals.

The importance of fully educating oneself on the study topics and species cannot be overemphasized. Reading additional resources (e.g., regional guidebooks, applicable literature), consulting with local herpetologists and experts, as well as attending hands-on training workshops about these techniques is an essential part of the learning process. Furthermore, new techniques are always being discovered and developed, and some of these may eventually be proven very effective. For example, researchers are experimenting with the use of dogs for tracking and locating fossorial snakes, but this technique has yet to be accepted as a standard practice.

An extensive table of recommended techniques for sampling the native reptile and amphibian species of the U.S. and Canada, specific to life-stage, is provided at the end of this chapter. The recommended techniques are broken down into four categories, according to the definitions given in Chapter 1 for the two sampling intensity levels of inventory (rapid assessment and comprehensive survey) and two levels of monitoring (presence/absence [i.e., detection/non-detection] and population status). The Species x Techniques Table (Table 5.1) should serve as a guide to which techniques may be most appropriate for a particular species in a particular region, but not necessarily viewed in a rigid manner. However, the techniques listed in this table were suggested and reviewed by numerous experts, and have been well thought out. We suggest you first consult the table for ideas and then investigate the techniques suggested to determine their appropriateness for your objectives and the conditions in which you are working (e.g., habitat, environmental factors, availability of resources).

SELECTION OF TECHNIQUES

Gabrielle J. Graeter

To select a technique suitable for the objectives of a study, the detectability of the amphibians and reptiles of interest and their habitats and activity patterns need to be carefully considered. As mentioned in Chapter 4, environmental conditions, fluctuations, and seasonality need to be considered during the planning and design process. For example, the type of technique you choose to use will depend on whether it will effectively sample a particular species during the sampling period. Other techniques may be more applicable under certain conditions. For example, the amount of precipitation and the humidity and temperature will affect the activity patterns of many

salamanders, so some techniques will have a better capture rate than others.

In addition to considering the effectiveness of a technique according to environmental, seasonal, and habitat-related factors, the cost, time, and resources required must be taken into account when deciding which technique(s) to use. A good example of the tradeoffs involved and factors that should be considered when selecting a technique is provided in a table by Heyer et al. (1994, p.77). Some techniques can be expensive to install, implement, and maintain (e.g., drift fences), whereas others require a lot of personnel to yield useful information (e.g., auditory surveys). Often, the more time- and resource-intensive methods do provide more information and can be used for more robust statistical analyses, but that type of data may be unnecessary for many inventory and monitoring programs. The techniques used should depend on the program goals, the effectiveness of the technique under the given conditions, and the resources available. Thus, in many cases, the simpler, less intensive methods may be the most appropriate because they provide the needed information, yet are affordable and feasible with the resources available.

While each of the following techniques can be implemented as a singular technique, in many cases it is preferable to diversify the techniques used (Gunzburger 2007). Some techniques are commonly used in conjuction with one another. For example, egg mass counts and auditory surveys are often used in combination (e.g., Corn et al 2000), as are funnel traps and drift fences. Using multiple techniques may be necessary when study objectives involve detecting species with varying life history characteristics, habitat use, and behavior, or if they are rare or uncommon in the landscape.

Steps involved in Technique Selection

Step 1: Determine study objectives and questions

Step 2: Consider study design and associated outcome (data and analysis)

Step 3: Make a list of potential useful techniques with consideration to the following constraints: 1) species detectability, 2) habitat, environmental, and seasonal factors, and 3) cost, time, and resources

Step 4: Select techniques to use that meet study objectives, provide the desired outcome, and fit within constraints.

TECHNIQUES WITH A DESIGN COMPONENT

TIME-CONSTRAINED SEARCHES

Brian D. Todd

Time-constrained searches (TCS) can be applied to many monitoring and inventory techniques, including visual encounter surveys, dipnetting, hand collection, and others. The basic premise of this technique is to actively search for animals in a given area or habitat for a predetermined amount of time. The presence and abundance of different species, sexes, and life stages of amphibians and reptiles are recorded as the animals are observed. The time it takes to measure, mark, or record captured or observed animals is not counted against the initial time allotted in the predetermined time constraint. As an example, a 30-minute time-constrained search may take well over 30 minutes if several animals are collected or observed. However, the time spent actively searching must only last for the allotted 30 minutes. Results from a time-constrained search are reported as number of species collected per person-hour. The amount of area covered during a time-constrained search is often dependent on the species of interest. For small woodland salamanders, an area of a few square meters may require 30 minutes to survey intensively. However, for larger species such as some colubrid snakes, a 30-minute search may cover a hectare.

Jamie Bettaso

Conducting time constrained surveys in Northwest California.

Utility

Time-constrained searches are typically used in terrestrial surveys, although they are occasionally used in aquatic habitats. Time-constrained searches

are also frequently used to standardize amphibian surveys, particularly for woodland and stream salamanders (Petranka et al. 1994; Herbeck and Larsen 1999), but also for anurans. However, time-constrained searches can be appropriately applied to nearly all terrestrial herpetofauna and have been used for lizards (Gienger et al. 2002) and snakes (Oliveira and Martins 2001). Time-constrained searches identify the presence of species and can also help reveal life history information such as timing of reproduction and egg deposition, larval presence, and activity patterns and habitat usage.

Limitations

Time-constrained searches have several limitations. Importantly, searches may require significant time commitments by survey participants. Very long periods of time may be required to thoroughly sample species-rich habitats. One must also be aware that periods of active, short-term sampling such as time-constrained searches may be heavily influenced by environmental factors, animal activity, time of day, season, and interactions between these factors. For example, amphibians are intimately associated with rain; weather just prior to, and during searches, may influence the likelihood of correctly identifying the presence of a species. Likewise, environmental variability may influence the number of animals captured during time-constrained surveys, possibly biasing abundance results. Some species are also seasonally active. Pond-breeding salamanders are more frequently encountered during their breeding migrations and many snakes and lizards are seasonally active. Therefore, changes in abundance over time, or among years, may have more to do with the timing of surveys and environmental conditions during surveys than with changes in population status of the species. For these reasons, it is highly recommended to record environmental conditions during the survey, including, at a minimum: temperature, humidity, rainfall the previous day, and rainfall the day of the survey.

Observer bias is yet another possible complication in time-constrained searches. People experienced with the study area or species of interest will often be more likely to encounter animals during the search than novices or participants unfamiliar with the habitat or study species. This needs to be kept in mind when designing studies, especially for the purpose of comparing abundance by habitat, season, or year. Lastly, when comparing multiple time-constrained surveys or designing a study with repeated surveys, it is important that the same type of active sampling be conducted during each survey to standardize the results. In other words, if cover objects are searched and animals are hand collected in the first surveys, the same methods should be followed in subsequent surveys.

Inventory

The goal of time-constrained searches when used for inventories is to maximize the number of species accumulated in a given region or habitat in a standardized format. Due to the inherent short-term nature of time-constrained sampling and the role of weather in determining animal captures, it may be necessary to repeat the sampling to encompass several days with different climate conditions and/or seasons to maximize the chance of accumulating many species. Very rarely will a single time-constrained search yield a complete species inventory for a given habitat. In fact, constraining the amount of time spent searching may be undesirable when a thorough inventory is the objective. Nonetheless, species accumulation rates from time-constrained searches may still be useful for comparing habitats.

Monitoring

It is critically important to standardize the amount of time and methods used during time-constrained searches conducted for monitoring programs. The same amount of time should be expended conducting the initial TCS as in each subsequent search. Likewise, the same level of effort and methods should be used across all searches. If all leaf litter and cover objects are turned and disturbed, this should be repeated in all subsequent searches.

As discussed previously, weather can play an important role in the number of species and individuals captured during a TCS. Therefore, careful consideration must go into research design when using TCS to monitor animal populations. It is possible that perceived changes in abundance or species composition are a reflection of the timing and environmental conditions of TCS and not representative of true changes in population status. Therefore, repeated TCS over consecutive days and across seasons are recommended to reduce the effects of chance variation in animal captures.

Conclusions and recommendations

Time-constrained sampling can be a very useful technique for both inventory and monitoring programs. However, for this technique to prove useful, time spent searching and methods used must be standardized and the sampling protocol must take environmental factors, animal activity, and potential observer bias

into account. Before undertaking a time-constrained sampling program, it is a good idea to consult with a regional herpetologist to determine the best sampling protocol for the particular species of interest that will allow you to meet the goals of your project.

Equipment Checklist

Installation:

Determine study objectives and questions
Flagging/stake flags for marking study area

Sampling:

Data recording materials
Stopwatch
Measuring and marking materials (e.g., scissors, metric ruler)
Containers
Equipment for recording environmental data
(e.g., thermometer, rainguage)

* See Appendix IX for basic equipment lists

AREA-CONSTRAINED SEARCHES

Brian D. Todd

Area-constrained searches (ACS) are very similar to the aforementioned time-constrained search, with the obvious difference that you search a predetermined area rather than search for a predetermined time. Area-constrained searches can be combined with many searching techniques, depending on the goals of the survey. For woodland and stream salamanders, visual encounters, turning cover objects and leaf litter, and hand collection may suffice. However, for larval amphibians in aquatic habitats, dipnetting may be more useful. As with time-constrained searches, the presence and abundance of different species, sexes, and life stages of amphibians and reptiles are typically recorded as the animals are observed.

Valorie Titus

Area-constrained searches can be appropriate in small wetlands

The size of an area that is intensively searched during an ACS can be as small as 1 x 1 m or as large as a 50 x 50 m plot or an entire pond. The size of the area chosen in an ACS typically depends on the habitat type and focal species as well as the time available for completing the search. At most, a search should not last longer than one day. For species-rich habitats where salamander densities may be high (e.g., Appalachian woodlands), a thorough search of all litter and cover objects in a 5 x 5 m area may take a few hours. If your goal is to find and identify large colubrid snakes, the area of interest may be several hectares. Lastly, if you are generating a species inventory for an entire pond or wetland, the pond itself may be used as the areal constraint. Environmental conditions and the amount of time spent searching the constrained area during the ACS should be recorded.

Utility

Area-constrained searches are useful for both aquatic and terrestrial surveys and are generally appropriate for all herpetofaunal taxa, although they are frequently used for sampling salamander populations (e.g., Corn and Bury 1989; Harpole and Haas 1999; Butts and McComb 2000). Area-constrained searches are often used to standardize surveys to ensure that equal portions of a habitat are sampled among sampling intervals and locations. Area-constrained searches identify the presence of species as well as identify life history information such as timing of reproduction and egg deposition, larval presence, and activity patterns and habitat usage. Area-constrained searches are particularly useful because they can generate density estimates of species or individuals per unit area.

Limitations

Area-constrained searches share the same limitations as time-constrained searches and many of the same precautions must be taken when designing studies that implement area-constrained searches. See the section on time-constrained searches for complete discussion of limitations.

Inventory

The goal of area-constrained searches when used for inventories is to maximize the number of species accumulated in a given region or habitat in a standardized format. Due to the inherent short-term nature of area-constrained sampling and the role of weather in determining animal captures, it may be necessary to repeat the sampling to encompass several days with different climate conditions and/or seasons to maxi-

mize the chance of accumulating many species. Very rarely will a single area-constrained search yield a complete species inventory for a given habitat. Additionally, spacing multiple area-constrained searches across a habitat to encompass any heterogeneity will maximize the likelihood of catching different species.

Monitoring

Area-constrained searches are an effective way of standardizing herpetofaunal monitoring surveys and have been used extensively in such capacity (e.g., Brown 2001; Meik et al. 2002; Bailey et al. 2004c). They are particularly well-suited for use in a monitoring program where estimates of animal densities and abundances are required. The same level of effort and methods should be used across all searches, which necessitates using the same size areal constraint across samples. By standardizing the size of area searched, this technique is useful in comparing among-habitat differences in species richness, abundances, or densities. The time required to search areas of the same size may differ among habitats based on topography, extent of available ground cover and leaf litter, or differences in habitat heterogeneity. However, in all cases the defined area must be exhaustively searched.

As discussed in the time-constrained search section, weather can also play an important role in the number of species and individuals captured during an ACS. Therefore, similar consideration must go into research design when using ACS to monitor animal populations. It is not unlikely that perceived changes in abundance or species composition may be a reflection of the timing of, and environmental conditions during the ACS. Repeated ACS over consecutive days and across seasons may reduce any effects of chance variation on animal captures.

Conclusions and recommendations

ACS techniques can be useful for both inventory and monitoring programs and have been used extensively for both purposes. Area-constrained sampling shares many of the same limitations as time-constrained sampling and these constraints must be carefully considered when designing a sampling protocol. As always, it would be wise to consult with a regional herpetologist during the planning process to get advice about project design.

Equipment Checklist

Installation:

- Map of area and design plan
- Flagging/stake flags for marking study area

Sampling:

- Data recording materials
- Measuring and marking materials (e.g., scissors, PIT tags, metric ruler)
- Containers
- Equipment for recording environmental data (e.g., thermometer, rainguage)
- Quadrats for demarcating search zones (if necessary)

* See Appendix IX for basic equipment lists

QUADRAT SAMPLING

Luke A. Fedewa

Quadrat sampling is the use of random sampling arrays within a study area to determine the presence, abundance, and density of representative fauna. A series of randomly chosen quadrats (i.e., squares) are laid out and thoroughly searched for the species of interest (Jaeger and Inger 1994).

Criteria for using the quadrat method (Jaeger and Inger 1994):

1. Animals will not leave the quadrat before being observed
2. Random (not haphazard) placement of quadrats
3. Quadrats are independent

Another sampling method or technique may need to be included in the assessment if one of these criteria cannot be met. In addition, if this methodology is adapted due to taxon specific biology, it is important to justify changes before survey implementation and on all subsequent reports.

A modified version of quadrat sampling, termed patch sampling, involves haphazard placement of sampling arrays within an area due to some known attribute of target species or communities (Jaeger 1994a). Thus, patch sampling is a non-random assessment of relative population size that focuses efforts on microhabitat usage of a particular species or taxon of interest. For example, the redback salamander (*Plethodon cinereus*) is known to defend territories under rocks in forest habitats (Mathis 1990), and in this case, each rock would be considered a patch (Jaeger 1994a). In the case of patch sampling, the patch itself acts as the quadrat for statistical purposes. Both of these methods implement various techniques that increase the

validity and accuracy of population estimates. Quadrat sampling is also discussed some in Chapter 3 (Essentials of Sampling Design) and Chapter 6 (Analysis of Inventory and Monitoring Data) of this book.

Criteria for using the patch sampling method (Jaeger 1994a):

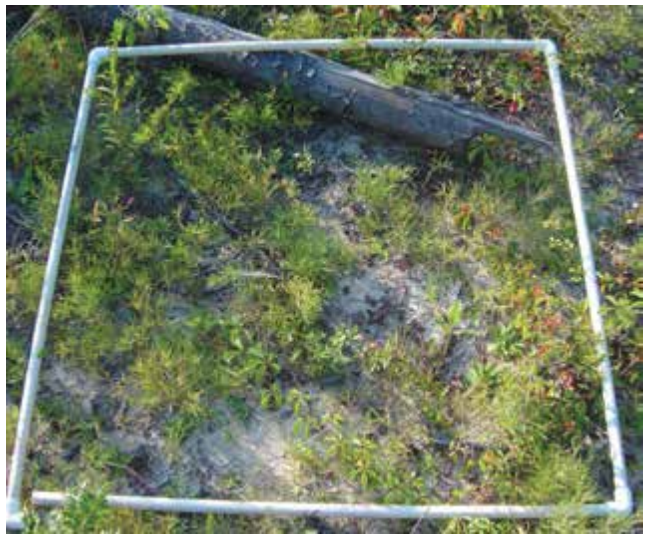
1. Each patch can be defined precisely (e.g., a rock)
2. Patches are operationally definable
3. All patches (or same proportion) equally locatable by observers, in an unbiased manner
4. All individuals in the patch can be located and counted (e.g., no escapees)

Quadrat sampling has been demonstrated to be an effective manner to search for forest ground-dwelling amphibian and reptile species. Rodda and Dean-Bradley (2002) demonstrated a prime example of the quadrat technique. They conducted a multiple site test using randomly placed quadrats throughout forest ecosystems on Pacific and Atlantic islands. The researchers had great success in accumulating data on forest floor-dwelling lizard communities through their total removal technique. Griffis and Jaeger (1998) used quadrat sampling to determine the densities and locations of two terrestrial salamander species (*Plethodon shenandoah* and *P. cinereus*) in Shenandoah National Park, Virginia; they used the proper randomization techniques to select the locations of 50 1-m^2 quadrats throughout the area of interest.

Utility

Quadrat sampling, like transect sampling (see the following section), can be used to assess amphibian and reptile populations at many scales, including at the microhabitat, habitat, and ecosystem levels. However, landscape level data may need to be extrapolated from multiple quadrat studies. Heatwole (2007) advises that quadrat sampling be targeted at species or taxon groups because that is the scale at which the technique is most effective.

Point sampling (Jaeger and Inger 1994) and large fenced quadrat technique (also known as broad sampling; Jaeger and Inger 1994; Heatwole 2007) are the two major types of quadrat sampling used to sample forest dwelling herpetofauna. Point sampling is usually employed when sampling for a single species and when study species are small and densely distributed, whereas broad sampling is used for multi-species studies and for large-bodied, widely dispersed species (Jaeger and Inger 1994). Within the first technique the assessor chooses a point within a predetermined area and randomly selects distances and directions to place 1 x 1 m frames on the forest floor. Then the researcher removes leaf packs, debris, and soil to locate the individuals within each quadrat (see Fig. 5-1 for photo of quadrat).



Gabrielle Graeter

Figure 5-1. Quadrats are often made with PVC piping

The large fenced technique begins by randomly selecting 10 x 10 m quadrats within the study area. The quadrats are enclosed by installing a fence into the ground (see Heatwole 2007) and systematically surveying the species within the fenced area by examining the surface, removing leaf litter, roots, and topsoil. The species are recorded, measured, and collected to achieve demographic and population assessment numbers. When these two techniques are implemented, it is important to properly replicate in order to apply statistical analysis to your conclusions.

The following guidelines will aid in the application with this methodology

1. Use the appropriate sampling techniques within each quadrat. The Reading (1997) assessment is a good example.
2. Record environmental conditions to ensure that results are not biased toward meteorological fluctuations.
3. Target specific species of interest (Heatwole 2003). This will allow you to use specific techniques that will enhance your success.

Limitations

The quadrat sampling method is an effective tool to gain accurate abundance and density estimates for amphibian and reptile populations within forest floor

communities. The following limitations should be kept in mind when designing and implementing this method:

1. This method will not be suitable for some species (e.g., turtles, large snakes, and alligators).

2. The method may only record a certain portion of the population due to sexual or life stage differences in niche selection (Heatwole 1989).

3. Environmental fluctuations and taxon-specific behavior may confound results.

4. When the three criteria for using quadrat sampling (in box on page 66) are not met, biases can result. For example, if species are mobile and not contained within each quadrat throughout the sampling effort, then the estimates of abundance and density will likely be biased.

Inventory

When using this technique for inventory purposes, particularly where interest lies in the differences among different areas or habitat types at a given time, the locations of the quadrats should be determined and then sampled in a random sequence (Jaeger and Inger 1994). This can be a very effective technique for inventory purposes, although it can be quite time-consuming, depending on the size of the area and number of quadrats required.

Monitoring

Heatwole (2007) has reviewed quadrat sampling as a technique for monitoring reptiles and recommended that it be used for those that are forest floor dwelling if population estimates are the end goal. Other adaptive methods (e.g., patch sampling) can be used to gain relative abundance data to assess a population that cannot be monitored via strict adherence to quadrat or transect methodologies. This technique is effective for determining abundance and density of species and populations over time when the criteria for random sampling and independence of data sets are met. Quadrat sampling can also be useful for investigating spatial patterns of a species or taxa.

In addition to randomizing the placement of quadrats, the quadrats should be sampled in a random order to minimize the effects of local environmental conditions (Jaeger and Inger 1994). For monitoring changes in an area over time, consider whether the animals will be removed from the quadrats during sampling. If animals are removed, different randomly selected quadrats should be used the next time; however, if they are not removed, the same quadrats can be reused (Jaeger and Inger 1994).

Conclusions and recommendations

The application of quadrat sampling can be applied to assess forest floor dwelling amphibian and reptile communities. In addition, adaptations such as patch sampling can be used to gain relative abundance data that may not be achievable through other techniques. The strict quadrat method should not be applied outside of forest dwelling herpetofauna unless the changes are justified due to the biology of target species and/or taxon groups (such as specific microhabitat requirements).

Equipment Checklist

Installation:

- Map of area and design plan
- Flagging/stake flags for marking study area
- Fence material (for "broad sampling")
- Tools for installing fencing (for "broad sampling")

Sampling:

- 1 x 1 m quadrat (for "point sampling")
- Snake sticks and rakes for removing litter and debris in sampling plots
- Data recording materials
- Equipment for recording environmental data
- Measuring and marking materials (e.g., scissors, PIT tags, metric ruler)
- Containers
- Flashlight or headlamp if sampling at night

* See Appendix IX for basic equipment lists

TRANSECT SURVEYS

Luke A. Fedewa

Line transect sampling is a powerful tool used by managers and scientists for inventorying and monitoring herpetological communities across environmental gradients or within a single habitat (Jaeger 1994b). To set up a line transect, a string is laid out and then either the entire narrow strip is searched for animals or, if the transect is long, subsets are searched at randomly selected points along each transect. If a properly randomized design is used (i.e., transect placement, subsections to be sampled, sampling order), this technique effectively determines the number of species, relative abundances, and densities across an environmental feature (e.g., elevation, habitat gra-

dients). In fact, this method is better suited to gradient studies than are the visual encounter surveys (see section on Visual encounter surveys; Jaeger 1994b).

Transect sampling is a systematic, repeatable method used to measure population size and structure within animal communities. The scale can encompass the following environmental stratifications: habitat, ecosystem, and landscape. The focus of most transect efforts will pertain to habitat levels. Ecosystems and landscapes may become a focal point during larger surveys by implementing other techniques such as aerial surveys (Mourao et al. 2000).

Transect sampling is often misused, as a concept and in its use, and care should be taken when implementing it as a sampling technique in order to optimize amphibian and reptile population and community estimates. The theory and practice of line transect sampling has been outlined by Jaeger (1994b), Burnham et al. (1980), and Anderson et al. (1979).

Utility

When applying the line transect method toward community or population analysis, it is important to work within the following assumptions:

1. Animals are randomly distributed throughout the transect
2. Transect lines must be randomly chosen
3. Animals on transect lines will not be missed
4. Animals will not be counted twice during each run of the transect and are independent
5. Animal distances from the transect line and angles are precise (Burnham et al. 1980)

This method has three major types of applications: it can be used 1) to assess differences across habitats for the entire study area (i.e., along a gradient) so that replicate transects are placed parallel to the gradient (e.g., lines run from the creek to the uplands or from low elevation to higher elevation); 2) to assess changes in species parameters along the gradient, replicate transects are placed perpendicular to the gradient (e.g., in the stream to uplands example, transects would be parallel to the stream with multiple transects at increasing distances from the stream); and 3) in homogeneous areas, transects are sometimes used. However, in this case, quadrat sampling is a more effective technique (see Quadrat sampling section; see Jaeger 1994b for a detailed explanation).

A good application of the line transect methodology was implemented by Cassey and Ussher (1999) during a population census of the endangered tuatara (*Sphenodon* sp.) that has been extirpated from most of New Zealand by non-indigenous wildlife. Through a well-planned program, they were able to attain a working population assessment of an endangered reptile. Though this method may be inappropriate for herpetofauna that perform behaviors or have ecology that may make them more secretive, it has been proven effective within the context of the biology of the tuatara. An example of a study that examined the numbers and density of salamanders from streams to upland areas is provided by Hairston (1980).

Guidelines for use

Consider the following when applying the transect methodology

Different species require different collection techniques
Line transect methodology using visual analysis may be ineffective for sampling secretive species. Other techniques such as coverboards, drift fence arrays, or aquatic traps may be used if they are implemented under the outlined assumptions in a systematic manner (i.e., randomly assigned traps placed in a systematic manner along a transcect).

Use multiple transect lines (i.e., replication) to ensure that population estimates are accurate (Burnham et al. 1980).

Transect lines should be assigned in a random pattern (using a random number table) to avoid biases (Jaeger 1994b).
It is also necessary to proportionately study habitat types according to their presentation within the given study area. This should be done so that your overall results will represent the habitat and ecosystem parameters that may influence species relative abundance.

Researchers and managers should not adapt sampling methodology based on perceptions of success or failure after the sampling effort has been initiated.
It is more important to accumulate distributional data within the sampling area that it is to capture individuals.

Seasonal, temporal and meteorological variants should be recorded and analyzed to examine potential biases (Burnham et al. 1980).

At times, transect methodology may need to be adjusted due to the biology of a particular species. For example, in assessing a population of ground skinks (*Scincella lateralis*) in the southeastern United States (Akin 1998), the transect lines were placed systematically due to the species' mobility patterns. Similarly, an estimation of the density of the fossorial Red Hills salamander (*Phaeognathus hubrichti*) was determined by counting the number of burrows along line transects (Dodd 1990). A combination of the quadrat

and transect techniques may also prove useful for certain situations; see Messerre and Ducey (1998) for an example of how to do this.

Limitations

This method has a variety of potential problems when applied to amphibian and reptile community assessment and monitoring and each should be recognized and appropriately mitigated before sampling is initiated:

1. Some species may not be detected with this technique. Be sure this technique is suitable for the species of interest.

2. Potential for observer bias (Taylor and Winder 1997). This can be minimized by randomizing observer sampling assignments (Jaeger 1994b).

3. Species may flee or hide from investigator (Rodda et al. 2001). An understanding of the species' natural history and behavior is important. Sampling subsections of each transect may mitigate this issue somewhat (Jaeger 1994b).

4 Assumptions and criteria may be violated: for example, species may not be randomly distributed throughout study area, so might need to choose a different technique such as patch sampling. Also, it may be impossible to have multiple transect lines (e.g., when sampling one narrow stream).

The costs associated with this method will vary depending on sampling techniques used within the line transect methodology. The primary costs incurred from this method will be in the amount of personnel hours required due to the repetition involved in traversing the transect lines. However, costs may increase if other taxon-specific capture strategies are necessary. For example, if aquatic snake communities need to be sampled, aquatic snake traps would need to be purchased and checked daily for captures.

Inventory

When program objectives involve a comparison of the number of species, abundance, or density at a given time at multiple areas, each area is sampled one time. Replication within each area is essential for obtaining accurate population estimates. For transects that are relatively short, the entire transect can be sampled, whereas sections of each transect will need to be randomly chosen and sampled for longer transects.

Monitoring

To monitor the differences in a population over time in a specific area, randomly chosen transects should be sampled at each sampling period, with the constraint that transects that were previously sampled are not resampled (Jaeger 1994b). This is an excellent method for monitoring temporal changes, provided that care is taken to use proper randomization.

Conclusions and recommendations

When conducted properly, with the objective of studying changes in a species or population along an environmental gradient, line transects can be an effective method for assessing amphibian and reptile abundance and density. Accurate population assessments of most herpetofauna can be achieved through the proper application of transect methodology (outlined by Burnham et al. 1980). Random placement, selection, and sampling of transect lines in the given study area, along with the necessary replication, should help ensure optimal results. Furthermore, the target species or taxonomic group should be well studied before the line-transect is implemented. Thus, any violations of the five assumptions previously listed must be justified by the species' biology or ecology. If the method cannot be adequately adapted to the desired taxon, another method should be employed. In addition, multiple methods should be used when assessing overall herpetological communities to ensure that the optimal results are reached and species are not omitted (Ryan et al. 2002).

Equipment Checklist

Installation:

- Map of area and design plan
- Flagging/stake flags for marking study area
- String
- Meter tape
- Compass

Sampling:

- Data recording materials
- Equipment for recording environmental data (e.g., thermometer, raingauge)
- Measuring and marking materials (e.g., scissors, metric ruler)
- Containers
- Meter tape
- Flashlight or headlamp if sampling at night

* See Appendix IX for basic equipment lists

ACTIVE SAMPLING

HAND COLLECTING

Brian D. Todd

The most important aspect of collecting herpetofauna by hand is being able to recognize and distinguish venomous species from harmless ones. Under no circumstances should venomous animals be free-handled or hand-captured. Only experienced and trained professionals should attempt to capture and handle venomous snakes, and only then if it is absolutely necessary. Fortunately, "hand collecting" encompasses the use of many tools and utensils that allow for the safe capture of venomous animals, including snake hooks and snake tongs, described below.

Capture methods and tools

Hand collection in its simplest form is the literal capture and collection of harmless reptiles and amphibians by hand for further study and investigation (see Conant and Collins 1998). However, hand collection also encompasses the use of several tools that can facilitate the capture of herpetofauna. Perhaps the most frequently used tools are snake hooks and snake tongs (Fig. 5-2). Both hooks and tongs are useful for manipulating venomous snakes and keeping them at a distance from the researcher. They are not well-suited for handling harmless colubrid snakes due to these animals' slender bodies, agility, and quickness. Snake hooks are also useful for lifting and overturning cover objects at ground level. Using a hook to flip a cover object protects the researcher's hands from venomous snakes, as well as centipedes and other potentially harmful animals underneath. Snake hooks can be purchased from several leading herpetological or research companies and crude hooks can be constructed at home. However, tongs are more difficult to manufacture and are easily purchased from a supply company.

Gabrielle Graeter

Figure 5-2. Snake hooks and tongs

A common technique used to collect many terrestrial amphibians and fossorial reptiles when hand collecting is searching through ground litter. Many reptiles and amphibians are highly cryptic and searching through leaves and pinestraw and checking under cover objects often reveals hidden animals (see Fig.5-3). Potato rakes and garden rakes are useful in removing dense leaf litter and vegetation from the ground, but can be quite destructive to the habitat and possibly injurious to delicate and small animals. It is generally recommended to leave searched habitats as undisturbed as possible, including returning cover objects such as rocks and logs as they were found. This preserves the quality of the microhabitat and its suitability for future use by other animals.

Joe Mitchell

Figure 5-3. Searching under logs is one tactic employed for salamanders.

Lizards

Lizards that are difficult to capture barehanded can often be hand collected using one of two methods. First, a small lizard noose can be constructed of rigid thread, dental floss, or fishing line and attached to a bamboo pole or other long, slender stick. Telescoping fishing "crappie poles" make especially good lizard noose poles. Many lizards will initially flee when approached, but often have the habit of freezing shortly thereafter. When the lizard has stopped fleeing, the noose on the end of the stick can often be gently slipped around the lizard's head from a distance without causing any alarm to the animal. The pole is quickly snatched upward and the animal is thus captured by the researcher. This technique takes considerable practice but can be very effective. The second method is the use of blowguns with cork projectiles. With practice and careful aim, the cork is shot at the stationary or fleeing lizard, temporarily stunning the animal, allowing the researcher to quickly apprehend it. However, the combination of large projectiles and small quarry can make this method inhumane and result in animal death.

Frogs and aquatic turtles

The capture of many frogs and aquatic turtles requires remarkable agility by the researcher and is often aided by the use of a long handled dipnet (Fig. 5-4). For frogs that remain in place when spotted, slapping a cupped hand down over them can prove effective. It also may help to avoid looking directly at the frog and to move towards the animal very slowly. Snapping turtles and many softshell turtles are easy to capture on land, but must be handled carefully and with respect as they can deliver powerful bites.

Kurt Buhlmann

Figure 5-4. A long-handled dipnet can be useful for capturing turtles and larval amphibians in the water.

Crocodilians (including alligators)

Crocodilians can often be captured with a noose and the help of several people. It is important to securely bind the mouth of crocodilians as soon as possible during capture and transport due to their tremendous jaw strength and the risk of serious injury to researchers. Because many crocodilians can be quite dangerous, capture of these animals should only be attempted by well-experienced researchers. Researchers should also keep their distance from small (young) crocodilians because they are often under the watchful care of a protective mother which may be lurking nearby. Young crocodilians are considered the most dangerous size for this reason.

Handling and housing

Any animals collected from the field must be housed in containers appropriate for transport to the lab. Amphibians should always be kept in moist substrate in containers or sealed plastic bags. A moist paper towel or standing water in the container usually is effective depending on the needs of the species in question but be careful not to drown animals. Small containers with ventilation are useful for holding small snakes, small turtles, and most lizards. Cloth bags of all sizes, including pillow cases, are useful for temporarily holding even the largest lizards, turtles, snakes, and small crocodilians when knotted closed at the open end. Researchers must be careful when transporting venomous snakes in cloth bags to always carry the bag with the hand firmly above the knot and away from the animal. Additionally, researchers should hold the bags well away from their bodies with the contents clearly labeled to prevent injury to themselves and any colleagues. A momentary lapse in concentration can be dangerous when a bagged venomous snake is inadvertently tucked into a waist belt or otherwise carried inappropriately. It is a wise practice to place cloth bags containing venomous snakes into larger plastic buckets with tight-fitting lids. However, one must be careful not to allow the animals to suffocate or be placed in direct sunlight where they can rapidly overheat and die. It is very important to take every precaution to ensure the health and safety of all specimens captured and transported from the field. See Appendix I on Handling Live Amphibians and Reptiles for more information.

Jamie Bettaso

When it is necessary to hold amphibians in captivity, be sure they are kept in the proper conditions; for many amphibian species that means keeping them in containers with moist paper towels.

Sarah Snyder

Turtles should be kept and transported in the proper container to ensure their safety and prevent escape.

Sarah Foster

View of a captured snake in a snake bag

Equipment Checklist

Pre-sampling:

Map of area and design plan
Plan for where to sample

Sampling:

Snake hooks and tongs
Garden rakes (optional and can be destructive)
Cloth bags and containers
Paper towels
Plastic buckets with lids
Nooses for capturing lizards and crocodiles
Blowgun with cork projectile for stunning lizards (optional)
Long-handled dipnets for capturing frogs and aquatic turtles
Data recording materials
Measuring and marking materials (e.g., scissors, metric ruler) if applicable

* See Appendix IX for basic equipment lists

VISUAL ENCOUNTER SURVEYS

Xavier A. Glaudas

A visual encounter survey (VES) is a time-constrained method (Campbell and Christman 1982; Corn and Bury 1990) in which observers sample species richness and/or abundance by walking a pre-defined area in search of reptiles and amphibians. Time spent in the field and numbers of observers need to be recorded if comparisons between areas are to be made. It differs from a transect survey (see Techniques (Ch 5) on transect surveys) in that the survey paths do not need to be a linear transect (Crump and Scott 1994). For example, a VES can be performed by walking along a pond or around a rock outcropping. The VES is used for both inventorying and monitoring of amphibians and reptiles.

Utility

The VES may be the best technique to monitor the occurrence of rare species and species not likely to be caught in traps (Crump and Scott 1994). This technique is especially useful when the objective is to establish the species composition of an area. In contrast, population density cannot be accurately estimated using the VES method solely (Crump and Scott 1994; Funk et al. 2003).

The effectiveness of the VES methods in inventorying and monitoring reptile and amphibian species varies according to habitat types and species habits: habitats with good visibility are more effectively surveyed than habitats with poor visibility (e.g., low- as opposed to high-understory vegetation). Likewise, fossorial and high-canopy species may not be as efficiently detected as terrestrial species that spend most of their time in the open (Crump and Scott 1994).

THREE SAMPLING DESIGNS ARE COMMONLY USED FOR VES (SEE CRUMP AND SCOTT 1994):

1. Randomized-walk: prior to conducting the survey, a series of compass directions, the starting point for the survey, and the distance to be walked from the starting point are randomly selected. All amphibians and reptiles observed within a set distance on each side of the path are then recorded. This design is commonly used to sample large areas.

2. Quadrat: this design is the most appropriate when a given – usually small – area needs to be thoroughly sampled. The quadrat can be systematically surveyed by walking parallel or diagonal pathways within the plot.

3. Transect: one or several linear transects are established within an area and walked systematically by the observer(s). All amphibians and reptiles observed within a set distance on each side of the transect are recorded. This design is commonly used to evaluate potential differences in species richness or abundance across multiple microhabitats.

Limitations

There are several limitations to using the VES method. First, all the microhabitats available in a given area cannot be sampled with the same efficiency. For instance, species living high in trees are more likely to be overlooked than terrestrial species. Second, the efficacy of the VES methods varies by habitat types (e.g., open vs. closed habitat), so that the data collected cannot be compared across habitat types. Finally, time of day and the environmental conditions can influence the activity as well as the detectability of amphibians and reptiles. Therefore, if comparisons are to be made across several sites, observers need to control for these variables.

Assumptions

The VES also has many assumptions (Crump and Scott 1994) and some of them may not be met:

1. Every individual of every species and each species have the same probability of being encountered. The assumption is that there is no intra- or inter-specific variation in morphology, behavior, diel activity, etc. For instance, many species exhibit sexual dimorphism, which would make some of them more conspicuous than others, and that may in turn affect their behavior. Additionally, coloration may be highly variable within and among species, and some individuals and/or species are thus more cryptic than others.

2. Each individual is recorded only once during a survey. This assumption can be met if observers keep track of the movement of the individual (which might be difficult!) or capture the animal and mark it.

3. High inter-observer reliability. This assumes that if two observers were to walk through the same area, they would be expected to have identical results. However, some people are better at finding animals than others. Also, search image is important and some people may be better at finding one particular species than others. Nonetheless, this assumption can be tested by comparing results of observers within the sample area.

Inventory

The VES technique is particularly useful when the objective is to obtain the species composition of an area. The surveys can be accomplished at different levels of intensity. At low-intensity, the method consists of walking and recording the presence of surface-active animals only. However, low-intensity searches may yield very low return per unit time invested because many amphibians and reptiles spend a considerable amount of time under cover (Crump and Scott 1994). Higher intensity surveys are usually more rewarding and consist of recording surface-active animals and animals found under cover objects, as well as opening decaying logs and raking the leaf litter. The major downside of these high-intensity methods is that disturbance to the habitat is greater.

Monitoring

The VES method alone is appropriate to monitor population change in space and time using population indices (i.e., including only the portion of the population that has been observed). However, population indices are frequently biased. In other words, the true population size is different from the expected value of the population estimate (Lancia et al. 1994). If this bias is consistent over space and time, population indices can be used to estimate spatial and temporal population change (Jung et al. 2000). On the other hand, total population size and density cannot be estimated by using only the VES technique because it is unlikely that all the individuals in the studied area will be surveyed (Crump and Scott 1994). Mark-recapture studies can be used in conjunction with VES (Crump and Scott 1994) to estimate these population parameters because the two techniques together allow the calculation of a detection rate, *p*, which eliminates (or reduces) bias (Jung et al. 2000).

Conclusions and recommendations

The VES is a straightforward technique that has been used for a long time and that requires minimal equipment. It is appropriate and useful for inventorying the species present in an area. In terms of monitoring, this technique alone allows the estimation of population indices to examine spatial and temporal population change. Training personnel prior to the survey is crucial to eliminate (or reduce) any possible inter-observer differences in the ability to detect reptiles and amphibians. Also, if comparisons are to be made among sites (only for similar habitats), it is crucial to control for as many factors as possible (including environmental conditions) that may affect the activity and/or detectability of reptiles and amphibians.

Equipment Checklist

Installation:

- Map of area and design plan
- Compass
- Flagging/stake flags for marking study area (e.g., quadrant, transect)

Sampling:

- Compass
- Meter tape
- Snake stick
- Garden rakes (optional and can be destructive)
- Data recording materials
- Measuring and marking materials (e.g., scissors, PIT tags, metric ruler) if needed
- Containers

* See Appendix IX for basic equipment lists

DIPNETTING AND SWEEP SAMPLES

Brian D. Todd

Dipnetting and sweep sampling is when a net is swept through an aquatic habitat in order to capture herpetofauna (Shaffer et al. 1994; Fellers and Freel 1995; Dodd 2003). Dipnetting can be done with many different objectives in mind, including a complete species inventory, or simply to discern whether a particular species is present. Sweep sampling refers to standardizing the dipnetting process so that the number of sweeps is recorded and can be compared among habitats or over time.

Kurt Buhlmann

Dipnets come in a variety of sizes. Diameter of dipnet mesh should be appropriate for target species. Here is a dipnet with very fine mesh that is most useful for catching small salamander larvae.

Utility

Dipnetting is a method useful for sampling herpetofauna in small aquatic habitats. Suitable habitats may include tree holes, sinkholes, springs, puddles, bogs, isolated ponds, Carolina bays, streams, rivers, ephemeral wetlands, permanent wetlands, swamps, and lakes. Large aquatic habitats may require the use of seines and nets (Fig. 5-5). Dipnetting is particularly useful for sampling aquatic amphibians and amphibian larvae as well as some aquatic reptiles. This technique is most effective in small, shallow bodies of water because the smaller the area, the more likely that all herpetofauna present will be captured.

Jamie Bettaso

Figure 5-5. Seining a pond for herpetofauna.

Limitations

The main limitation of this technique is that animals may not be equally catchable, so that some species may be present but not represented and others may be easily captured, thus skewing species inventories or estimates of abundance or density. Similarly, more diverse habitats or a diverse species assemblage may introduce bias into dipnetting monitoring surveys because some species may be very difficult to catch and some habitats may be more difficult to sample than others. The effectiveness of this technique declines with size and depth of the aquatic habitat, such that estimates of abundance in large or deep habitats may be skewed.

Inventory

Dipnetting reveals the presence of many species in aquatic habitats and is especially useful when searching for evidence of amphibian reproduction. The goal in dipnetting during species inventories is to be as thorough as possible when sampling. This is accomplished by searching as many microhabitats as possible in the overall aquatic habitat of interest. This also means conducting enough sweeps of the net through the water to exhaustively reveal the presence of species that occur in the habitat. Because mesh size of the net will affect the speed with which the water can be strained, different nets may be required to sample different species, dependent on the agility of the species and its ability to avoid capture. Some amount of trial and error may be necessary to determine which nets are most appropriate. Lastly, it is advisable to time dipnetting sessions to coincide with portions of the year when species will most likely be found in aquatic habitats. For aquatically-reproducing amphibians, this will occur during the breeding or larval seasons. For aquatic snakes, this may be in the warmer parts of the year when the animals are most active. And for completely aquatic amphibians, this may be many times of the year.

Kurt Buhlmann

Inventory & Monitoring Workshop participants dipnetting in a Carolina bay wetland, Aiken, SC, November 2004.

Monitoring

A key to accurately monitoring animal populations by dipnetting is to ensure that repeated samples have equal sampling effort. One way of ensuring equal sampling effort when dipnetting is to conduct a standard number of sweeps with the dipnet or to record the number of dipnet sweeps used. Alternatively, dipnetting a prescribed, standardized area or using removal sampling (when possible) can provide estimates of animal abundance or density. Another potential bias with dipnetting occurs when animals are not equally catchable. This may not be a problem when monitoring one species in one habitat. However, diverse habitats or diverse species may introduce bias into dipnetting monitoring surveys. Habitats with significant heterogeneity, such as streambeds, may be particularly difficult to sample without introducing bias into the study. Variation in sampling depth or the amount of water strained with the net can also influence the number of animals captured.

Conclusions and recommendations

This can be a very useful and effective technique, particularly if the program involves sampling one or more habitats that are somewhat homogeneous. A careful consideration of the variation in the catchability of different species and the timing of sampling is essential.

Equipment Checklist

Installation:

Map of area and design plan
Flagging/stake flags for marking study area (e.g., quadrant, transect)

Sampling:

Dipnets (of various sizes and mesh sizes)
Seine (optional - only necessary in some habitats)
Data recording materials
Measuring and marking materials (e.g., scissors, PIT tags, metric ruler)
Containers

* See Appendix IX for basic equipment lists

STOVEPIPE SAMPLING

Mary Beth Kolozsvary

Stovepipe sampling is an active, quantitative method for sampling aquatic amphibian larvae that involves trapping animals inside of an enclosure (Schaffer et al. 1994). Once trapped, larvae are removed from the enclosure with a net. This is done repeatedly until all or a high proportion of larvae are removed, identified, and counted. For determining the total number of dipnet sweeps needed, a "giving up" algorithm can be used; the algorithm may vary with the size and dimensions of the specific enclosure. An algorithm commonly used with a typical pipe sampler (approximately 30 cm diameter x 1 m high) is to continue to remove larvae with a net until you get 10 consecutive sweeps that yield no larvae (Skelly pers. comm.).

Jonathan Richardson

Biologist using a pipe sampler in a wetland.

Enclosures can be made of a variety of materials, typically either shaped like a pipe (e.g., air conditioning duct, culvert, stove pipe, PVC pipe; Alford 1986; Skelly 1996) or rectangular box (Harris et al. 1988). For a pipe sampler, larvae are captured by sweeping a dipnet along the edge of the pipe, working from the bottom up through the water column. In contrast, for a box sampler the dipnet is dragged across the substrate along the length of the box sampler and up through the water column to capture the larvae. When placing the sampler in the water, care needs to be taken to be quick and cause minimal disturbance to prevent animals from swimming away before the enclosure is firmly set in the substrate (Shaffer et al. 1994).

Actual sampling locations are identified prior to sampling (by some method of randomization), located along a standard grid, or at random distances along transects. If the number of larvae at a site is to be computed, the number of samples within a habitat type should be in proportion to how much of the entire site is made up of that habitat type. If comparisons are to be made between study sites, the number of samples to be taken at a site should be in proportion to the overall area of the site.

Utility

Stovepipe sampling is time intensive to implement, but yields quantitative estimates of larval densities and can be used to estimate population size. The dimensions of the sampler are needed when extrapolating number of larvae captured to the area of a given habitat type or the overall site. If depth of each sample is recorded, then estimates of number of larvae per unit volume can be calculated. If estimates of larval density are not desired, then a less time intensive sampling method (e.g., dipnetting) may be more time efficient. However, it is important to realize that all sampling methods have biases and it may be best to use multiple sampling techniques that, together, are best suited for the overall objective.

Limitations

Enclosure sampling is limited by the height of the sampling apparatus. Thus, this method is ideal for small, shallow bodies of water. The enclosure needs to have a tight seal with the substrate, or animals can escape. Therefore, substrates (e.g., sand, muck, gravel) that enable a good seal work well, but large, irregular rocks or rock slabs and downed logs and branches interfere with sampling or may prevent this method from being used at certain sites. Pipes are easier to use than box samplers in shrub swamps and other vegetated habitat types. Box samplers are only useful in open basin ponds (Skelly pers. comm.).

Samplers are generally rigid, bulky, and heavy, making it difficult to use this method for sampling at remote sites. These difficulties, however, can be minimized if lighter materials (e.g., air conditioning duct) are used in constructing the sampler. Sampling at larger sites can also be more efficient if several people conduct the sampling while a person remains “on shore” to record data for the team.

Inventory

Stovepipe sampling can be an effective technique for inventorying larval amphibian populations in small, shallow bodies of water with suitable substrate. If the primary goal of a study is to determine presence of a particular species that is rare at sites at which it occurs, then other methods may be more effective. Alternatively, stovepipe sampling can be supplemented with other sampling techniques (e.g., funnel trapping, dipnetting, call surveys).

Monitoring

This sampling method can also be useful for monitoring. However, this technique is time intensive and when larval amphibians occur at very low densities, the number of samples taken is often insufficient for detecting a species at a given site. Thus, valid population size or density estimates of species in very low abundance are difficult to estimate using this method.

Conclusions and recommendations

Stovepipe sampling is an established technique to consider for obtaining population size and densities of aquatic amphibian larvae, as well as to contribute to documenting species presence and overall species richness of a site. Because this sampling method focuses on larval amphibians, stovepipe sampling gives a better indication of actual breeding effort and high potential for reaching metamorphosis than methods that merely document species presence (e.g., call surveys).

Equipment Checklist

Pre-sampling:

- Map or sketch of area to be sampled, by habitat type
- Plan for where to sample

Sampling:

- Stovepipe (or alternative enclosure) sampler
- Dipnet (fitted to sampler)
- Plastic freezer baggies and/or containers (e.g., urine cups) for holding/sorting samples
- Rigid plastic container (for holding plastic baggies/cups with samples; floats the container alongside the sampler)
- Small ruler or caliper (if larval measurements are not to be taken)
- Data recording materials (e.g., field notebook, datasheet, pencils)

* See Appendix IX for basic equipment lists

KICK SAMPLING

Jason L. Jones

Kick sampling (or “rubble rousing”) generally consists of lifting and removing all loose cobble substrate from the stream bottom and hand raking or kicking loose pebbles into a net or a series of nets securely placed downstream of the habitat to be sampled. D-frame nets are most commonly used. Depending upon the environment, hardware cloth screen, Surber nets, or small fish aquarium nets may be used. Block or seine nets can be used in conjunction with D-frame nets to assist in the capture of any individuals missed by the

Jason Jones

Kick Sampling using D-frame nets.

Jason Jones

Examining the catch.

Jason Jones

Tailed frog larvae (*Ascaphus montanus*) in net.

Jason Jones

Underwater view of tailed frog larvae attached to rock in front of net.

nets. Time and area constrained searches are two approaches used in kick sampling (see Time-constrained searches; Dupuis and Friele 2006; and Area-constrained searches). Area constrained kick samples are the most common means of quantifying sampling effort and can be easily compared among habitats and over time (0.1-m²: Hawkins et al. 1988; 1–10-m long: Bury and Corn 1991; 0.6-m long: Welsh et al. 1997; 10-m²: Lohman 2002). Sampling areas are generally placed within stream reaches (100-m – 1,000-m in length). Kick sampling data can be used for species inventory, species occupancy analysis, and species abundance, biomass, and density estimates.

Working in a two-person team appears to be the most efficient method for kick sampling (Bury and Corn 1991). Following the secure placement of the nets, one worker holds the nets firmly in place while the other worker gently dislodges cobble and woody debris, firmly kicking or hand raking through the stream substrate. Any herpetofauna encountered are hand captured or displaced downstream into the secured nets. Nets should be cleaned of debris and checked for herpetofauna in short time intervals (≤5 minutes) to decrease animal stress or mortality (Jones, pers. comm.). It is important to note that any large habitat item (e.g., ≥200 mm sized cobble, large woody debris) dislodged while kick sampling should be repositioned back into the stream to minimize the ecological impact associated with this technique.

Many stream dwelling amphibians are agile, mobile, and commonly avoid capture; to address this issue the entire sample unit of interest must be thoroughly sampled. Moreover, the net and mesh size need to be carefully chosen (e.g., 0.8 mm mesh for *Ascaphus spp.* larvae). Several stream dwelling reptiles and amphibians are long-lived and remain in the stream year round; nevertheless, it is advisable to schedule sampling efforts during the season that the species is most likely to be found in the stream. In addition to timing sampling efforts with species presence, it is also important to time sampling events with annual flow events to allow for adequate sampling depths (see “Limitations”) for capturing the species of interest.

Utility

Kick sampling is a useful method for determining occupancy and abundance for stream dwelling amphibians, particularly in small to intermediate sized, fast flowing streams (1st-3rd order streams: 0.5-m - ≤ 5-m). This technique is most effective in streams that are relatively small and shallow, due to limitations on net heights. Slow flowing streams, pools, or rivers may require the use of snorkeling or alternative

techniques (see Snorkeling surveys and Electrofishing). Kick sampling provides an effective means of sampling complex stream systems (e.g., log snags, cascades, undercut banks). Nets can be obtained that vary in overall size and mesh size and are lightweight, durable, and collapsible for ease in transportation of sampling gear into remote wilderness stream ecosystems. Although a two-person team appears to be the most effective way of thoroughly sampling a stream network, kick sampling can also be accomplished successfully with one person using one larger net or several smaller nets (Lohman 2002).

Kick sampling is particularly useful for sampling headwater stream dwelling amphibian larvae, such as tailed frogs (*Ascaphus spp.*). This technique can also be used for a variety of stream dwelling salamander species, including giant salamanders (*Dicamptodon spp.*), torrent salamanders (*Rhyacotriton spp.*), and a variety of plethodontid salamanders (Bury and Corn 1991; Lowe and Bolger 2002).

Limitations

A major limitation to kick sampling is that herpetofauna may not be equally catchable. More mobile and less abundant species, like the giant salamander, may be present but rarely captured, and therefore underrepresented, while other more abundant species, like the tailed frog, may be easily captured and adequately represented, thus biasing species inventories or abundance and density estimates. Additionally, more complex habitats can be more difficult to sample and thus species inhabiting these habitats may be overlooked.

Although kick sampling may result in relatively unbiased abundance estimates, it is labor intensive, and therefore may not be the most cost effective sampling approach for determining occupancy (Quinn et al. 2007). The effectiveness of kick sampling declines with increasing stream size and depth, such that estimates of abundance in large or deep habitats may be biased. Larger streams (≥3rd order) which are shallow enough to sample may bias abundance and density estimates because as stream width increases, the likelihood of capturing all amphibians present decreases (Jones, pers. comm.). Additionally, larger streams can become less efficient to kick sample; in this case the snorkeling technique may provide a more efficient and effective means of sampling.

A drawback to kick sampling is the technique's potential impact to the stream ecosystem. Dynamic streams experience high annual discharge, which may result in annual disturbance/scouring events in the headwater. The less dynamic the stream, the more kick sampling may add to the overall disturbance regime experienced by the stream, which may affect species richness, abundance, biomass, and density estimates.

Inventory

In general, kick sampling can be used to adequately sample certain amphibian species inhabiting a stream ecosystem. Area-constrained kick samples can result in comparable estimates of abundance, biomass, and density of these amphibian species. When kick sampling during species inventories, one must establish a long enough sample unit (e.g., stream reach) to adequately determine the presence of a species or species assemblage. This may require workers to employ kick sampling through a variety of riffle, cascade, and pool complexes. Additionally, the kick sample units should be placed across all possible habitats, ideally approximating the proportion of habitat where the species are likely to occur (Hawkins et al. 1988), and placed adequately across the stream reach or subbasin so inference can be drawn from the data (see Chapter 3: "Essentials of Sampling Design").

Jason Jones

Ascaphus montanus tadpoles, illustrating age classes and sizes (from left to right: first year age class, second year age class, and three tadpoles in differing stages of development in the third year age class).

Monitoring

Given that kick sampling may cause varying degrees of perturbation on the stream ecosystem, one must use caution when selecting sites for use in monitoring surveys. If established sampling units have been monitored from year to year, exercise caution when comparing estimates of occupancy, and subsequently abundance, biomass, and density. To avoid sampling modified habitat, one can define a sampling unit (i.e., stream reach), and randomly select sub-sample units that are nested within the larger sample unit so as to minimize the chance of repeatedly sampling an area impacted by kick sampling. Additional bias may be introduced when sub-sampling stream units due to the inherent heterogeneity of stream systems. To decrease

bias and ensure equal sampling effort, constrained searches in area or time can be conducted, which should allow for temporal and spatial comparisons.

Conclusions and recommendations

Kick sampling provides an efficient method of determining the presence and abundance of stream dwelling amphibian species, particularly in shallow, fast moving streams. If thoughtfully designed, inventory and monitoring programs can yield insight into species occurrence patterns, age class structuring, larval recruitment rates, and abundance, density, and biomass estimates across multiple spatial scales. Kick sampling may be particularly useful for inventorying purposes, but may have limited use in intensive monitoring programs for specific stream sites (see "Limitations"). Consideration must be given for the timing of sampling (e.g., high water events) and the sampling season so as to avoid biasing the results. The overall affordability, ease of equipment transport, and thoroughness of the kick sampling technique makes it an effective method for assessing amphibian population parameters.

Equipment Checklist

Installation:

- Map of area and design plan with list of stream coordinates
- Flagging/stake flags for marking study area

Sampling:

- D-frame dipnets or Surber nets (of appropriate sizes and mesh sizes)
- Seine net with stakes (optional)
- Data recording materials
- Camera (water resistant or with an underwater casing)
- Tape measure
- Measuring and marking materials (e.g., metric ruler, scales, scissors)
- Containers for holding amphibians (collapsible, light weight ball bucket)
- Hip-waders with boots (e.g., neoprene legs with felt bottom boots)
- GPS
- Stop Watch

* See Appendix IX for basic equipment lists

EGG MASS AND NEST COUNTS

Aaliyah D. Green

Egg mass and nest counts are easy, inexpensive methods used during breeding periods to monitor reproductive activity in reptile and amphibian populations. Egg mass counts involve searching the pond's perimeter, and if possible, the interior, for globular masses of eggs attached to vegetation. Many amphibians reproduce in large aggregations during a specific time window of a few to several weeks. This allows investigators to collect data from several masses in a small area over a short period of time.

Nest counts are usually conducted over a larger area than are egg mass counts, as reptile nests may be more spatially distributed. This method entails searching known nesting grounds (specific to the species of interest) for evidence of underground nests. In the case of gopher tortoises (*Gopherus polyphemus*), burrows can be carefully probed to confirm the presence of a nest (Butler and Hull 1996).

Whit Gibbons

Egg masses come in many shapes and sizes.

Kurt Buhlmann

Some eggs are easily spotted because of large numbers or size.

Whit Gibbons

Some eggs are sheathed in a gelatinous covering.

Whit Gibbons

Some eggs are difficult to see and may be most easily counted in a dish.

Whit Gibbons

Some eggs are laid in strands, an important feature for identifying the species.

Tracey Tuberville

Careful probing of a gopher tortoise burrow can determine the presence of a nest.

Utility

Egg mass and nest counts can provide information on presence, reproduction, and population parameters of amphibian and reptile populations. Egg mass counts are performed in streams, ponds, or seasonal wetlands where adult amphibians assemble to breed (Fig. 5-6). This technique is particularly useful for amphibian species that are explosive breeders or reproduce in communal aggregations, such as wood frogs (*Rana sylvatica*), spotted salamanders (*Ambystoma maculatum*), gray treefrogs (*Hyla versicolor, H. chrysoscelis*), and green frogs (*R. clamitans*) (Crouch and Paton 2000; Dodd 2003). Periodic counts can also be used to identify environmental conditions most conducive to oviposition.

Figure 5-6. Observer bias can be reduced by having more than one person do the survey. Here, four researchers survey for salamander eggs.

Nest counts are conducted to investigate reptile populations, and are most useful for turtles and crocodilians with easily identifiable nests. In some cases, predation can make nests easier to find and count. For example, *Malaclemys terrapin* nests have an extremely high rate of depredation (Feinberg 2004), so counts of these nests provide a close underestimation of the total number of nests.

Limitations

Egg mass counts are not as useful for amphibians that lay eggs on land, and nest counts are not as useful for snakes and lizards. These populations are best studied using other techniques. If conducting a site inventory, egg mass/nest counts do not indicate species that are present but are not breeding. Counting nests or egg masses also offers no information about the number of juveniles, males, or non-breeding females in a population. A single egg mass/nest count does not provide a reliable estimation of population parameters. Counts must be conducted periodically throughout the breeding season to account for new nests/masses.

Egg masses can be inconspicuous and are easily overlooked, especially when they are separate from large aggregations. For some amphibian species, one mass may contain the eggs of more than one female. Conversely, one female may deposit more than one egg mass in a breeding pond. These factors must be considered when egg mass counts are used to estimate population parameters.

Whit Gibbons

Marbled salamanders (*Ambystoma opacum*) lay their eggs on dry ground and guard them. When surveying for their eggs, carefully turn over cover objects in an area that becomes wet on a seasonal basis.

Inventory

During a particular species' reproductive season, counts can often be used to determine if that species is present in a habitat. Egg masses of many amphibian species look similar, so it is important to positively identify the mass when conducting an inventory. If identification cannot be made in the field and no camera is available, researchers should use detailed language to describe the mass so that it can be identified later (Mitchell 2000). Some helpful observations to make are:

- Is the mass globular or round?
- Are the eggs clumped, separated, or on a string?
- What color and shape are the embryos?
- Is the jelly surrounding the eggs firm or loose?
- Is there a film on the surface of the mass?
- To what type of vegetation is the mass attached?

In cases where nests are difficult to find, signs of nesting activity can be used to confirm a species' occurrence. Remnants of nest materials (broken eggshells, dead hatchlings, etc.) indicate the prior presence of a nest. For turtles and other reptiles that bury their eggs, holes left by emerging hatchlings are also indicative of successful nests (Butler and Hull 1996).

Michael Marchand

Predated eggs of a painted turtle, *Chrysemys picta*

Monitoring

Counting egg masses/nests requires little effort and resources compared to other monitoring techniques. To accurately estimate reproductive parameters, counts should be taken periodically throughout the season. As egg masses/nests can be difficult to detect, more than one observer should perform the survey to minimize the number of egg masses/nests missed. Grant et al. (2005) concluded that using two observers to conduct egg mass counts significantly decreased the proportion of egg masses that were overlooked. For species that lay one clutch per season, egg mass/nest counts provide a reliable estimate of the number of breeding females in a population. Crouch and Paton (2000) found a strong correlation between numbers of *R. sylvatica* egg masses counted in breeding ponds and numbers of males and females captured in drift fences surrounding the ponds.

Egg masses and nests can be marked with flags to evaluate spatial distribution of oviposition sites. Marking nests also allows researchers to revisit individual nests after hatching has occurred and determine hatching success. Mazzotti (1999) used nest counts as a part of an extensive monitoring program to conclude that the number and spatial distribution of American crocodile (*Crocodylus acutus*) nests in Florida Bay increased between 1970 and 1995. When marking nests, researchers must take care to cause as little disturbance to the eggs as possible, as this can cause maternal nest abandonment (Mitchell 2000).

Conclusions and recommendations

Egg mass and nest counts can be simple, yet powerful and informative methods for determining presence/absence (i.e., detection/non-detection) and estimating population parameters for some amphibian and reptile species, provided that certain factors are taken into account. Note that these methods are not appropriate for all species (see limitations). Egg mass and nest counts are most effective when the researcher has a solid understanding of a species' natural history (especially the details about reproduction), and has taken the project objectives, timing of the study (e.g., season), and limitations into account during the design phase of a project.

Equipment Checklist

Installation:

Map of area and design plan

Sampling:

Dipnet (if necessary to scoop eggs out and into something)
Dish (for counting eggs in)
Data recording materials
Camera
Stake flags for marking location of nests/eggs
Measuring materials (e.g., metric ruler)
Magnifying glass
Containers
Burrow probe (for gopher tortoise nests)

* See Appendix IX for basic equipment lists

SNORKELING SURVEYS

Kurt A. Buhlmann, Jason L. Jones, and
W. Jeffrey Humphries

Snorkeling is a useful technique for studying amphibians and reptiles in aquatic environments including clear-flowing rivers, headwater streams, clear lakes, and large karst springs. Species that may be detected by snorkeling include a variety of aquatic turtles (Iverson 1977; Graham 1981; K. Buhlmann pers. comm.) and several species of pond and stream dwelling amphibians, ranging in size from tailed frogs (*Ascaphus spp.*) to the large aquatic hellbender salamander (*Cryptobranchus alleganiensis*; Tyler et al. 1998; Adams and Frissel 2001; Nickerson and Krysko 2003; J. Humphries, pers. comm.). The effectiveness of snorkeling surveys depends on the natural history of the target species; species with terrestrial life stages (e.g., tailed frogs) may be underrepresented (Hoffman et al. 2005), whereas species that are aquatic (e.g., some turtles, the hellbender salamander) can be sampled more thoroughly.

Jason Jones

A researcher surveying for tailed frog larvae in Flathead National Forest, Montana

Jason Jones

Tailed frog larvae (*Ascaphus montanus*).

Utility

Depending upon the environment, snorkeling may require a proficiency in swimming ability by the researcher. The technique requires that certain safety protocols be established. Teams of two or three persons should work closely together and maintain eye contact with each other. In small streams, a team of two is sufficient, but in more rapidly flowing rivers, you may need an additional person to follow along shore or in a drifting boat for safety purposes. In sensitive stream and pond ecosystems, snorkeling can provide a low impact means of estimating species occurrence and abundance if the substrate is not substantially altered during surveys. In remote lakes and streams, snorkeling may provide the best estimates of occupancy and densities given the constraints of time and equipment (Tyler et al. 1998).

Large riverine turtles, such as river cooters (*Pseudemys concinna*), are difficult to approach when basking, and will often drop off of their basking sites and quickly travel to deeper flowing water where they will hide among large rocks and rooted aquatic vegetation (Buhlmann and Vaughan 1991). In clear water conditions, river cooters hiding in such areas will remain motionless and can be captured by hand. Being able to reach river cooters in these locations requires mask, snorkel, fins, a boat, and more importantly, a companion for safety. This technique is useful for presence/absence surveys, as well as repeated long-term monitoring of adult members of local populations. Unlike river cooters, many species of map turtles (genus *Graptemys*) will drop to the river bottom from basking sites, but stay nearby. Snorkeling for these turtles is often successful, but one needs to be careful about getting tangled up in log snags that typify this microhabitat (K. Buhlmann, pers. comm.). Snorkeling has also been used successfully to find alligator snapping turtles (*Macrochelys temminckii*) in clear flowing spring runs in Georgia (J. Jensen, pers. comm.) and western pond turtles *(Clemmys marmorata*) in California (J. Holland, pers. comm.).

Snorkeling surveys are useful for estimating occurrence and abundance of amphibians in the deeper offshore areas of ponds and lakes. Surveys can easily be conducted during the day, and can be performed at night with the aid of handheld dive-lights (Hoffman et al. 2005; R. Thurow pers. comm.). Snorkeling surveys have typically been used for estimating larval salamander occurrence and abundance in mountain ponds and lakes in the Pacific Northwest and Rocky Mountains (Olson et al. 1997; Tyler et al. 1998; Brokes 2000; Pilliod and Peterson 2001; Hoffman et al. 2005). During pond and lake surveys, flippers can be used, but are not necessary, and may be more troublesome than helpful (Hoffman et al. 2005).

This technique is also useful for estimating occurrence and abundance of stream dwelling amphibians across a variety of stream sizes and habitats (Adams and Frissell 2001). In fast moving stream systems, snorkeling provides a time efficient measure of occupancy and abundance of tailed frog (Ascaphus spp.) larvae and egg masses (J. Jones, pers. comm.), which are adhered to moderately sized cobble in shallow riffles (Nussebaum et al. 1983). When snorkeling in fast moving streams and rivers, caution should be taken to avoid log snags, cascades, and other hazardous obstacles. Tennis shoes or light weight, felt bottom boots should be worn in the place of fins.

Hellbenders are usually active at night, but hide beneath large rocks during the day (Nickerson and Mays 1973; Peterson 1987; Humphries and Pauley 2000). Snorkeling, coupled with lifting rocks by hand or with the aid of a cant hook (or log peavey), is one of the best methods for finding hellbenders. Diurnal activity of hellbenders is common throughout the summer in some streams (Humphries and Pauley 2005), but activity peaks during the fall breeding season in most populations (Nickerson and Mays 1973; Humphries and Pauley 2005). Snorkeling during periods of high diurnal activity may reveal many hellbenders not able to be seen from above the stream surface, especially in deeper or very fast-moving stream sections. Additionally, snorkeling is the best method for detecting larval and juvenile hellbenders because small size classes are often overlooked using other methods (Nickerson and Mays 1973; Nickerson and Krysko 2003; Humphries and Pauley 2005). Studies on hellbender demographics that do not include snorkeling may underestimate juvenile recruitment, thus providing unreliable data about population health (Humphries and Pauley 2005). Finding larval and juvenile hellbenders requires meticulous inspections of the stream bottom, including the interstices of cobble and gravel beds (Nickerson and Krysko 2003; J. Humphries, pers. comm.). Because many hellbender streams are extremely cold, even during summer, a thick wetsuit and neoprene gloves and boots are usually needed. Flippers are not needed and usually only hamper survey efficiency.

Jeff Humphries

An adult hellbender salamander, *Cryptobranchus alleganiensis*

Jeff Humphries

Researchers snorkeling for hellbender salamanders in Chattahoochee National Forest, Georgia.

Limitations

Snorkeling techniques are limited to relatively clear waters (Helfman 1992). Turbid or silt-laden conditions will not be conducive to snorkeling surveys. In streams, seasonal run-off events and storms can greatly reduce underwater visibility, which can directly influence occupancy and density estimates. If snorkeling is used as a repeatable monitoring method, then we recommend that turbidity measurements (e.g., with a Secci disk) be taken during each monitoring session, so that the numbers of animals observed is not affected by environmental conditions. A Secci disk has black and white sectors and is lowered into the water from the surface. Increasing turbidity will cause the black and white elements to fade into one another and the disk will slowly disappear from sight. A formula determines the amount of turbidity by measuring the depth at which the disk was no longer visible (www.oceanservice.noaa.gov/).

In large, deep ponds and lakes, water depth appears to influence amphibian counts (Brokes 2000). In deep water, SCUBA diving will likely be a much more effective technique than snorkeling. Additionally, the complexity of the habitat can influence the amount of time needed to complete a survey. Therefore, habitat complexity should be taken into account when deciding whether to use time- or area-constrained surveys. In small, shallow streams, snorkeling surveys may require more sampling effort than kick sampling, particularly if copious amounts of large woody debris and other in-stream obstructions impede the snorkeler from crawling upstream. Additionally, stream snorkeling is limited by the ability of the observer to submerge their mask underwater; streams that are exceptionally shallow (≤ 30 cm) may require an alternative method (Nickerson and Krysko 2003; See Kick Sampling). For some species, snorkeling appears to bias population estimates in favor of larger, mobile animals, and therefore small, bottom dwelling animals

may be underrepresented in such estimates (Helfman 1992; Tyler et al. 1998). For hellbenders, however, snorkeling is superior to other methods for finding the smallest individuals in a population (Nickerson and Krysko 2003; J. Humphries, pers. comm.). Finally, in cold aquatic environments, snorkeling requires the use of a wet or dry suit (Hoffman et al. 2005), which is considerably more expensive than the equipment needed to perform alternative methods (Nickerson and Kryskno 2003).

Inventory

Snorkeling as an inventory technique is probably most appropriate in conjunction with other techniques, such as basking surveys and setting hoop traps for aquatic turtles, visual encounter surveys and frog call surveys for pond amphibians, and lifting large rocks on mountain stream and river bottoms for stream amphibians. Riverine turtles are easily located by searching around the sites of basking logs or by snorkeling over beds of rooted aquatic vegetation. Pacific pond turtles (*Clemmys marmorata*), river cooters (*Pseudemys concinna*), various map turtles (*Graptemys spp.*), painted turtles (*Chyrsemys picta*), and sliders (*Trachemys spp.*) are some of the species likely to be detected. Other more secretive turtles, such as loggerhead musk turtles (*Sternotherus minor minor*) or alligator snapping turtles (*Macrochelys temminckii*) may also be found by snorkeling; these species are not likely to be seen by basking surveys.

Kurt Buhlmann

Snorkeling and canoeing for Loggerhead musk turtles (*Sternotherus minor*) in spring runs.

Tracey Tuberville

Snorkeling for alligator snapping turtles (*Macrochelys temminckii*).

Pond and lake dwelling amphibians are commonly inventoried by swimming across transects located perpendicular to the shoreline and counting the number of amphibians observed within the water column (Hoffman et al. 2005). Pond and lake dwelling amphibians may include a variety of Ambystomids, Bufonids, and Ranids. Stream dwelling amphibians in the Pacific Northwest may be found by turning over large, loosely embedded substrate, or by searching near undercut stream banks. Species in the Pacific Northwest likely to be detected by snorkeling include tailed frogs (*Ascaphus spp.*), giant salamanders (*Dicamptodon spp.*), long-toed salamanders (*Ambystoma macrodactylum*), northwestern salamander (*A. gracile*), torrent salamanders (*Rhyacotriton spp.*), and a variety of plethodontid salamanders.

Snorkeling, coupled with rock lifting, is an excellent method of inventorying hellbenders. A group of between two and six people works best for hellbender surveys, with the group moving upstream during surveys. In slower moving sections of streams, a person should very slowly lift rocks from the upstream side with a cant hook or log peavey while one or two people remain below water looking for hellbenders beneath the rock as the rock is lifted. Hellbenders usually remain relatively motionless as rocks are lifted, and lifting from the upstream side helps to keep the area beneath the rock clear from siltation. In areas with faster moving water or in silty stream bottom conditions, the person lifting rocks should lift from the downstream side while the person underwater feels for hellbenders beneath the rock as it is lifted. Other persons involved in surveys can remain downstream of the main crew, snorkeling for escaped hellbenders or turning cobble and small rocks in search of larvae and juveniles. Rock lifting surveys should be avoided during the hellbender breeding season, but snorkeling surveys not involving rock lifting can be productive during the breeding season when diurnal activity dramatically increases (Nickerson and Mays 1973; Humphries 2007).

Zach Felix

Lifting rocks and snorkeling can be an effective method for sampling hellbenders.

Monitoring

Permanently established transect lines may also be established in certain stretches of stream or river habitats, due to the relatively low habitat disturbance associated with snorkeling. Searching along established transect lines by snorkeling may provide an index to turtle and amphibian abundance, although other methods of monitoring might be preferable. Additionally, snorkeling may allow for *in situ* behavioral observation and monitoring of turtles and amphibians. Estimates of river cooter and Florida red-bellied turtle (*Pseudemys nelsoni*) abundance have been estimated in certain Florida karst springs by time- and distance-constrained snorkeling surveys (Meylan et al. 1992).

Kurt Buhlmann

A Florida red-bellied cooter (*Pseudemys nelsoni*) captured by snorkeling in a clear, karst spring.

For monitoring pond and lake dwelling amphibians, transects should be located along the perimeter of a site and perpendicular to the shoreline (Hoffman et al. 2005). The snorkeler swims along the transect, counting the number of amphibians observed within the water column. Area constrained searches are recommended, since the time required to conduct a survey is dependent upon the habitat complexity. Estimates of tailed frog larvae densities may be monitored using the same surveyed areas during subsequent sampling dates.

Hellbenders may also be monitored using transects provided that the same areas are surveyed at the same season and time in subsequent years. Usually, transects of several hundred meters are needed to obtain enough meaningful capture data. The time actually spent searching for hellbenders, as well as the number of people searching, should be recorded in order to develop an index of abundance during each survey effort. In addition to relative abundance estimates, hellbenders can easily be marked using PIT (Passive Integrated Transponder) tags to obtain population and density estimates for stream sections. Streams should probably not be surveyed more than every other year, especially if rock lifting is a major component of monitoring efforts. Lifting rocks can damage habitat, dislodge nests and larvae, and may make rocks more vulnerable to becoming dislodged during flooding events.

Conclusions and recommendations

Snorkeling is an effective technique for sampling and monitoring some turtle and amphibian species. For some species (e.g., hellbenders), snorkeling is the most effective method for locating small size classes often overlooked using other sampling techniques. Snorkeling requires relatively expensive equipment (e.g., wetsuits) and can be dangerous in some situations. In addition, numerous variables including water clarity, time of year, substrate composition, water temperature, water depth, and velocity can affect sampling effectiveness, and these variables should be considered when comparing data among surveys or locations.

Equipment Checklist

Installation:

- A Partner - NEVER ATTEMPT THIS METHOD ALONE
- Understanding of river of lake depths, currents
- Transect end markers

Sampling:

- Mask
- Snorkel
- Flippers (for use in lakes)
- Tennis shoes or Felt wading boots for streams
- Knee pads for small streams
- A small aquarium net and plastic container
- Log peavey or cant hook (for lifting rocks)
- Canoe or Boat (remote areas: inflatable kayak)
- Water-proof Watch
- Digital or disposable underwater camera
- Properly fitting Wet or Dry Suit
- Snorkeling hood
- Neoprene gloves
- Nylon mesh bag
- Underwater flash light
- Secci Discs
- Underwater writing board and pencil (or waterproof paper)
- Water flow measuring equipment
- Marking equipment (PIT tags)
- Measuring equipment (tape, scales, thermometer, etc.)

* See Appendix IX for basic equipment lists

ELECTROSHOCKING

Lawrence A. Wilson

Electroshocking is the technique of using electricity to capture aquatic animals, primarily fish (Murphy and Willis 1996; Cowx and Lamarque 1990), but also aquatic amphibians (Meffe and Shelton 1987; Williams et al. 1981), a few selected reptiles (L. Wilson, unpublished), and macroinvertebrates (Taylor et al. 2001). The development of electrofishing as a fisheries sampling technique began after World War II. During the 1960s-1990s it was observed by many fishery biologists that electroshocking was equally effective at collecting many species of aquatic salamanders. Since that time, numerous studies have utilized the technique of electroshocking for the capture and sampling of salamanders.

Steve Fraley

Biologists electroshocking a small stream.

Electroshocking is a wildlife sampling technique which uses high currents of electricity to temporarily stun fish or other animals. Sighting and capture are much easier than with more traditional techniques such as dip-netting. Shockers can be powered by either batteries or a gas powered electric generator. The current can be AC current, DC current, or a DC pulsed current. The use of DC pulse has proved to have a less lethal effect on amphibians and also is the most effective current for drawing hiding animals out from burrows or under rocks. A current produced by electrodes separated by a potential of 50-990 volts can be used, but generally the voltage is in the 500-700 volt range, depending upon water conductivity. Many environmental variables can affect the efficiency of using electroshocking. The most important parameter is water conductivity but other factors include water temperature, water transparency, and dissolved oxygen. Biological variables, such as innate differences in species' reaction to electroshocking, morphology, physiology, behavior, and specimen activity and size, also affect the efficiency of successful surveys.

Utility

Electroshocking has several advantages over conventional hand-collecting, dip-netting, and seining methods. Electroshocking is best used to survey the presence of animals rather than a day by day monitoring of populations. The technique is useful in "extracting" animals which are hiding under bottom structure or burrowed into bottom substrate. Several studies have used electroshocking to successfully find salamanders (e.g., sirens) during diurnal periods of inactivity (Dundee and Rossman 1989) and in situations where other methods have failed to reveal any animals (Rossman 1960; Bennett and Taylor 1968).

This survey method is effective at sampling a variety of age classes so that adults are not the only age class collected. Electroshocking surveys can be conducted much more quickly and with less manual effort and less disturbance to the habitat than netting or methods that involve dredges, Peavy hooks, and crowbars (Nickerson and Krysko 2003). Shocking is most effective in shallow clear waters with depths not exceeding one meter. This technique has been successfully used to survey hellbenders (*Cryptobranchus alleganienesis*), sirens (*Siren intermedia* and *S. lacertina*, and *Pseudobranchus species*), mudpuppies (*Necturus sp.*), and *Leurognathus* (*Desmognathus*). Other species can be shocked, including *Gyrinophilus*, *Eurycea*, *Desmognathus*, *Apalone*, and *Nerodia,* but these have been incidental captures while conducting other surveys.

Limitations

It should be strongly noted that electroshocking has some real limitations as a technique. First and foremost, it is a potentially dangerous activity using electrical currents that can be lethal to both researcher and specimens. Most states have restrictions on the use of electroshocking and almost all require a scientific collecting permit specifying the use of electroshocking as a method of capture. Both a federal and state permit are required if surveys are conducted on federal lands (e.g., National Forests). Secondly, when conducting

surveys using this method, at least one member of the survey team should be experienced and certified to use shocking equipment. Proper safety equipment (insulated chest waders, insulated gloves if operating the shocker) should be utilized by all members of the survey team.

The equipment required for this technique is costly and there are only a few companies that commercially manufacture electroshockers. The traditional favorite has been manufactured by Smith-Root, Inc, a company located in Washington State. Their electroshockers range in price from low end shockers of $4400 to the deluxe models of approximately $7220 (as of 2007). The other manufacturer of shockers, which has been cited in many papers (Coffelt Manufacturing Inc.), has gone out of business.

Another major limitation of this technique is the actual physiological impact that electroshocking has upon the animals. The effect on the animal is dependent on the voltage received and the duration of the shock received (Dwyer and White 1995). Environmental parameters of the water (conductivity, temperature, bottom structure) and biology of the animal (age, health, and activity) greatly influence the effect that shocking has on specimens. The animal's survivability also depends on how the animal is handled, housed, and returned to the water. Many studies have shown that larval fish and eggs suffer ill effects much greater than do adults, which probably translates for amphibians as well. Therefore, this technique should not be used during egg deposition and nesting seasons (September through November), especially when endangered species such as the hellbender (*Cryptobranchus alleganiensis*) are present.

Inventory

Electroshocking is a technique best used to survey streams and lakes for the presence of amphibian species. It can be a very effective method compared to netting or turning rocks and logs by hand. In order to standardize collections care should be taken to adjust for nocturnal versus diurnal sampling and seasonal effects as the technique is less effective during periods of the year when animals are inactive. Another factor that should be considered is returning animals as close as possible to their capture site. Salamanders, such as hellbenders are very territorial and dislodging them from prime territories might stress them, especially during the courting and nesting season.

Monitoring

Electroshocking is a method that is not recommended to be part of the monitoring protocol for populations. Populations should not be subjected to a repeated regimen of surveys. Once a population is found to exist in a section of stream or body of water, other methodologies need to be employed to assess population structure and long-term health of the population.

Conclusions and Recommendations

Electroshocking is a useful sampling technique with very limited application. It is especially useful for conducting inventories in bodies of water. It is best to coordinate with skilled fishery biologists in order to dovetail surveys, but equally important to utilize their equipment and expertise in using electroshocking equipment. It should be noted that electroshocking is a potentially dangerous and expensive technique

Sampling Protocol and Operating Guidelines

1. Electroshocking is a hazardous activity in which **safety is the primary concern**. The electrical energy used is sufficient to cause death by electrocution.
2. **Never electroshock alone!**
3. Teams should be comprised of **2-3 people** to avoid communication problems. Each team should have at least one experienced biologist/technician.
4. During operation it is critical to **avoid contact with the electrodes** and water.
5. There should be a **safety switch** to cut off the power source. It should remain off until the signal is given to the entire crew that electroshocking is to begin.
6. If any **emergency occurs immediately turn off the main power** supply.
7. **Rest often to avoid fatigue**. Most accidents occur when operators are tired.
8. **Dry skin and clothing** are good protection against electroshock. Avoid operating the electroshockers during rain or if necessary wear protective raingear.
9. At least two members of the team must have **knowledge of CPR and first aid**.
10. **Communication is important** and a hand signal system should be reviewed before shocking begins.
11. **Stunned fish or amphibians should be removed from the electric field as soon as possible** and not subjected to continuous elecroshock by being held in the water in a dip net. Using the anode as a dip net is unhealthy and dangerous to both operator and specimens.
12. When electroshocking, **conduct the survey slowly and carefully**. Many sites are hazardous with deep holes and slippery rock surfaces. Also, slow movement avoids scaring animals out of the electrical field.
13. **Do not operate the electroshocker too close to bystanders** such as fisherman, pets, or livestock.

and should be undertaken by only trained personnel. Before conducting an electroshocking survey, proper permits should be obtained and local wildlife authorities should be notified as to where and when surveys will be conducted. Although electroshocking is a useful technique for aquatic amphibians, its utility for any other group of herptiles is limited at best. The technique has many drawbacks, so in most cases other methods should be employed whenever possible.

Equipment Checklist

Installation:

- Map of the area and design plan
- Flagging, tape measure for delineating stream length to be sampled
- Proper permits

Sampling:

- Electroshocker backpack with battery or gas generator
- Hip waders (check to make sure there are no leaks)
- Rubber gloves
- Wooden or fiberglass handled dip nets
- Collection containers for specimens
- Fuel or extra batteries for shocker unit
- Ear protection if operating gas generator shocker
- Buckets and coolers for collected specimens
- Data sheets, data logger
- Seines to catch drifting specimens and delineate the collection area (Optional)
- Salt block (if needed in low conductivity waters)
- Yellow Springs or similar meter to measure dissolved oxygen, conductivity, and water temperature.

* See Appendix IX for basic equipment lists

AUDITORY SURVEYS

Thomas M. Luhring

Monitoring adult amphibians at breeding sites is an effective way of gathering data to estimate species' richness and abundance (Scott and Woodward 1994). Male anurans in particular are fairly conspicuous during the breeding season as they call or vocalize to attract mates and warn other nearby males (Mitchell 2000). The frequency or intensity of these calls can be used to successfully estimate population sizes (Nelson and Graves 2004, but see limitations section below).

The North American Amphibian Monitoring Program (NAAMP) has established a unified protocol for conducting auditory surveys throughout North America (Weir and Mossman 2005; http://www.pwrc.usgs.gov/naamp/; see sample NAAMP datasheet in Appendix VIII). Basically, ten randomly selected (i.e., computer-generated) listening stations are placed near potential breeding habitat or equidistantly (0.5 miles apart) along secondary or smaller roads. A volunteer observer then visits all ten sites in one night and listens for anuran calls for five minutes at each station. The observer records time, wind and sky conditions, species' presence and relative abundance, and any outside noise (e.g., traffic, trains) that could affect the results. Observers return to their sites repeatedly during three seasonal sampling periods.

Tom Luhring

Utility

Auditory surveys have been successfully used in a number of states and provinces in North America (Scott and Woodward 1994). Notable programs include initiatives in Wisconsin and Illinois that were started in 1984 (McDiarmid and Donnelly 1994). Auditory surveys from roadsides work best in areas of North America where the anuran species are fairly predictable in their vocalizing behavior (Weir and Mossman 2005). Call surveys are advantageous because they cover a large area, are fairly non-invasive, and can be done effectively by a group largely composed of volunteers.

Limitations

One limitation of this sampling technique is that not all anuran species are equally detectible (Scott and Woodward 1994). For example, Bridges and Dorcas (2000) found that *Rana sphenocephala* (unlike many species) call after midnight in summer months, with peak activity between 0100 and 0530 hours. In addition to temporal differences in calling activity, they also found that not all anuran species in a breeding area are aurally detectable each night.

While the calling index feature of the NAAMP protocol is useful in determining relative abundance of a species, it reaches a threshold when the chorus reaches a level 3 rating (Weir and Mossman 2005). Level 3 occurs when calls in the chorus are constant, continuous, and overlapping. This results in an inability to detect fluctuations in population sizes above the minimum number of adults that it takes to reach a level 3 chorus. Species-specific relationships between chorus intensity and population levels are still needed because qualities such as call length could skew the number of vocalizing males required to reach each level of abundance.

Species detection ability does not appear to vary much between experienced and novice observers, however, calling index values can have much more variation (Shirose et al. 1997). Weir et al. (2005) suggested recording observer experience in addition to other data presently taken. Call surveys are also limited by the amount of time that technicians or volunteers have for monitoring sites and an inability to monitor multiple areas at the same time without observer bias. Likewise, the effects of observer bias should be taken into account (Lotz and Allen 2007). In some cases, automated recording devices may be more appropriate (see section on automated recording devices).

Inventory

Regular anuran call surveys are helpful in determining species composition for inventory purposes (Mitchell 2000; Scott and Woodward 1994). Inventories of anurans via call counts are fairly easy and can result in the detection of species that are otherwise hard to detect away from their breeding area (Dodd 2003). This is an especially applicable technique if the goal of the inventory is to focus more on presence versus absence of a species rather than population estimates of anurans (see limitations section above).

Monitoring

When used over successive years, call count surveys can be used to monitor changes in population levels of a species as well as changes in species assemblages (Scott and Woodward 1994). However, using call count indices as sole indicators of population levels over time can result in a bias toward observing declines (see Weir and Mossman 2005). Additionally, populations of amphibians fluctuate naturally and care should be taken to discern natural population fluxes from those caused by anthropogenic agents (Pechmann et al. 1991). While call count surveys can provide basic information about presence or absence, population estimates used for monitoring purposes may be more accurate if coupled with other various sampling techniques presented in this book.

Conclusions and recommendations

This technique may be most useful for inventorying purposes and detecting presence versus absence, and care should be taken when using call surveys for population monitoring. When used for monitoring programs, auditory surveys may be most useful if supplemented with other sampling techniques. Relative to the use of auditory recording devices (i.e., frogloggers; see section on automated recording devices), auditory surveys may be most applicable under situations where capable and committed technicians and/or volunteers are plentiful and sufficient funding for recording devices is not.

Equipment Checklist

Installation:

Map of area and design plan
Flagging/stake flags for marking listening stations

Sampling:

Data recording materials (e.g., datasheet, pencil)
Thermometer
Vehicle (if need to drive between listening locations)

* See Appendix IX for basic equipment lists

BASKING SURVEYS AND BASKING TRAPS

Kurt A. Buhlmann

Aquatic turtles that frequently bask on rocks and logs, as well as certain watersnakes, may be inventoried and even monitored through the use of basking surveys. Basking surveys involve scanning with binoculars for basking individuals of the target species while floating in a boat (if on a still body of water) or moving parallel to the river bank at an appropriate rate of speed. A basic inventory can be accomplished for most basking species and in some cases, information on sex ratios and juvenile recruitment may be obtained (Buhlmann and Vaughan 1991; Lindeman 1998). Brown watersnakes (*Nerodia taxispilota*) have been surveyed as they frequently bask on overhanging branches along southeastern rivers (Mills et al. 1995).

When further information is needed, basking traps can be used. These traps are particularly useful when the water depth and velocity of large rivers prevents trapping turtles with aquatic traps (see aquatic and terrestrial funnel trapping section). Wire traps created from chicken wire may be attached to the undersides of logs used by basking turtles. Turtles drop from the logs into the traps, where their first instinct is to dive to the bottom of the trap, giving nearby researchers an opportunity to get to the trap and extract the turtles. Basking traps should be designed to allow captured turtles to simply climb out of them by themselves if a researcher is not present to collect them (Sexton 1959; Ream and Ream 1966; MacCulloch and Gordon 1978; Buhlmann and Vaughan 1991).

Kurt Buhlmann

Basking surveys can be supplemented by setting traps along basking sites.

Utility

Rivers are particularly well-suited for basking surveys because of limited access. The effectiveness of basking surveys is often determined by 1) the amount of substrates (i.e., logs, rocks) available for basking, 2) the time of day and year during which they are conducted, 3) the species' proclivity for basking, and environmental conditions during the survey, such air temperature, water temperature, and cloud cover. Some turtle species, such as river cooters, will bask on cool days if air temperature exceeds that of the water (usually in spring or fall), but may bask cryptically in beds of aquatic vegetation that become quite warm during the summer. In some species males seem to bask regularly throughout the active season, while females commonly bask only during the spring (Buhlmann and Vaughan, 1991), perhaps to assist egg development. It is likely that aquatic snakes behave in much the same way as the turtles. In many large rivers, water depth and velocity prevent trapping of turtles with aquatic traps. Therefore, wire traps may be attached to the undersides of logs used by basking turtles.

Limitations

A lack of suitable, or at least abundant basking sites, does not necessarily mean a species is absent if it is not observed by a basking survey. Therefore, the availability and abundance of suitable basking structure will determine if a basking survey is a suitable survey technique. Calculating the amount of basking structure available relative to the amount occupied by turtles or watersnakes is necessary to make relevant inference about abundance and to compare among different sites (Mills et al. 1995; Lindeman 1996; Lindeman 1997). Some turtle species, notably cooters and map turtles, are wary when basking, and often drop from basking sites before positive identifications can be made via binoculars. Although cooters and map turtles are usually distinguishable by their uniquely different shell silhouettes, in some rivers more than one map turtle species may occur, adding to the difficulty in making positive identifications. Some species will allow observers close enough to confirm identification with the naked eye; others will require binoculars, and still others spotting scopes. Note that spotting scopes are actually difficult to use from moving boats and are better suited for identifying turtles on opposing river banks from land. Turtles that do not frequently bask should be inventoried using other methods (see Table 5.1).

Inventory

Long stretches of river habitat may be quickly and easily assessed for presence of select turtles by basking surveys. Certain watersnakes, notably brown watersnakes (*Nerodia taxispilota*), diamondback watersnakes (*Nerodia rhombifer*), and others may be inventoried by quietly drifting downstream in a canoe and observing carefully for snakes basking on overhanging branches (Mills et al. 1995). Basking traps, such as those made from wire and attached to basking logs, can be used to capture turtles in large, fast-flowing, deep rivers where the use of hoop traps or fyke nets is not possible. Note: hoop traps set in flowing water will collect floating debris and will eventually collapse and be submerged, thus drowning any captured turtles. Basking traps are subject to fluctuating water levels, and some turtles will stop using a favorite basking log once a trap is attached to it (K. Buhlmann, pers. comm.). Basking traps need to be

attached to favorite basking sites, meaning that these sites must be identified through previous confirmation (i.e., a basking survey) that turtles are using them. Given the wariness of certain species, hand-captures are often unsuccessful, and basking traps are a preferred alternative.

Monitoring

The numbers of basking turtles of target species observed per defined distance, such as river mile, may be used to develop a relative index of abundance. Noting general size categories of turtles may also help identify recruitment in the population. Using basking surveys to monitor populations also requires the same periodic assessment of basking site availability. Conversely, the local abundance of a map turtle (*Graptemys* sp.) was related to basking site availability (Lindeman 1998).

Use of basking traps may help to observe the turtles first hand and establish a relationship between age class and size categories. Basking traps are useful to monitoring programs when used in conjunction with mark-recapture studies and when the goals include monitoring growth of individuals, changes in sex ratios, conducting health assessments, or any other data collection that requires that the turtle or watersnake be captured and examined. It is also important to understand differences in basking frequencies between males and females, especially with regard to time of year. In general, a greater proportion of female turtles may bask during the spring and early summer months when a raised body temperature may be necessary to produce a clutch of eggs. The proportion of basking female turtles observed, relative to males, declined in a West Virginia population of river cooters between April and August (Buhlmann and Vaughan 1991).

TABLE 3. Mean basking frequency (see text for explanation of basking frequency calculations) of marked basking river cooters (*Pseudemys concinna*) observed monthly in the New River, West Virginia, 1985.

Month	Surveys	Males	Females	*P*
April	16	17.2%	24.6%	ns
May	15	15.5%	27.1%	ns
June	24	20.1%	21.8%	ns
July	4	16.0%	6.3%	ns
August	3	21.0%	0.0%	<0.01
September	10	15.6%	1.3%	<0.01

Kurt Buhlmann

Example of type of frequency data that can be obtained from basking surveys

Conclusions and recommendations

Basking surveys conducted using binoculars may provide a quick and efficient method to determine the presence of certain turtle species and some watersnakes that are frequent baskers. For some turtles, limited data about sex ratios and juvenile recruitment may be obtained. Any further assessments of population size, health assessments of animals, and reproductive condition require handling of the turtles. Basking traps are a useful technique to capture individuals of frequently basking species, especially in situations where water depth, velocity, or other factors prevent use of baited hoop traps.

Environmental conditions affect the frequency of basking by turtles. Factors to consider when planning a basking survey include: time of year, time of day, cloud cover, wind speed, water temperature, and air temperature. Surveys should be conducted during late spring and early summer when turtles are active and females are basking, but before high summer air temperatures and warm summer water temperatures reduce basking behavior.

Equipment Checklist

Installation:

- Map of river area
- Canoe or boat
- 4ft tall x 50ft roll of chicken wire
- Rope
- Tin snips
- Work gloves

Sampling:

- Data recording materials (e.g., field notebook)
- Binoculars
- Marking and marking equipment

* See Appendix IX for basic equipment lists

SIGN AND TRACKING

Ria N. Tsaliagos

Various signs created by herpetofauna can lend helpful evidence to an individual's whereabouts, activities, and habits. The main use of sign and tracking for locating or studying an amphibian or reptile is as a supplement to other types of survey techniques or field methods. Much of the literature regarding comprehensive studies that use sign and tracking pertains to mammals (Stokes and Stokes 1986; Stall 1989), however, amphibians and reptiles also leave many

different types of signs. Herpetofauna may be located by following distinctive footprints, by locating recently used burrows, hibernacula, or nests, and by identification of scat (Murie 1954; Stall 1989). Some specific techniques that herpetologists use for locating signs are road cruising (see road cruising section), burrow and nest surveys (see egg mass and nest counts section), and surveys for turtle shells and shed snake skins. Sign and tracking can be used as part of a visual encounter survey (see visual encounter survey section), as in the case of road cruising, or simply as a method for collecting more information on an individual, species, or population. To the best of our knowledge, with the exception of locating eggs, no sign or tracking techniques have yet been developed and firmly established for amphibians.

The tracking technique is useful in any habitat in which taxon-specific signs can be located. Some species may leave easily spotted evidence of their presence wherever they are (e.g., scat is commonly used for a variety of mammal species), whereas others may only leave obvious signs in certain microhabitat (e.g., tracks from a snake in sand or loose soil; alligator scat in clear waters). However, tracking reptile signs is most applicable in deserts, sandhills, roads, and semi-aquatic areas. Amphibians may be most easily located near their breeding sites, as higher density may result in a greater abundance of signs in the immediate area; however, thus far, the tracking technique has not been used effectively for amphibians.

Gabrielle Graeter

Fragments of a box turtle shell.

Jeff Beane

A hatchling Southern Hognose snake (*Heterodon simus*) and it's track in the sand.

Jeff Hall

Shed skin of an eastern indigo snake at the entrance to a gopher tortoise burrow.

Jeff Hall

Tracks left in the mud by an eastern mud turtle (*Kinosternon subrubrum*).

Utility

Tracking has been used for many different purposes in the study of reptiles. For example, to gather data on sidewinder populations, Lillywhite (1982) followed their tracks over desert sands for more than 1.2 km. Locating gopher tortoise burrows or nesting sites for aquatic turtles is one way of examining the movement and activity of an individual or population (see egg mass and nest counts section).

Limitations

A major limitation to this technique is that it is not a stand-alone method. It should only be used to supplement other methods, such as a visual encounter survey, or to gain additional information about a study population. A potential bias is that some individuals or species may be more easily located or detected than others, a limitation that other visual encounter techniques share. It may be impractical and an inefficient

use of time to attempt to locate or study some species this way; they may be much more easily located using other methods.

Inventory

The use of signs for locating or tracking herpetofauna is a very useful technique for a basic or comprehensive inventory when used in conjunction with other survey methods. For example, even if a particular lizard or snake species is not spotted, a recent sign of their presence, such as scat, indicates that individuals are indeed inhabiting or using the area. Similarly, if a nest known to be specific to a particular turtle species is found, this is definite evidence of their presence. Thus, this type of information can be helpful when one is simply determining presence or absence of a species. However, this technique is limited if not used along with other techniques.

Monitoring

Using signs to locate an individual is not as useful as a monitoring technique because it is difficult to standardize this method. However, it could be used for monitoring if employed along with other techniques and if effort is standardized. It may be most appropriate when a monitoring program is aimed at a particular species or set of species that are easily and reliably located with the use of signs. For example, this method could be used for monitoring gopher tortoises because of the relative ease of telling whether a burrow is in use during their active season. It could also be used for monitoring reproductive activity in reptile species whose nests are easily located, such as crocodilians.

Conclusions and recommendations

Use of signs should be used only as a supplementary technique to other methods, such as visual encounter surveys. It can be useful for obtaining additional information about some species, but is limited in that some species may not leave easily discernable or detectable signs. This reemphasizes the importance of consulting guidebooks and experts about the natural history and background on each species of interest before adopting a specific sampling plan.

Equipment Checklist

Sampling:

Data recording materials (e.g., field notebook)
Camera

* See Appendix IX for basic equipment lists

ROAD CRUISING

Kimberly M. Andrews

Road cruising is a technique commonly used by herpetologists for surveying and collecting local herpetofauna. The premise of the technique is that the road serves as a survey transect that is methodically driven. This technique has been used to document factors such as species diversity and richness, in addition to demographical and behavioral attributes of the herpetofaunal community (e.g., amphibians, Shaffer and Juterbock 1994; reptiles, Sullivan in press). Road use by herpetofauna has been documented since the beginning of the 20th century (e.g., Stoner 1925) which led to the development of road cruising to collect data on local species compositions in the deserts in the western United States (Klauber 1931, 1939).

Although some anuran and salamander species can be found on roads at night in punctuated densities during or following rain, road surveys are not as consistently effective as with applications for reptiles. However, there are exceptions to this. Road cruising can be most useful with amphibians in areas where the road immediately bisects or is peripheral to an inhabited body of water for documenting highly vagile species during breeding or foraging movements (e.g., Shaffer and Juterbock 1994). Additionally, US Geological Survey's (USGS) North American Amphibian Monitoring Program (NAAMP) uses road survey routes in a stratified random block design for volunteers to conduct seasonal sampling of frog calls (NAAMP, www.pwrc.usgs.gov/naamp, 27 March 2006).

Michael Marchand

Spotted salamander (*Amybstoma maculatum*) crossing a paved road.

The cryptic and secretive nature of reptiles, particularly snakes, makes designing road surveys for reptiles challenging and the surveys themselves incredibly time-consuming due to difficulties in detecting some species. Crocodilians occasionally cross roads (e.g., American Alligator, *Alligator mississippiensis*, Smith and Dodd 2003; American Crocodile, *Crocodylus acutus*, Gaby 1987), although these events are usually on roads bisecting wetlands and other aquatic habitats immediately encroaching the road edge. Snakes and turtles can be observed crossing roads in their peak activity periods, such as species-specific nesting or breeding pulses. When certain habitats are bisected by roads, snakes and turtles can be found both alive-on-roads (AOR) and dead-on-roads (DOR) on almost a daily basis (e.g., Smith and Dodd 2003; Aresco 2005a; Andrews and Gibbons 2006). Obviously, marine turtles cannot be adequately sampled using road surveys although sometimes they are captured on roads bordering beaches due to an attraction to lights for both hatchlings and nesting females (e.g., McFarlane 1963).

Lizard encounters on roads in the United States are not necessarily rare, but in most regions are not frequent enough for the road cruising method to be a productive sampling technique. This trend could be a result of rapid deterioration of small-bodied carcasses, a reduced level of mortality due to an ability to move quickly across the road, or a lower encounter rate with roads as many species have smaller home ranges and high site fidelity (Andrews et al. 2007). Road cruising for lizards is likely most productive in the western regions of the United States (e.g., Kline and Swann 1998) where you are more likely to encounter this taxon on the road due to both a higher diversity of native species and presumably a lower rate of road avoidance due to roads being a more continuous environment (i.e., open) with their resident habitats. Road cruising for lizards in Florida could also be productive; due to the increasing number of invasive species, some of which are large in body size, there could be a greater chance to observe lizards on roads.

Utility

First and foremost, it should be established that road cruising should never be used as the sole technique to achieve a complete census of local populations and their ecological dynamics. Data are inherently confounded due to the many biological biases and inconsistencies associated with the technique (e.g., Case 1978) and the multitude of impacts on the surrounding landscape from the road itself (e.g., Forman et al. 2003). As roads affect local populations, on-road data cannot be considered statistically independent or representative of the dynamics that would occur in the absence of roads. For research objectives that do not involve measuring an effect that is directly related to the sampling technique itself, the selected sampling technique ideally should not directly influence the organisms of study; this mantra is simply not achievable with the road cruising technique.

Kurt Buhlmann

Timber rattlesnakes can be censused by road cruising.

Most legitimized sampling techniques can lend scientifically useful data but only when shortcomings are recognized and respected. The following assumptions stated by Shaffer and Juterbock (1994) must be met when performing road surveys: 1) roads are not a barrier to movement for target organisms; 2) individuals are not deterred or attracted to roads; 3) characteristics of the road do not influence richness or abundance in the immediate vicinity of the road; and 4) individuals are only sampled once per day. Due to emerging data on the breadth of impacts from roads (e.g., Andrews et al. 2006, Andrews et al. 2007), these assumptions are becoming more challenging to meet. Although road cruising data should be handled delicately, this technique has proven to be incredibly useful for either a simple sampling objective or when directly measuring road impacts.

Considerations and limitations

Road cruising is useful as a quick and easy way to explore a new area for areas of prime habitat for particular species. However, the use of the technique for scientific data collection is most successful when targeting specific areas, species, and times, rather than when conducted haphazardly or randomly. Organismal responses to biotic and abiotic conditions vary within and among species and therefore, the productivity of road cruising is dependent on these conditions of influence. The biases of the road cruising technique are commonly represented in the literature (see references in Andrews et al. 2006) and are more effectively mitigated for than corrected (e.g., Case 1978).

Characteristics of roads – Many factors of roads can be influential and therefore should be considered when designing road surveys. First, road density can be a substantial factor in the ability to detect animals on roads. Generally, areas of lower road density can yield a higher frequency in observations as populations often suffer local declines and fragmentation with increased road density (e.g., amphibians, Vos and Chardon 1998; reptiles, Rudolph et al. 1999). The age of the road is also a factor to consider. The composition of on-road observations can shift across time with correspondent shifts in the surrounding wildlife community and habitat (e.g., Mendelson and Jennings 1992). Shifts could also be attributed to behavioral or genetic shifts in the local population by which individuals avoided roads through behavioral modification or evolutionary pressure (Seigel and Pilgrim 2002). Also, as roads can instigate population reductions over time (e.g., Boarman and Sazaki 1996), surveying newer roads can be more independent of the impacts that roads themselves have on the surrounding landscape. In terms of road substrate, asphalt roads are easier for readily spotting animals (e.g., Bugbee 1945). Dirt roads will permit more individuals to cross successfully because they are usually present in habitats that have experienced minimal disturbance and conversion and therefore assume minimal wildlife impacts. The size (width and presence of a median) can also influence the level of road impacts. For instance, surveying two-lane roads with moderate traffic densities is more worthwhile than surveying interstate segments. Initially, roads with higher traffic densities can result in more frequent DOR individuals, but over time highly traveled roads can become less productive routes as mortality impacts of local herpetofauna take their toll on local abundances (e.g., amphibians, Fahrig et al. 1995; reptiles, Klauber 1939). Lastly, speed limits will influence mortality rates (e.g., Case 1978; Cristoffer 1991) and presumably levels of behavioral avoidance (e.g., Klauber 1931). These factors must be applied with caution as a certain road characteristic that increases the chance of observing an individual of one species may not apply to all species (Mazerolle 2004).

Surrounding landscape – The habitat bisected by the road must also be considered. For instance, amphibians and some aquatic reptile species are generally only found in large numbers on roads immediately bisecting wetland habitat (Bernardino and Dalrymple 1992; Smith and Dodd 2003). As many herpetofaunal species are amphibious, using both aquatic and terrestrial habitats, landscape-level configuration of habitats in the area surrounding the road also becomes a critical determinant in the composition and abundances of animals using the road. Therefore, many animals can be observed on roads if they cross a migratory route between two critical habitats or locations. This is particularly true with organisms reliant on metapopulation dynamics, such as seen with amphibians (Marsh and Trenham 2001). However, roads can reduce the biological connectivity of a landscape (e.g., Andrews 1990), and result in reduced species richness (e.g., amphibians, Vos and Stumpel 1996; reptiles, Kjoss and Litvaitis 2001). These types of impacts must be considered in road surveys as they affect presence and abundance parameters.

Kurt Buhlmann

Copperhead found dead on a road (DOR) next to a recent clearcut.

Abiotic conditions – Climatic conditions should also be considered when determining the potential productivity of a road cruise. Abiotic conditions can instigate or inhibit movement for different species on natural substrate and unnatural substrates, such as roads. Heavy precipitation and warm weather may foster the movements of amphibians (e.g., Duellman 1954) and other aquatic herpetofauna (e.g., Hellman and Telford 1956) while deterring that of terrestrial species (e.g., Klauber 1931). Cloudy, overcast days will frequently prove less fruitful than a sunny day for many diurnal reptiles. Collecting environmental data is recommended as it may prove helpful in understanding road cruising data.

Detectability – The size and patterning of animals can affect how readily detectable an animal is on the road, a factor that is further complicated with dead specimens that are nearly obliterated. Slender, small snakes or darkly patterned species are more difficult to detect (Mendelson and Jennings 1992; Sullivan in review). Enge and Wood (2002) found that pedestrian road surveys, in which surveys are conducted on foot, yield the maximum number of individuals and species. Observer bias, or the researcher's ability to detect snakes on the road, will naturally influence survey results (Case 1978) and will vary with the experience level of different researchers.

Activity patterns and ecological strategies – Targeting the daily and seasonal activity patterns of local species is one of the most important steps in designing road surveys. If one is new to the area or animals, previously collected data can be found in some of the

most remote areas in the United States by contacting the closest museum, university, biologist, or state herpetologist. Selecting appropriate times of day or night to perform the surveys will determine which species could be observed. Extreme summer temperatures can suppress the activities of many amphibian and reptile species, but there are times that a species is active but will not use roads. For example, a summer afternoon in the southeastern United States yields road temperatures of 52°C (130°F), which most diurnal herpetofauna species will avoid with the exception of fast-moving species, such as Black Racers (*Coluber constrictor*) or Eastern Coachwhips (*Masticophis flagellum*; K. M. A., personal observation).

Gabrielle Graeter

Black rat snake (*Elaphe obsoleta*) crossing a paved road

The ecological requirements of the animals relative to the timing of the road survey will influence the outcome of a road cruising survey. For instance, canebrake rattlesnakes (*Crotalus horridus*) have large home range requirements and can be found more frequently than snakes which remain in more centralized locations (Andrews and Gibbons 2005), such as ringneck snakes (*Diadophis punctatus*), because wider ranging species are more likely to encounter roads (e.g., amphibians, Carr and Fahrig 2001; reptiles, Bonnet et al. 1999). Additionally, generalist species are more likely to encounter and cross roads as their habitat requirements tend to not be as restricted as specialists (Case 1978; Shaffer and Juterbock 1994).

Most amphibians and reptile species exhibit intraspecific variation in ecological requirements and strategies between sexes (e.g., salamanders, Johnson 2003; lizards, Koenig et al. 2001; snakes, Reinert and Zappalorti 1988; turtles, Morreale et al. 1984). Therefore, sex ratios calculated from on-road observations are likely to be skewed in a manner that is not representative of surrounding populations. Additionally, these data are not independent from the road as road mortality of one sex can lead to a population biased towards the opposite sex (e.g., amphibians, Fukumoto and Herrero 1998; reptiles, Steen et al. 2006). For example, female turtles often experience differential mortality on roads because many species use road shoulders for nesting habitat (Wood and Herlands 1997; Aresco 2005b). Conversely, a higher proportion of male lizards (e.g., Sherbrooke 2002) and snakes die on roads due to wide-ranging mate-locating behaviors (e.g., Sealy 2002; Andrews and Gibbons 2006). Biases also exist due to movement patterns that differ intraspecifically between juveniles and adults. Therefore, road cruising data alone will not lend an accurate picture regarding the composition of life stages within an area. Further, there are seasonal peaks in road crossings so time of year will influence the interspecific and intraspecific composition of species observed. For instance, adult male gopher snakes (*Pituophis catenifer*) were more susceptible to crossing roads in the spring whereas dispersing subadults comprised the majority of roadkill in the fall (Jochimsen 2006).

Behavioral variation – Road crossing time also influences the likelihood of observing particular species and therefore the survey results. Therefore, differential crossing speeds among species creates differential levels of mortality (e.g., anurans, Hels and Buchwald 2001; snakes, Andrews and Gibbons 2005; turtles, Gibbs and Shriver 2002), resulting in differential detection probabilities. For example, turtles are frequent victims of road mortality due to a physical inability to move fast. Lizards, however, are generally active only during warm spells in which they consistently have the ability to move quickly. Snakes are presumably the herpetofaunal group exhibiting the most variation in speed, with faster snakes being less vulnerable than slower snakes (e.g., Fitch 1949). Crossing angle is another factor that can influence crossing time but has been quantified only in snakes which typically crossed perpendicularly to the road, taking the shortest route possible (Shine et al. 2004; Andrews and Gibbons 2005). Behavioral responses to passing vehicles also vary among species, but many immobilize when vehicles are approaching (amphibians, Mazerolle et al. 2005; reptiles, Andrews and Gibbons 2005). Human behaviors can also create biases as herpetofauna, particularly snakes, are often intentionally killed on roads (Langley et al. 1989).

Michael Marchand

Blanding's turtle (*Emydoidea blandingii)* crossing a paved road.

Inventory

Road cruising can be a useful technique for inventorying local populations provided that the objective of the data collection does not entail a comprehensive survey of the local wildlife community in representative proportions. Depending on time and budgetary limitations, a local inventory would include seasonal diurnal and nocturnal surveys across multiple habitat types. Additionally, existing inventory data can assist in directing focal habitats or species for longer-term monitoring initiatives. This technique should be supplemented by additional sampling techniques.

Monitoring

Monitoring local populations using road surveys can best be accomplished by collecting baseline data not only from initial road surveys, but from the literature in regards to activity patterns and specific behaviors that might expel or exaggerate road use. In the case where a community-level survey is the objective, surveys must be conducted across daily and seasonal times, across habitats, and throughout a variety of weather conditions and levels of habitat disturbance. Additionally, long-term data must be collected due to a high level of annual variation. However, there is an ultimate tradeoff between incorporating variation in data collection and being strictly consistent for purposes of statistical quantification of your results. It is imperative for data analyses that surveys are conducted across these variables in a consistent and predetermined fashion as best as possible. Additionally, effort (researcher hours and kilometers covered per individual or species) should be recorded so that diversity and abundance estimates can be calculated relative to each other and off-road data. Lastly, when the objective is to determine diversity and abundance, individuals need to be marked so that they are not counted more than once and road cruising data should be supplemented with data collected off-road in the surrounding area. These latter objectives cannot be calculated from road cruising data alone and the former parameters are not recommended.

Conclusions and recommendations

Road cruising is incredibly useful due to the time efficiency and ease of driving the vehicle while surveying in comparison to time-constrained searches or trapping. For animals that are difficult to find, such as snakes, it is undoubtedly a productive method. However, additional survey techniques (e.g., coverboards) should be implemented to detect species not using the road or not clearly detectable due to some nature of the species, particularly with long-term monitoring data. As all techniques experience sampling biases, road cruising is best offset by a technique with different biases. For instance, species that are hesitant to cross roads and open spaces are often underestimated in samples and can be supplemented by sampling techniques that provide refugia, such as coverboards. Additionally, due to the multitude of biases inherent in road cruising, data are most robust when focal species or habitats are identified as opposed to an experimental design that is spatially or temporally randomized. Lastly, identifying abiotic and biotic biases within your data will allow for the most appropriate mitigation.

As roads can be lethal environments to humans as well, it is first and foremost essential that road surveys are designed in a manner that ensures the safety of the researchers. Using low beams when surveying at night can assist in creating a reflection off smaller snakes that might be difficult to see otherwise. Secondly, collect venomous snakes only if you have the proper equipment and are professionally trained (even dead specimens!). Many species are very defensive when captured in the middle of the road and can be challenging to contain. Thirdly, if storage, processing, and preserving capacities allow, collecting dead specimens can provide for more direct documentation and allow for future research. However, some roads are not safe to pull over on or to collect carcasses off the road. The safety of the driver and any passengers must never be neglected amidst the excitement of a find. Lastly, a speed limit would ideally be maintained that maximizes the detection level specific to the researcher. However, again, in prioritizing safety, it is best to maintain local speed limits.

Equipment Checklist

Installation:

- Map of area and road cruising plan

Sampling:

- Vehicle to drive
- Data recording materials
- Measuring and marking materials (e.g., scissors, PIT tags, metric ruler)
- Snake hook and tongs
- Containers and cloth bags
- Bucket with lid for venomous snakes
- Bright flashlight for night surveys
- Closed shoes, preferably boots
- GPS unit (optional, to record location of animal)
- Equipment for recording environmental or weather data

* See Appendix IX for basic equipment lists

AERIAL SURVEYS

Xavier A. Glaudas

Estimating population size and distribution of large-bodied animal species can be more challenging than for small-bodied species. Large-bodied species generally have smaller population densities, meaning that a greater area must be surveyed to collect sufficient data. A survey technique that copes with the difficulty of covering extensive areas is the aerial survey using helicopters or small planes. Aerial surveys are specifically used for the monitoring of large reptiles.

Before conducting an aerial survey, the study area should be partitioned into units. These units can be transects, large quadrats, small quadrats, or sections, but transects are usually preferred (Caughley 1977). In the case that transects are used, rods can be attached to the wings of the plane to delineate the transect width (Mourao et al. 2000). Units to be sampled within the area surveyed are then randomly selected. The animals detected from the air during the survey are then counted.

Utility

Due to the limitations of detectability from altitude, application of this method to reptiles has been restricted to surveys of crocodilians (Magnusson et al. 1978), sea turtles (Marsh and Saalfield 1989), and gopher tortoise burrows (J. Jensen, pers. comm.), which are large enough to be detected from the air. Open habitats maximize the probability of detecting animals from the air, so densely vegetated areas may not be appropriately sampled with aerial surveys. Distribution, abundance, and aspects of the courtship behavior and thermal biology of loggerheads (*Caretta caretta*) and Kemp's Ridley sea turtle (*Lepidochelys kempii*) have been reported using aerial surveys (Coles and Musick 2000; Frick et al. 2000; Gomez de Segura et al. 2003). Gopher tortoise burrows can be censused by aerial surveys of certain landscapes, especially recently burned habitats, powerline right-of-ways, old fields, and other similarly open habitats (J. Jensen, pers. comm.).

Limitations

Even though this method is one of the most practical when sampling extensive areas, the estimate of population size is usually inaccurate. However, methods have been designed to detect, reduce, or eliminate the inaccuracy associated with aerial surveys (Caughley and Goddard 1972; Caughley 1974; Caughley et al. 1976). Many parameters have to be held constant when using transects: flight elevation, speed, and the width of the area sampled. These are some of a number of factors that can be difficult to control, but can strongly affect the detectability of individuals (Caughley 1974). Additionally, as with other visual techniques, the ability to detect animals may vary greatly among observers (Caughley et al. 1976).

Inventory

This technique is appropriate to inventory the presence of large-bodied species that are in relatively open habitats. It is also particularly useful in surveying areas that are difficult to access or even inaccessible by foot (Mourao et al. 2000).

Monitoring

This technique has been used to monitor crocodilian, sea turtle, and gopher tortoise populations. Even though estimates may be inaccurate, this method can be useful to estimate population characteristics (e.g., rate of increase) in relative numbers rather than in absolute numbers, assuming that biases are held constant (Caughley 1974). The total population size can be estimated by multiplying the mean density of animals per sample unit by the total number of units assuming that these units are of equal size. If sampling units differ in size, the density of animals per sample has to be corrected using the ratio estimate (for more information on sampling in aerial surveys see Caughley 1977).

Conclusions and recommendations

Aerial surveys are most applicable for alligators, sea turtles, and gopher tortoise burrows and have mainly been used for sea turtles on the southeastern coast. This method can be a great inventorying technique for surveying these large herpetofauna, particularly in open habitats. When used as a monitoring technique, one must consider and adjust according to potential limitations, such as that population estimates may be biased. Another critical consideration when using aerial surveys for monitoring purposes is that flight parameters must be properly standardized

Equipment Checklist

Installation:

Map of area and design plan with flight transects marked

Sampling:

Airplane or helicopter with rod to delineate transect width
Binoculars
Data recording materials

* See Appendix IX for basic equipment lists

EASY PASSIVE SAMPLING

ARTIFICIAL COVER

Tony M. Mills, Lucas R. Wilkinson, and Gabrielle J. Graeter

Turning over cover objects, such as logs, rocks, and human debris, that provide refuge for many herpetofauna, is one of the most productive ways to capture amphibians and reptiles in the field. While this method is effective, effort can be difficult to quantify, microhabitats are sometimes disturbed, and there is great potential for observer bias. Using artificial cover objects, often referred to as coverboards, makes use of this proven field technique while allowing standardization of sampling effort, maintaining natural habitats, and limiting observer bias.

Materials used to create artificial cover include solid wood boards, plywood boards, corrugated metal strips, tar paper, and horticultural plastic sheeting (Fig. 5-7). Coverboards should be placed in organized arrays, making them easier to find when covered with debris and increasing the standardization of effort among sites. Common array designs are linear transects, rectangular grids, and webs (Fig. 5-8). Choice of array design depends on the species and habitats sampled and the desired data. For example, a linear array paralleling a river or stream may be appropriate for sampling aquatic snakes, whereas a large grid may provide information about movement and home ranges for species with relatively small home ranges (e.g., small fossorial snakes and some terrestrial salamander species).

Materials for creating artificial cover:

Plywood: 1/2 inch thick or greater. 2'x4' or 4'x4' are workable sizes
Pros: 1) good insulation, 2) effective in warm to hot weather, 3) lasts 2-4 yrs
Cons: 1) expensive 2) heavy

Galvanized roofing tin: comes in various lengths, usually about 2' wide
Pros: 1) lasts 20+ yrs, 2) effective in cooler weather, 3) relatively lightweight
Cons: 1) too hot in summer, 2) may be somewhat costly

Tar paper: comes in long rolls up to 50'
Pros: 1) maybe be easily cut into section, 2) lightweight, 3) inexpensive
Cons: 1) hot in summer, 2) breaks down in water

Figure 5-7. Pros and cons of different materials used to create artificial cover.

Coverboards should be quickly lifted from one side and animals underneath immediately captured before they retreat into burrows or cracks in the soil. Captured animals should be handled and processed according to the recommendations in Appendices I, II, and III. Individuals must be marked with individual or cohort marks to prevent the same individuals being counted repeatedly (Appendix III).

Employing artificial shelters is relatively inexpensive and requires little labor compared with other sampling methods. A single individual can easily check hundreds of boards in a day; however, the time required will increase with the number of animals captured and the amount of data taken at each capture.

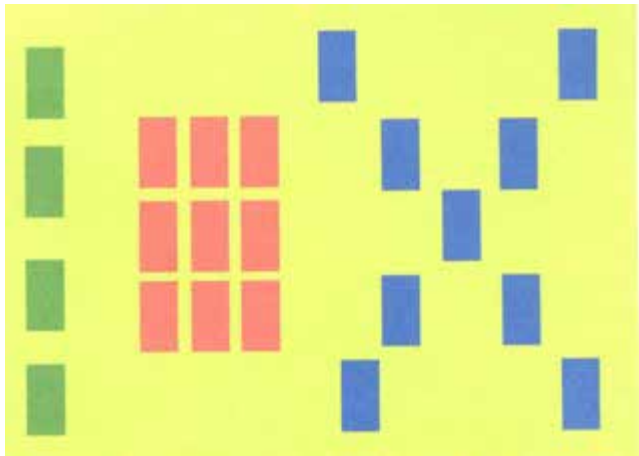


Figure 5-8. Coverboards can be placed in various patterns.

Snake hooks should be used to check under coverboards safely.

Different types of boards attract different species. For example, coachwhip snakes may be captured more often under weathered boards.

Utility

Aspects of sampling amphibians and reptiles with artificial cover have been addressed by several field studies (Grant et al. 1992; Parmelee and Fitch 1995; Sutton et al. 1999; Houze and Chandler 2002; Ryan et al. 2002; Smith et al. 2006). Using artificial cover has several advantages over other sampling techniques. Since coverboards provide suitable habitat for herpetofauna and do not restrict movement, they do not require constant surveillance (e.g., daily checking of pitfall traps) and pose a lower risk of injury or predation to captured animals than live-trapping techniques (Fitch 1992). Employing and maintaining coverboards is also relatively easy and inexpensive when compared with drift fence/pitfall trap arrays. Coverboards may be preferable to time-constrained searches in that they nearly eliminate observer bias and do not necessitate destruction of existing microhabitats to locate animals. A monitoring program which uses this technique in combination with others may be efficient in providing an "early warning system" for population declines of amphibian species (Fellers and Drost 1994).

Artificial cover objects have been effectively used to sample a large number of snake and lizard species. Fitch (1992) found that artificial cover was especially useful for small secretive snake species that were too small to be captured in wire-mesh funnel traps. Artificial cover can be deployed in virtually any terrestrial habitat and is often effective along the margins of aquatic habitats. Obviously, where artificial cover is placed can determine which species may inhabit it. For example, coverboards in areas with sandy soil and high small mammal populations are often quite effective for capturing pine snakes (*Pituophis melanoleucus*) and corn snakes (*Elaphe guttata*). Placing coverboards on the waterline of a stream, pond, or wetland can be very effective for attracting aquatic snake species like the black swamp snake (*Seminatrix pygaea*), various watersnakes (*Nerodia sp.*), and others. However, some reptilian species, primarily turtles and crocodilians, will rarely be detected under coverboards.

Coverboards can also be useful for sampling some amphibians, although the conditions under which they are captured under coverboards may vary from those of many reptiles (e.g., moist, mild conditions for many amphibians; sunny, warm conditions for many reptiles). Salamanders that are terrestrial and commonly found under natural cover objects, such as slimy and two-lined salamanders, can be sampled using coverboards (Fellers and Drost 1994; Harpole and Haas 1999; Houze and Chandler 2002). For example, various species of Coastal Plain salamanders have been effectively captured with this technique (J. W. Gibbons and B. W. Grant, pers comm.). In addition, ambystomatid salamanders will use coverboards as refuge sites. For example, *Ambystoma opacum* will occasionally lay and brood eggs under coverboards on the edge of wetlands. Anurans may occasionally be captured under coverboards, but in most cases additional sampling techniques must be employed because of low capture rates.

Kurt Buhlmann

Example of coverboard layout design for salamanders.

Kurt Buhlmann

Setting up and installing tin coverboards along the edges of an aquatic habitat.

Limitations

Despite the above advantages, coverboards will rarely yield encounter rates as high as drift fence and pitfall trap arrays. Unlike live-trapping methods that

integrate captures over the entire period of deployment, coverboards only capture those individuals using the coverboard at the time it is checked. Several studies have compared coverboard arrays paired with drift fence/pitfall trap arrays (Grant et al. 1992; Sutton et al. 1999; Ryan et al. 2002) and in each case drift fence/pitfall trap arrays caught more individuals of more species than coverboard arrays. It is important to note, however, that coverboards detected species that other techniques did not, emphasizing the point that to detect all species multiple methods must be employed. Furthermore, this technique may not be the most effective method for capturing amphibians in some habitats (e.g., Houze and Chandler 2002; Smith et al. 2006).

Like visual encounter surveys, surveys employing artificial cover will be sensitive to the conditions under which the survey is performed; environmental conditions such as time of day, temperature, rainfall, cloud cover, and humidity will bias the number and identity of species captured (Fellers and Drost 1994). Parmalee and Fitch (1995) found that encounter rates were reduced at midday in the summer, when temperatures beneath artificial cover became too hot for many species. Similarly, Grant et al. (1992) found that encounter rates for reptiles were maximum at ambient temperatures of 20-25 °C, however this may vary depending on local fauna.

Another consideration is that coverboards may not produce data comparable to those gathered by searching natural cover objects. No studies document whether coverboards provide the same physical environment (e.g., temperature, moisture) as do natural cover objects (Houze and Chandler 2002). Likewise, boards will break, warp, and disarticulate over time, leading to possible sampling bias. However, boards can be replaced to ensure microhabitat is kept as consistent as possible.

Considerations

Anecdotal evidence suggests that coverboards may require some time after installation to attract many amphibians and reptiles. However, experimental studies have found relatively rapid use by many species. For example, Grant et al. (1992) found that after two months newly placed tin coverboards had a capture rate equivalent to coverboards that had been in place for a year, and Parmelee and Fitch (1995) found no significant difference in encounter rates of snakes between new coverboards and boards that had been in place for seven years. Some have suggested that litter under coverboards be removed to make specimens more visible. However, Parmelee and Fitch (1995) found that preparing the substrate under cover objects reduced the number of snakes captured, suggesting that removal of debris may be counterproductive. Removing the vegetation underneath a coverboard places it in direct contact with soil, which may increase moisture retention and potentially favor use by some amphibian species (Dorcas pers. comm.).

Checking cover objects using a commercially produced snake hook (see section on hand collection in Chapter 5) or a similar tool (e.g., garden hoe) can greatly reduce the chance of an accidental bite from a venomous snake, centipede, scorpion, fire ants, or other venomous animal while flipping the board. The board can be flipped toward the researcher so the coverboard stands between any animal under the board and the researcher (Fig. 5-9). Tapping the ground with a snake hook can also help locate coverboards that have become buried in leaf litter or pine-straw. Fitch (1992) found that a high percentage of snakes using artificial cover were digesting meals (e.g., 25.6% of *A. contortrix* under artificial cover compared with 6.1% caught in funnel traps). Thus, coverboards may be exceptionally useful if snake diet is of interest.

Fig. 5-9. The proper technique for checking a coverboard.

As discussed above, encounters may be biased by environmental conditions; at a minimum, record the time, temperature, cloud cover, and recent rainfall at the time of capture. Grant et al. (1992) and Fellers and Drost (1994) recommend checking artificial cover objects at many different times of day and under different weather conditions to maximize the number of taxa encountered. However, for comparisons among sites, the conditions under which cover objects are checked should be identical.

Inventory

It may be an effective technique for assessing what species are present in a particular habitat or region, especially when employed along with other techniques. To maximize the number of species captured, this method must be used in a wide variety of conditions, including the time of day, the season, and environmental conditions. To detect the presence of more rare species, the density of coverboards may need to be increased. Likewise, coverboards should be placed in all of the various habitat types within a sampling area to increase the likelihood of capturing those species that are habitat specialists or have small home ranges.

Kurt Buhlmann

Some of the materials needed for coverboard installation: numbered flags, coverboards, rakes.

Monitoring

This technique can provide a reliable index of population size for many herpetofauna, provided that proper standardization measures are taken. It can also prove useful in evaluating the condition and changes in each species' population (Fellers and Drost 1994). Whereas placement of boards in all habitats is a focus when doing an inventory, a monitoring program requires more standardization. Coverboard type, size, and placement (e.g., parallel lines, grids, webs) should be carefully considered as these factors directly affect what is captured and the statistics possible. The conditions under which cover objects are checked should be standardized (e.g., time of day, season, habitat type) as much as possible and recorded along with the current environmental conditions (e.g., temperature, rainfall).

Conclusions and recommendations

Although this method is quite effective, it should be used in concert with a variety of other techniques. It is important to carefully consider experimental design and program objectives when placing coverboards in arrays to ensure that data will be useful during analysis. It is also important to consider the species in question since this technique may not be amenable to collecting all species. Remember to consider environmental conditions, such as temperature and time of day, as well as seasonality and the specific habitat and microhabitat of the sampling area when establishing a sampling regime.

Equipment Checklist

Installation:

- Map of area and design plan
- Measuring tape for setting up the transects, grids, or webs
- Coverboard material options: solid wood boards, plywood boards, corrugated metal strips, tar paper, and horticultural plastic sheeting
- Flagging/stake flags
- Paint or large permanent markers for numbering the coverboards
- Rake for clearing area prior to placing board

Sampling:

- Snake stick or garden hoe
- Data recording materials
- Measuring and marking materials (e.g., scissors, PIT tags)
- Containers
- Equipment for recording basic environmental data (e.g., thermometer, rainguage)

* See Appendix IX for basic equipment lists

POLYVINYL CHLORIDE (PVC) PIPE SURVEYS

Tracey D. Tuberville

Ground-placed or tree-mounted polyvinyl chloride (PVC) pipes are an easy, inexpensive, passive method for sampling hylid treefrogs (see Fig. 5-10). Ground-placed PVC traps are typically about 1 m in total length installed so that about 60 cm remain above the surface. The bottom end of the pipe can be cut at an angle to facilitate insertion into the ground (Moulton et al. 1996; Zacharow et al. 2003). Tree-mounted designs vary widely in both trap characteristics (e.g., length, shape) and placement (e.g., tree-type, tree size, mounting height). In general, however, greatest trapping success has been associated with 60 cm vertical pipes mounted so that the

bottom of the pipe is about 2-4 m off the ground and capped at the bottom so that the pipes can hold water (Boughton and Staiger 2000; Dodd 2003). A small drainage hole should be drilled into the pipe about 15 cm from the bottom. In addition, tree-mounted traps are more effective when placed on hardwood trees vs. pine trees and larger vs. smaller trees (Boughton and Staiger 2000). For both ground-placed and tree-mounted traps, relatively small inner diameters (2 - 5 cm) are recommended (Boughton and Staiger 2000; Zacharow et al. 2003).

John White

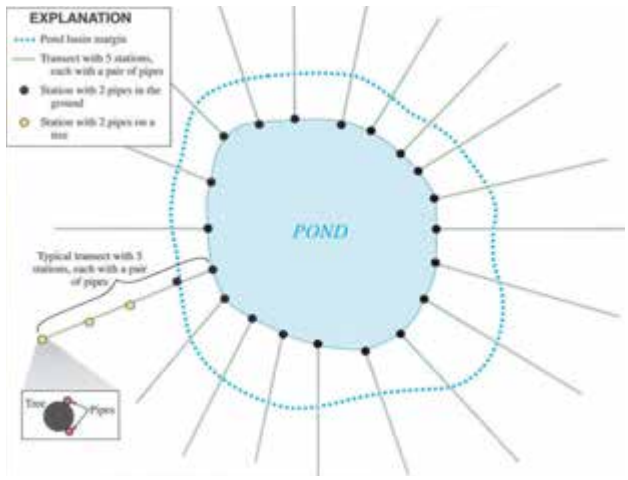
Figure 5-10. Cope's Gray Treefrog (*Hyla chrysoscelis*) on a PVC pipe mounted on tree

Hylid species vary in body size and possibly their preference of retreat placement and other characteristics (Boughton and Staiger 2000; Zacharow et al. 2003), so it may be necessary to use a range of pipe diameters and/or mounting heights to sample all hylid species present. Both tree-mounted and ground-placed traps can be installed in grids or as transects surrounding the perimeter of the breeding site, or radiating from it. PVC pipes should be spaced at 1-5 m intervals for smaller sites and at 5-10 m intervals for larger sites (Moulton et al. 1996; Dodd 2003).

Utility

Because the PVC pipes are a passive trapping technique, the risks of trap mortality and disruption of animal activities are eliminated (Moulton et al. 1996; Boughton and Staiger 2000). Therefore, frequency and timing of trap checks can be flexibly scheduled to target sampling for particular species of interest and can accommodate other project logistics such as personnel, time, and travel constraints. PVC pipes are a particularly appealing sampling technique for remote field sites.

This technique is only effective with hylid treefrogs and trapping success may vary among species depending on trap design and placement and availability of natural refugia (Boughton and Staiger 2000). In at least one study, PVC pipes were not used by *Pseudacris crucifer* (spring peeper) even though the species was known to be present at the site (Boughton and Staiger 2000). However, PVC pipes are currently the most effective method for capturing adult hylids—a group of amphibians that are otherwise notoriously difficult to capture.



Schematic of PVC pipe survey design.

Ground-placed PVC pipes are most successful when used in close proximity to the aquatic breeding sites and can be used to sample grassy, treeless areas. Ground-placed PVC pipes can be used as a supplementary technique to pitfall traps and drift fences, which treefrogs can easily trespass (Dodd and Scott 1994). Tree-mounted pipes, unlike ground-placed pipes, can be used to sample treefrogs during the non-breeding season after frogs have dispersed from the breeding site (Dodd 2003).

Limitations

Unfortunately, PVC pipes are conspicuous, and therefore subject to theft or other disturbances to equipment. This problem can be alleviated by painting pipes a camouflage color (Dodd 2003). Otherwise, the primary limitation to the technique (particularly as a monitoring technique – see below), is the lack of information on potential capture biases.

Inventory

PVC pipes are an excellent technique for evaluating presence/absence (i.e., detection/non-detection) of hylid species. However, calling surveys can be equally effective and may be preferable to the labor

and expense of installing PVC pipes unless additional life history data are needed (Dodd 2003) or scheduling constraints prevent sampling during times when species would most likely be calling. PVC pipes can also be used to determine the distance, direction, and timing of dispersal to and from the breeding sites (Dodd 2003).

Monitoring

Although PVC pipes are a good inventory technique, potential trap biases must be carefully evaluated before implementing them as a monitoring technique. Because frogs frequently take up residency in pipes, it is necessary to mark individuals to distinguish new captures from recaptures (Dodd 2003) in order to estimate species abundances using mark-recapture models. Additionally, although PVC pipes could potentially be used to collect additional life history data such as sex ratios and size distributions (Dodd 2003), preliminary studies must first be conducted to identify size- or sex-related capture biases (Boughton and Staiger 2000). Finally, it may be inappropriate to make comparisons among study sites (even when the same protocols are used at each site) due to different species composition at the sites, differences among species in their proclivity to use artificial refugia, and the local characteristics and availability of natural refugia (Dodd 2003). Therefore, PVC pipes may be most appropriately used as a monitoring technique for monitoring at a single site, whether documenting temporal changes or differences among habitat types or management treatments within the site. Additionally, researchers must pay close attention to whether or not territoriality or site-defense by individuals affects the distribution of animals in PVC pipes, possibly hampering use of PVC pipe-collected data for relative abundance estimates. Furthermore, there may be more amphibians in the immediate vicinity than suggested by PVC pipes if all pipes are found to be in use and are a limiting factor.

Equipment Checklist

Installation:

- Map of area and design plan
- Flagging, stake flags for marking study area
- PVC piping (2 - 5cm diameter)
- Caps for lower end of each pipe
- Mounting materials (e.g., mounting brackets or bungee cords)
- Drill
- Paint (for camouflaging the pipes)

Sampling:

- Data recording materials
- Measuring and marking materials (e.g., metric ruler, PIT tags)
- Containers

* See Appendix IX for basic equipment lists

LEAF-LITTERBAG SURVEYS

Thomas M. Luhring

Salamanders are difficult to monitor in part due to their cryptic and fossorial nature as well as their lack of vocalizations (Jung et al. 2000). The sampling of stream habitats, in particular, differs in many aspects from sampling wetland habitats (Mitchell 2000). Litterbags have been commonly used over the past three decades as a standard technique for estimating leaf litter breakdown in streams (Benfield 1996; Peterson and Cummins 1974). Recently, this technique has been adapted to sample for stream-dwelling salamanders (see Pauley and Little 1998).

Tom Luhring

Spring salamander (*Gyrinophilus porphyriticus)*

Waldron et al. (2003) implemented this technique to successfully inventory various streams throughout the Great Smoky Mountains National Park. Their basic design consists of a square (50-90 cm per side, with 70 cm x 70 cm being the optimal size) piece of plastic netting with 1.9 cm mesh. Small rocks are placed on the netting in the field and covered with leaves before the corners are brought together and bound with cable ties to form the litterbag (Fig. 5-11). A piece of flagging tape is tied to the bag to aid in finding it during future surveys. Many variations on this design have also been successful and are featured in a number of studies (U.S.G.S., Manager's Monitoring Manual, (www.pwrc.usgs.gov/monmanual/techniques/leaflitter.htm, accessed 24 August 2011).

Finished bags are placed in the stream at regular intervals and care must be taken to weigh or stake the bags down so that they do not float away during periods of high flow (Pauley and Little 1998; Dodd 2003). Waldron et al. (2003) also tethered their bags to nearby roots or branches with monofilament fishing line to secure them in place. After an acclimation period of a couple of weeks, each bag is checked by

placing a dipnet underneath and lifting the bag into a bucket of water. To extract the salamanders from the bag, dip the bag repeatedly in the bucket and then pour the water through the dipnet (Jung et al. 2000), or alternatively, shake the bag over a white dishpan for 15 seconds (Dodd 2003). The salamanders are then processed and the bags are placed back into the stream.

Utility

Leaf litterbags are a non-destructive, passive, and efficient technique for sampling stream-inhabiting salamanders that may not be typically encountered during other types of surveys such as time-constrained or area-constrained searches (Waldron et al. 2003). Litterbags are particularly useful for sampling larval salamanders. However, surveys targeting larval salamanders should completely submerge litterbags in order to discourage colonization by predatory adults. The ease of using and deploying leaf litterbags for sampling streams as well as the relatively low cost per bag (around US $2.00) makes this technique applicable to large-scale standardized sampling efforts (Chalmers and Droege 2002; Waldron et al. 2003).

J. C. Maerz and T. Pierson

Figure 5-11. This litter bag is made of Tenax plastic deer netting and placed in a stream. The mesh is ~1/2 inch x 1/2 inch. The bag's final dimension is 50 cm X 25 cm. It is sealed on all sides by a fold that creates a full edge, and secured on three sides with zip ties. One side is sealed with binder clips that can be removed to add or remove litter. The bag has been placed near the stream edge with rocks to secure it in place.

Limitations

While effective at surveying for the presence of salamander species, this technique is not capable of indexing population sizes (Chalmers and Droege 2002; Dodd 2003; Waldron et al. 2003). Another inherent problem that should be taken into consideration is that identifying larval salamanders is often difficult for inexperienced observers (Waldron et al. 2003). Furthermore, litterbags may not be effective during seasons where leaf packs occur naturally in high abundance and can often be washed away and lost during times of high water flow. Seasonal use of the stream habitat varies by salamander species and may bias results depending on the time of year in which litterbags are deployed. Chalmers and Droege (2002) suggested that litterbags may need to accumulate prey items in order to become better at attracting salamanders.

J. C. Maerz and T. Pierson

Biologists sampling a leaf litterbag.

Inventory

Leaf litterbags are an effective way to detect species presence of stream-inhabiting salamanders and are a particularly efficient way to sample their larvae (Dodd 2003; Waldron et al. 2003). The results from litterbag surveys are comparable to other surveys (Jung et al. 2000), and can have better detection rates of secretive or rare species than techniques such as time-constrained or area-constrained (i.e., transect or quadrat sampling) searches (Waldron et al. 2003).

J. C. Maerz and T. Pierson

A salamander captured in a leaf litterbag survey.

Monitoring

This technique is not currently ready for use as a stand-alone monitoring program as it is incapable of tracking population indexes (see limitations above). Chalmers and Droege (2002) specifically tested the ability of litterbags to provide a population index in Maine populations of the northern two-lined salamander (*Eurycea bislineata*). They varied densities of *E. bislineata* within enclosures containing litterbags at three different streams and were unable to detect an overall significant relationship between population sizes and the number of salamanders present in each litterbag. Dodd (2003) also found that litterbags are unable to provide an estimate of overall abundance without the implementation of additional measures such as a mark-recapture regime.

Conclusions and recommendations

Leaf-litterbag sampling for salamanders is a relatively new technique, and one that has proven particularly useful for sampling larval salamanders in some streams. It can be especially useful for detecting the presence of the more rare and elusive stream-dwelling species. However, keep in mind that certain stream conditions (e.g., very high water flow) and the sampling season chosen will influence the results. Recent research has shown that this technique is not suitable as a primary monitoring method, mainly because there is no relationship between population parameters and litterbag captures.

Equipment Checklist

Installation:

- Map of area and design plan
- Measuring tape
- Flagging/stake flags
- Square plastic netting with small mesh (for making bags)
- Rocks and leaves
- Cable ties
- Flagging to tie onto bags
- Stakes or monofilament fishing line to secure bags

Sampling:

- Dipnet
- Bucket
- White dishpan
- Data recording materials
- Measuring and marking materials (e.g., scissors, metric ruler)
- Containers
- Equipment for recording environmental data (e.g., water depth, velocity, temperature

* See Appendix IX for basic equipment lists

AUTOMATED RECORDING SYSTEMS

John D. Willson

A variety of systems are available that allow automated recording of anuran vocalizations (also see Chapter 4 - Automated Data Acquisition). Recorders vary in complexity, capability, and cost. On the high-end, data logger-based systems are capable of recording multiple channels of data, including environmental data and sound recordings (for further information see Peterson and Dorcas 1994). Alternatively, a variety of less-expensive models have recently become available that combine a recorder, timer, microphone, and voice-print alarm clock. These "frogloggers" (www.frogloggers.com) are capable of recording more limited amounts of data on a set time schedule and are relatively easy to construct and operate (Fig. 5-12). Recently, digital technology has improved the data capacity, quality of recordings, size and cost of these models.

Bedford Technical

Figure 5-12A. Components of an automated recording device (i.e., froglogger).

John Jensen

Figure 5-12B Froglogger set up in field and camouflaged in a tree.

Regardless of the specific design, recorders can be set to record on regular intervals, yielding compilations of calling activity over relatively long timeframes. Recordings are replayed in the laboratory to evaluate species present and calling intensity at each time interval. Several studies have also used background noises (e.g., the sound of rain hitting the microphone) to generate useful covariates of anuran calling activity (e.g., Bridges and Dorcas 2000; Todd et al. 2003; Mohr and Dorcas 1999).

Utility

Automated recording devices have several advantages over traditional observer-based anuran call surveys: 1) they allow for 24-hour monitoring of potential sites, 2) they allow several sites to be monitored simultaneously without observer bias, and 3) they free up the observer for other activities (Peterson and Dorcas 1992; 1994). For simple inventory or common species (see section on "auditory surveys"), automated recorders may not be necessary, but the ability to monitor at intervals over long time periods makes automated recorders invaluable for inventorying rare or explosively-breeding anuran species, for surveying remote locations that are difficult to access regularly, and for sampling at many locations simultaneously. Automated recorders are particularly well-suited for intensive investigation of temporal calling patterns (i.e., reproductive activity) at specific locations and have been used to document previously unrecorded calling patterns in some species (Bridges and Dorcas 2000).

Limitations

Data logger-based recording systems allow recording of multiple channels of data and can maximize space by recording on complex intervals (e.g., only at night), but are expensive (about US $1700; Peterson and Dorcas 1994). Timer-based "frogloggers" that use digital recorders are less expensive (range from US $380-US $650) but require more frequent upkeep (replacement of batteries) and cannot record environmental variables. The cost effectiveness of automated recorders varies greatly depending on the goals of the study. If rapid inventory is the primary objective, observer-based surveys allow for assessment of large areas in a very short time period but yield limited data at each locality (e.g., only a "snapshot" of calling activity at one point in time). If intensive inventory or monitoring is the goal of a program, the high initial cost of automated recorders may be offset by reduced spending on mileage and personnel that would be necessary for observer-based surveys. An additional consideration when using automated recorders on public lands is that they may be a target for theft or vandalism.

Inventory

Calling surveys, in general, are one of the best methods for inventory of anurans (see section on "auditory surveys"), often yielding many species with relatively little effort. Automated recorders allow for sampling of multiple locations simultaneously and thus can be extremely useful when the area of interest is very large or when locations are geographically separated. By programming short recording intervals over an extended time period you can be reasonably sure that all species breeding at a given locality will be recorded. Generally, when using automated recorders for inventory, recorders should be placed in habitats most likely to harbor high anuran diversity (e.g., fishless wetlands), and sampling a variety of aquatic habitat types will often yield the most complete species list. Because breeding seasons vary by species, sampling for multi-species inventories should be conducted at several intervals throughout the year (e.g., winter, spring, summer). Alternatively, when specific species are of concern, recorders may be set to record frequently during the focal species' suspected breeding season. It is important to remember that many species do not breed successfully each year in any given habitat. Thus, absence of a species in a given year does not necessarily mean that it is not present at the site. An additional benefit of automated recorders is that recorded calls provide a voucher for each species without taking live specimens.

Monitoring

Designing a monitoring scheme based on call surveys requires a standardized sampling scheme that can be repeated over time (see Chapter 3). For this type of survey, automated recorders have obvious advantages over observer-based methods. Automated recorders can be used to sample numerous locations simultaneously with minimal observer bias. Additionally, recorders can be programmed to record short samples on regular intervals over long periods of time, allowing for a much more complete characterization of temporal calling patterns than would be possible via observer surveys (e.g., Bridges and Dorcas 2000). Although the specifics of recording frequency and duration will vary by species and study goals, generally recording at several regular intervals each night (or day) over the entire suspected breeding season will allow thorough characterization of breeding activity of the focal species. Ideally, call surveys can be supplemented with larval sampling to determine if reproduction was successful or with capture of metamorphosing juveniles to estimate recruitment into the population.

Conclusions and recommendations

Automated recording systems are a useful complement to anuran call surveys. Automated recorders allow for simultaneous recording of multiple locations and may be particularly useful for inventory of rare or explosively-breeding species, for sampling at inaccessible or widely-separated locations, or for intensive monitoring at specific sites. Thus, although somewhat expensive, the cost of automated recorders may be far less than mileage and personnel expenses of observer-based surveys and this technique should be considered whenever intensive inventory or monitoring of anurans is a priority.

Equipment Checklist

Installation:

To build a froglogger: cassette recorder, timer, microphone, voice-print alarm clock (alternatively, a froglogger can be purchased)
Datalogger can be used to record (instead of using a froglogger)
Materials for mounting the device in a secure location

Sampling:

Data recording materials (when listening to recorded surveys: datasheet, pencil)

* See Appendix IX for basic equipment lists

INTENSIVE PASSIVE SAMPLING

AQUATIC AND TERRESTRIAL FUNNEL TRAPPING

John D. Willson

Although numerous trapping methods have been developed for capturing reptiles and amphibians, funnel trapping remains a standard method for many species. Various types of funnel traps are available but all function on the same basic principle: animals are directed through a small opening in the trap via a funnel or ramp and, once inside, are unable to find their way out. This section describes the general utility of funnel trapping as a sampling technique and details several popular varieties of funnel traps for different applications.

Utility

Funnel trapping is one of the most effective general methods for capturing reptiles and amphibians and it is particularly useful for sampling rare or cryptic species. Funnel traps vary in design (Fig. 5-13) with a trap type available for most species or habitats. Trapping has several advantages over other sampling methods for reptile and amphibian monitoring. Because trapping schemes can be standardized, observer bias is minimized. Additionally, because trapping integrates captures over the entire time that traps are set (rather than being a snapshot of the time when the observer is at the site), this method is less sensitive to biases resulting from temporal variation in activity or detectability than methods that only sample for a short duration. Finally, in addition to generating relative abundance data (e.g., captures per trap-night), funnel trapping can be used in conjunction with mark-recapture techniques to estimate population parameters.

Limitations

A major limitation of funnel trapping is that it requires a substantial investment of time and equipment. Traps are expensive and must be checked regularly to avoid mortality of trapped animals.

Inventory

Funnel trapping is a useful method for herpetofaunal inventory, particularly when used in conjunction with other methods. Perhaps the greatest strength of funnel trapping for inventory is that this technique is virtually the only way to capture a variety of rare or cryptic species (e.g., highly aquatic or fossorial species) that are often overlooked by other survey methods. As assessing population status is not a priority for inventory, trapping for inventory purposes should be carried out with the goal of maximizing captures, even at the cost of standardizing sampling methodology. Thus, for inventory, trapping should focus on habitats and times when animals are most likely to be active (e.g., during rainy periods or in highly vegetated aquatic habitats). Additionally, trapping multiple habitat types will maximize the chances of encountering a variety of species. Although trapping is an effective method for sampling many species, capture rates are generally low (especially for snakes and uncommon amphibians). Thus, high intensity sampling will be necessary to detect all species present. Although efficacy varies by species and habitat we generally recommend several hundred trap-nights spread across seasons if a reasonably complete species inventory is desired.

Monitoring

Funnel trapping can be an extremely useful technique for herpetofaunal monitoring because it is relatively insensitive to a variety of sampling biases. For moni-

toring, a standardized trapping scheme should be designed that can be repeated, allowing comparisons of capture rates over time to assess population status. The goal in monitoring should be to replicate sampling intensity at several time intervals and, ideally, to sample equally across the population of interest. Thus, it is generally desirable to systematically or randomly place arrays of traps throughout suitable habitat, even at the expense of reducing capture rates and/or placing traps in less accessible locations. Trap capture rates from several trapping occasions can be compared to infer population status. However, when comparing capture rates it is important to remember that capture rates are measures of relative abundance and may not reflect true population size. At any given time, capture rates are a function of population size, level of activity, and propensity of a species to enter and remain in traps. Thus, capture rates should be compared with caution and ideally this technique should be used in conjunction with mark-recapture techniques to estimate changes in population size and detection probability over time.

Jeff Hall

Figure 5-13A. Minnow trap set in a wetland for aquatic snakes.

Tom Luhring

Figure 5-13B Trashcan trap set in water.

Kurt Buhlmann

Figure 5-13C Hoop trap set in a wetland so that captured turtles can breathe.

Kurt Buhlmann

Figure 5-13D Setting a fykenet in a gum swamp.

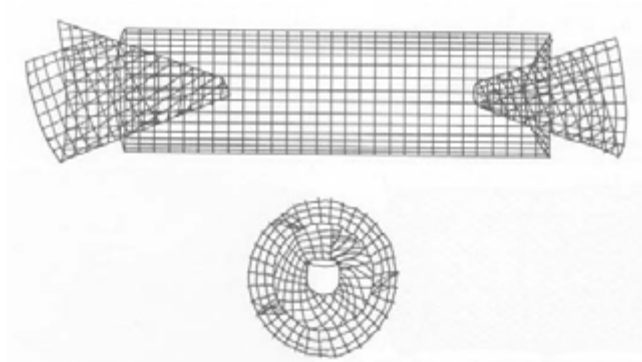


Figure 5-13E Side and head-on view of terrestrial funnel trap parts that can be installed along a drift fence (Jones 1986).

Kurt Buhlmann

Figure 5-13F Wooden funnel traps capture snakes along a drift fence.

DESCRIPTION OF FUNNEL TRAP APPLICATIONS

Small cylindrical and rectangular aquatic funnel traps – Aquatic funnel trapping is an effective method for capturing aquatic snakes, and may be the most effective method for inventory of secretive species that do not conspicuously bask (e.g., mud *Farancia abacura*, rainbow *F. erytrogramma*, swamp *Seminatrix pygaea*, and crayfish snakes *Regina sp.*; Willson et al. 2005). Additionally, aquatic funnel traps readily capture amphibians and are a good alternative to systematic dipnetting (see section on dipnetting) for standardized sampling of aquatic species that are infrequently captured in dipnets (e.g., large salaman-

ders, *Siren* and *Amphiuma*, adult frogs, and adult stream-dwelling salamanders). Aquatic funnel traps vary in size and design but the most readily available and effective varieties are commercially available minnow, crawfish, or eel traps (Fig. 5-13A; Adams et al. 1997). These traps may be cylindrical or rectangular but are typically double-ended and constructed of steel hardware cloth, plastic, or nylon mesh. Recent research has shown that steel and plastic traps capture more snakes than nylon mesh traps (Willson et al. 2005). Steel traps are easier to set than plastic traps and are effective in most situations. Additionally, if large species are targeted (e.g., large female watersnakes, *Nerodia sp.*) the funnel openings can be widened slightly, allowing larger snakes to enter (Keck 1994). However, as small snakes are easily entangled in the mesh of steel traps, plastic traps should be used when small species (e.g., swamp snakes, *Seminatrix pygaea*) are suspected to occur (Willson et al. 2005). Additionally, the smaller mesh of plastic traps makes them more effective for sampling small amphibians (e.g., larval salamander and tadpoles). Although ineffective for snakes, an inexpensive funnel trap for small amphibians can be made by inverting the top of a plastic soda bottle and anchoring the trap to the substrate with a garden stake (Griffiths 1985; Willson and Dorcas 2003b).

Aquatic funnel trapping is useful for sampling nearly any aquatic habitat but is most effective in shallow water bodies with abundant vegetation. Traps are generally set with approximately ¼ of the trap above the water, allowing captured animals access to air. Alternatively, Casazza et al. (2000) used styrofoam floats to allow for trapping of deep-water habitats without drowning captured animals. Traps are often set along shorelines, submerged logs, or other obstructions that may aid in guiding animals into traps (Fitch 1987). Alternatively, traps may be set along artificial barriers (e.g., nylon silt fencing) to increase capture rates (Willson and Dorcas 2004). Most amphibians presumably enter traps "by chance," but snakes are actively attracted to prey within traps (Winne 2005). Thus, although in many areas amphibian and fish bycatch may be sufficient to "bait" traps, purposefully baiting traps with fish or amphibians (live or dead) may increase capture rates (Keck 1994; Winne 2005). To avoid inadvertently killing trapped animals, traps must be checked regularly, generally at least once daily. As many snakes can readily escape from funnel traps if given sufficient time, checking traps more frequently will increase snake capture rates per unit time (Willson et al. 2005). In warm months, traps are best checked in the morning as animals captured at night may die when the sun heats shallow water around the trap.

Trashcan traps (Thomas M. Luhring) – Trashcan traps are a type of large aquatic funnel trap that can be used for subsurface sampling (up to 70 cm) of fishes, reptiles and amphibians (Fig. 5-13B). These traps are created by installing funnels into the sides of commercially available 32-gallon Rubbermaid® Roughneck® household trashcans ($11.92 in 2008). Trashcan traps are safe for obligate air-breathers and require a substantially lower financial investment than commercially available crayfish traps (Luhring and Jennison, 2008).

The size, number, and location of the funnels on the trashcan are determined by the objective of the project. Funnels placed along the lower sides (see Fig. 5-14) are effective for sampling permanently aquatic salamanders such as greater sirens (*Siren lacertina*), lesser sirens (*Siren intermedia*), and two-toed amphiumas (*Amphiuma means*) and for highly aquatic snake taxa such as *Farancia, Regina, Nerodia,* and *Seminatrix*. Trashcans with funnels located near the top of the sides are useful for sampling the top stratum of the water column and are most effective when floating next to large vegetation mats with the funnels at the same level or slightly below the vegetation. Funnels of various sizes can be placed at any height along the side of the trashcan to sample a finite stratum of the water column. Screen windows (optional) can also be installed along the sides to permit air flow and water circulation. The screen also functions as a fine filter through which to drain the contents of the trashcan when pulling it out of the water. Draining water through the side screen permits the capture of smaller animals (e.g., small amphibian and fish larvae) that would otherwise fit through the mesh of the funnels.

Trashcan traps have higher capture rates than minnow traps for several bottom-dwelling vertebrates and regularly capture multiple species of reptiles and amphibians at the same time (Luhring, unpublished data). The smaller interior size of minnow traps may limit the number of animals that can be captured inside a single trap at one time. For these reasons, trashcan traps may be particularly helpful for an inventory. As the use of trashcans as traps is a relatively new technique, the ability to determine population size without mark-recapture data is yet to be determined. However, trashcan traps do have a higher recapture rate than minnow traps for greater sirens (Luhring, unpublished data) and would most likely be useful for mark-recapture efforts targeting aquatic vertebrates.

Although advantageous in many ways, it is important to keep in mind that the traps are somewhat heavy, bulky, and cumbersome to carry for a long distance.

Likewise, although cheaper than crawfish traps, trashcan traps do require a considerable amount of time to assemble (30 minutes or more per trap). The greatest asset of the trashcan trap is its adaptability as it can be tailor-made to address particular questions and inventory or monitoring objectives.

C. A. Jennison

Figure 5-14. A trashcan trap for capturing sirens and amphiuma.

How to build a trashcan trap

1. Determine the size of the funnel (depends on your study objectives).
2. Trace the funnel openings and window openings on the outside of the trashcan using a template and a permanent marker (to ensure a tight fit, make sure opening is slightly smaller than the largest end of the funnel).
3. Cut out the openings with a utility knife.
4. Create small holes around the entire perimeter of the openings using an electric drill.
5. Place the funnel into the opening and cinch it in place with cable ties
6. If aluminum window screening is used for the window, small cable ties can be forced through the screen
7. To improve drainage when lifting the trashcan out of the water, drill holes through the bottom of the trashcan with the electric drill.

Note: Cost of one 32-gallon Rubbermaid® Roughneck® household trashcan was $11.92 in 2008.

Baited Hoop-nets for aquatic turtles – Large aquatic funnel traps or "hoop-nets" are an extremely effective method for capturing many aquatic turtle species (Iverson 1979; Lagler 1943; Legler 1960). Although almost any turtle species may occasionally be captured with such traps, they are most effective for sampling highly aquatic, carnivorous species (e.g., sliders *Trachemys scripta*, mud *Kinosternon* sp.*,* musk *Sternotherus* sp., snapping *Chelydra serpentina* and *Macrochelys temminckii*, and painted turtles *Chrysemys picta*). Hoop nets are commercially available in a variety of sizes and most are made of twine or mesh (Fig. 5-13C). Traps are easily set in shallow water but care must be taken to ensure that some part of each trap remains above water, allowing captured turtles access to air. Additionally, traps should usually be tied to a stake, tree, or other object to prevent large turtles from dragging traps into deep water. Hoop nets can be baited with nearly any type of fish or meat, however, a tin of sardines with the top slightly opened is a clean and readily-available bait (Ernst 1965). Traps should be checked at least once daily to avoid drowning turtles. Small mesh hoop-nets used primarily for sampling turtles will also occasionally capture large aquatic salamanders (e.g., *Siren* and *Amphiuma*) and large aquatic snakes (e.g., water *Nerodia* sp., mud *Farancia abacura*, and rainbow snakes *F. erytrogramma*).

Interruption traps and fyke nets for aquatic turtles – Aquatic turtles that are not captured effectively in baited hoop-nets can often be captured in "interruption traps" and "fyke nets." These traps are unbaited, but use nets or natural channels to guide turtles into funnel traps, essentially working like an aquatic drift-fence. Interruption traps are effective for sampling turtle species that inhabit shallow wetlands with abundant vegetation (e.g., bog *Glyptemys muhlenbergii*, spotted *Clemmys guttata*, and wood turtles *Glyptemys insculpta*). Interruption traps vary in design but may include simple unbaited hoop-nets, swing door traps, or pressure plate traps. Traps are set in shallow water in natural channels or bottlenecks in mud or vegetation, capturing turtles as they move along these routes. Rather than using natural channels, fyke-nets use nets to guide turtles into a funnel trap (Fig. 5-13D; Vogt 1980). These traps are extremely effective for trapping many hard-to-capture species in open water situations (e.g., chicken turtles *Deirochelys reticularia*, cooters *Pseudemys* sp., diamondback terrapins *Malaclemys terrapin*, softshells *Apalone* sp., and map turtles *Graptemys* sp.; Buhlmann 1998; Vogt 1980). Although both interruption traps and fyke nets are most commonly used for species that do not respond well to baited traps, these traps also readily capture many other species (Vogt 1980) and, like other hoop nets, may occasionally capture aquatic snakes and large amphibians.

Terrestrial funnel trapping – Funnel traps can also be effectively used to trap reptiles and amphibians in terrestrial situations and are often used in conjunction with pitfall traps along drift fences (see techniques section on drift fences). As with aquatic funnel traps, designs for terrestrial funnel traps vary. The most popular variation consists of a wire hardware cloth cylinder with inverted hardware cloth funnels pinned into each side (Fitch 1987; Fig. 5-13E). This type of trap can be set along a drift fence (Fig. 5-15) or placed along natural barriers such as rock outcrops or fallen logs, with open ends pressed against the ground to direct animals into the funnel (Fitch 1987). Traps are best set in a shaded location, and placing a board, piece of plastic, or similar artificial shade over the trap can make the trap more attractive to animals and protect captured animals from heat and rain (Enge 1997). As with other traps, terrestrial funnel traps must be checked each morning and preferably again in the late afternoon to avoid death of captured animals. In areas where amphibians are abundant and temperatures are high, it may be necessary to devise methods for providing moisture to captured animals (e.g., a moist sponge, water bowl, or moist hide-box in each trap).

Jamie Bettaso

Figure 5-15. Mesh funnel traps set next to a terrestrial drift fence.

Wooden box funnel traps have recently been designed that function similarly, but are far easier to check than hardware cloth traps and may be more effective at retaining many reptiles, particularly snakes (Fig. 5-13F; Burgdorf et al. 2005). Box traps, however, are time consuming and expensive to construct and are only effective when used in conjunction with an artificial drift fence. Although more difficult to construct and check than pitfalls, box traps have proven very effective for trapping almost any snake, lizard, or amphibian species (e.g., Burgdorf et al. 2005; Campbell and Christman 1982; Enge 2001; Greenberg et al. 1994; Todd et al. 2007). Variations of the basic box funnel trap have been developed, such as the large, square-shaped box traps that are watered daily so that they do not need to be checked every day. When used along a drift fence, funnel traps often capture many species that seldom enter pitfall traps (e.g., treefrogs and large snakes) and can be very effective for capturing rare and/or secretive species (Burgdorf et al. 2005; Campbell and Christman 1982; Enge 2001; Greenberg et al. 1994; Himes 2000; Jenkins et al. 2003; Zappalorti and Torocco 2002; Ryan et al. 2002).

Conclusions

Aquatic and terrestrial funnel trapping can be useful methods for sampling a variety of reptile and amphibian species in many habitats. Although somewhat expensive and labor intensive, trapping is perhaps the best technique for inventory of aquatic and cryptic terrestrial species. In addition, trapping is among the most repeatable of sampling methods, making it an invaluable tool for long-term monitoring of many species.

Equipment Checklist

Installation:

Map of area and design plan
Flagging/stake flags for marking trap locations

Trap building materials (many can also be purchased)

Aquatic traps:
- Minnow, crawfish, and eel traps can be purchased
- Steel hardware, cloth, plastic, or nylon mesh; cable ties and wire cutters
- Floats for keeping aquatic traps at surface (if necessary)
- Plastic soda bottle, garden stake to secure in place (for salamanders)

Trashcan traps:
- Trashcan (size can vary, but 32-gallon household trashcan recommended)
- Cable ties
- Template for size of funnel
- Utility knife
- Electric drill
- Aluminum window screening
- Funnel (either handmade from 1/4" hardware cloth or salvaged from other minnow traps)
- Permanent marker

Hoop nets:
- Purchase hoop nets
- Stake and rope for securing in place
- Bait for hoop nets (fish or meat, sardines recommended)

Interruption and fyke net traps:
- Purchase interruption traps and fyke nets
- Stakes and rope for securing in place

Terrestrial traps:
- Wire hardware cloth, heavy-duty stapler, cable ties
- Wooden box funnel traps (wood, stiff mesh for funnel, nails, hinges, latch)

Sampling:

Data recording materials
Measuring and marking materials (e.g., scissors, PIT tags, metric ruler)
Containers and cloth bags
Cable ties (for repairing traps as needed)

* See Appendix IX for basic equipment lists

TERRESTRIAL DRIFT FENCES AND PITFALL TRAPS

Brian S. Metts and Gabrielle J. Graeter

Terrestrial drift fences and pitfall traps have been used for many years to sample a variety of amphibian and reptile species. Basic design for a terrestrial drift fence is a straight fence buried slightly below ground standing up to 50 cm high. Drift fences are typically made of aluminum flashing, although other materials may be used (e.g., plastic silt-fencing, chicken wire, hardware cloth). Pitfall traps are then placed at prescribed intervals alongside the fence and buried flush with the ground (Fig. 5-16A). Animals traveling across the landscape surface are diverted by the drift fence and directed into pitfall traps. In most cases, drift fences are used as part of a mark-recapture study so that individuals can be distinguished from one another. Funnel traps placed along-side drift fences are also useful for capturing amphibians and reptiles (Enge 2001; see aquatic and terrestrial funnel trapping section).

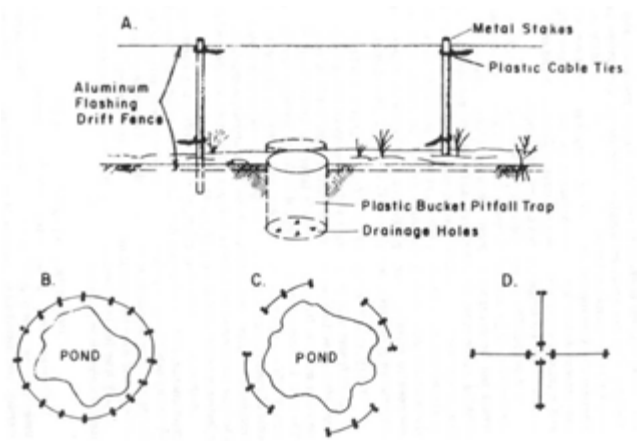


Fig. 5-16A-D. The basic design for a terrestrial drift fence with pitfall traps, and spatial arrangements. (A) Materials needed and recommended set-up for each drift fence segment; (B) top view of a drift fence that completely encircles a pond or wetland with pitfall traps at regular intervals; (C) top view of a partial fence, in that sections of drift fence can be installed around a pond or wetland; and (D) top view of an X-shaped fence array in which the four arms intercept migrating herpetofauna.

Drift fences can be divided into three types or spatial arrangements: straight-line, continuous, and partial drift fences. Continuous and partial drift fences are most often used to sample around wetlands, and as their names imply, either completely or partially encircle the wetland (Fig. 5-16B and 5-16C; Dodd and Scott 1994). Straight-line drift fences are often set up in X or Y-shaped arrays, and are used for sampling upland habitats (Fig 5-16D; Corn 1994). See Corn (1994) and Dodd and Scott (1994) for detailed explanations of the equipment required, design, and installation of drift fences and pitfall traps.

Utility

Drift fences with large pitfall traps have proven effective for sampling most amphibians and squamate reptiles (Nelson and Gibbons 1972; Semlitsch et al. 1981; Bury and Corn 1987; Hanlin et al. 2000; Enge 2001; Russell et al. 2002; Ryan et al. 2002; Todd et al. 2007). Fences, including those of chicken wire construction, are effective for catching turtles regardless of whether or not pitfall traps are present (Bennett et al. 1970; Gibbons 1970; Wygoda 1979; Burke et al. 1998).

Gabrielle Graeter

View into a pitfall trap full of spadefoot toads (*Scaphiopus holbrookii*). Traps can catch high numbers of individuals during peak breeding events.

Gibbons and Semlitsch (1981) and Todd et al. (2007) provide a discussion of several advantages and limitations associated with using drift fences. Gibbons and Semlitsch (1981) concluded that aluminum flashing was superior in comparison to other fencing materials because animals do not bypass it as easily and because it does not deteriorate or rust. Gibbons and Semlitsch (1981) and Todd et al. (2007) also concluded that 20 L plastic buckets were more effective than smaller pitfall traps at capturing many species of reptile and amphibians. However, terrestrial funnel traps may be needed to effectively capture many reptiles, particularly large snakes (Todd et al. 2007). Smaller volume traps have been used effectively for certain species of amphibians (Shoop 1965; Gill 1978; Douglas 1979; Todd et al. 2007), but double-pit systems may aid in trapping larger reptiles (Friend 1984). Enge (2001) pointed out that funnel traps will effectively capture most species, but that pitfall traps may need to be used in xeric upland habitats to increase the chance of capturing semifossorial or fossorial reptiles.

Kurt Buhlmann

This drift fence, constructed of hardware cloth, was used to survey for bog turtles.

Drift fences have been proven effective in most terrestrial habitats for sampling amphibian and reptile populations. However, capture rates and effectiveness of this technique can vary greatly among sites. Drift fences are particularly beneficial in determining species richness and relative abundance, and aid in detecting the presence of species that are difficult to document. For example, a study conducted in diverse habitats in Florida captured semifossorial lizard (e.g., *Eumeces egregius*) and snake (e.g., *Tantilla relicta*) species in pitfall traps (Enge 2001).

Ryan et al. (2002) sampled herpetofauna in three terrestrial habitats (recent clearcut, pine plantation, and mixed pine-hardwood forest) using three survey techniques, and found drift fences to be more effective than coverboards and time-constrained searches. The drift fence technique revealed the presence of more species and individuals in every habitat and was the only one to detect species dissimilarity among habitats.

Kurt Buhlmann

Terrestrial drift fences are effective for catching turtles regardless of whether or not pitfall traps are present because most turtles remain near or up against the fence.

Limitations

A major drawback of this technique is the investment of both time and funds to set up and run a drift fence study (Dodd 2003). This technique requires many tools (e.g., shovels, post-hole digger, possibly a motorized trencher), equipment (e.g., aluminum flashing, buckets or tin cans, sponges, shade covers), and hard work to install and maintain the pitfall traps and fences. Once installed, the traps must be checked daily, ideally before noon to prevent desiccation of animals in hot and dry weather. This can be very time-consuming if travel between different sites is necessary. When checking daily is impractical and waiting longer will not cause harm to the animals, Corn (1994) recommends checking at least every three days.

Differences in trap avoidance or attractiveness among species or individuals can lead to biases in the data. For example, southern toads (*Bufo terrestris*) have been known to hop around a pitfall trap and avoid capture (Graeter 2005), whereas leopard frogs (*Rana sphenocephala*) have shown a preference for the buckets, presumably for their moist and warm environment (Shields 1985). This can be problematic if one wants to compare relative abundance between different species, but not as serious a problem if comparing among one species at multiple sites.

Differential climbing or jumping ability of herpetofauna may also result in capture rates that are not representative of relative abundance. Several limitations and important considerations for using drift fences and accompanying pitfall and funnel traps related to this issue are discussed in greater depth in Todd et al. (2007). For example, many lizards, treefrogs, and ranids can climb or jump out of the pitfall (e.g., *Anolis carolinensis, Gastrophryne*, and most *Hyla* and *Rana*; Corn 1994). Fitting a plastic collar to the top of pitfall traps may keep some individuals from crawling and jumping out of traps (Vogt and Hine 1982). One solution for the escape of ranids is to use deeper pitfalls, thereby reducing the number of frogs that are able to jump out. Larger snakes will also escape medium and small sized pitfall traps. For example, more than 100 subadult rainbow snakes (*Farancia erytrogramma*) were captured in an area where large adults had never been seen, indicating that the larger-bodied snakes had escaped the traps (Gibbons et al. 1977). In these cases, double-pit systems may be useful. Double-pit systems are recommended for catching more species in a set time, but for comprehensive results they should be used in conjunction with conventional trapping and detection methods (Friend 1984). Setting funnel traps alongside the fence may also enhance the number of larger reptiles captured (Enge 2001).

In addition, species that have a close association with a certain microhabitat are less likely to be captured by this method because they rarely enter the area where the drift fences are (Corn 1994). For example, if all the drift fences in a study are located on arid ridgelines, then those salamanders that typically travel only a certain distance from a water source are unlikely to be captured. To remedy this, the project's goals should be carefully considered during the design stage.

Mortality of animals captured in pitfall and funnel traps can be a problem, particularly in dry or exposed habitats. To avoid high mortality rates in dry habitats, wet sponges and/or some form of shade should be placed in traps to provide some moisture to captured animals. Mortality rates of reptiles were found to be similar in pitfall and funnel traps, but in drier habitats, anurans were more prone to dying in funnel traps, despite the presence of shade covers and sponges (Enge 2001).

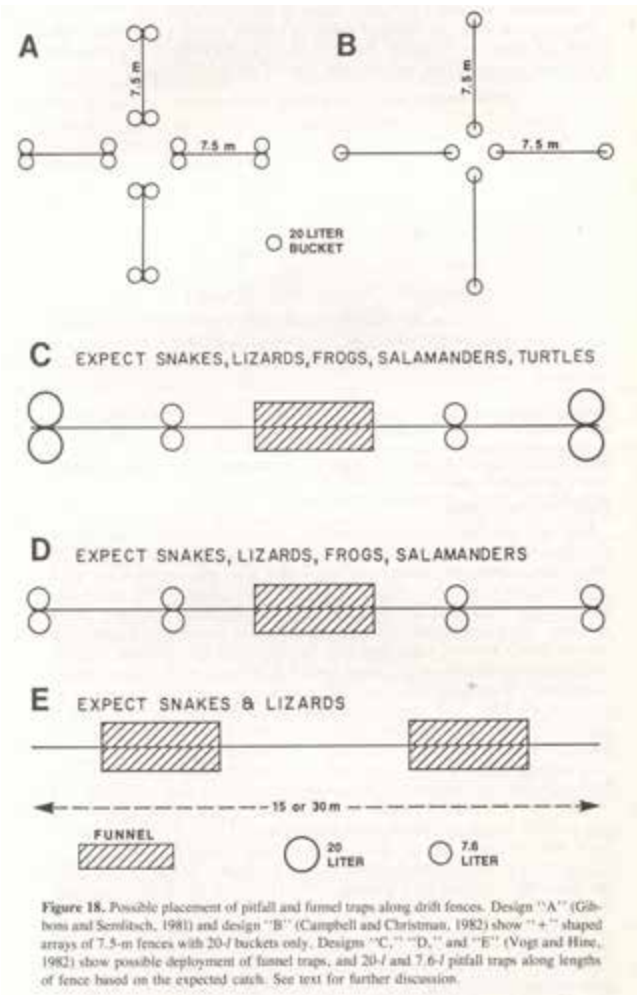


Figure 18. Possible placement of pitfall and funnel traps along drift fences. Design "A" (Gibbons and Semlitsch, 1981) and design "B" (Campbell and Christman, 1982) show "+" shaped arrays of 7.5-m fences with 20-*l* buckets only. Designs "C," "D," and "E" (Vogt and Hine, 1982) show possible deployment of funnel traps, and 20-*l* and 7.6-*l* pitfall traps along lengths of fence based on the expected catch. See text for further discussion.

Possible placement of pitfall and funnel traps along a drift fence. Reprinted from *Vogt, R.C. and R.L. Hine. 1982. Evaluation of techniques for assessment of amphibian and reptile populations in Wisconsin. Pp. 201-217 In N.J. Scott (Ed.). Herpetological Communities. U.S. Department of the Interior, U.S. Fish and Wildlife Service, Wildlife Research Report 13. Washington D.C.*

Inventory

Although generally used for monitoring because of the time and funding investment, drift fences can also be used in conjunction with species inventories. In this situation, traps would be opened only during certain seasons (e.g., breeding seasons) or environmental conditions (e.g., warm rainy nights). To maximize the number of species detected in an inventory, supplement your sampling program with other methods, such as coverboards, visual encounter surveys, PVC pipes, and auditory surveys (see the sections for each of these techniques).

Monitoring

This technique is ideal for long-term population studies and assemblage monitoring, particularly when placed around a breeding site (Dodd and Scott 1994). For example, Greenberg and Tanner (2005) used nine years of drift fence data at eight ponds in Florida's longleaf pine-wiregrass sandhills to study spatial and temporal patterns of breeding and recruitment in eastern spadefoot toads (*Scaphiopus holbrookii*). There are many different applications for using drift fences and pitfall traps to monitor amphibian and reptile populations. For example, herpetofaunal response to clearcutting was monitored at wetlands in South Carolina by assessing the changes in overall richness, abundance, and community similarity over a 3-year period (Russell et al. 2002). However, if species richness is an objective of the study, other techniques should probably also be employed.

Gabrielle Graeter

Drift fence materials: rolls of aluminum flashing and staking poles.

Conclusions and recommendations

Other methods may be a more practical use of your time and funding, but this technique can be incredibly useful for detecting the presence of rare species and monitoring changes in a population over time, and if used along with other techniques it can also be valuable in determining species richness at a site (Corn 1994). Because of the large time and funding investment this method can require, one must be realistic in what can be accomplished with available funding and personnel, both in building the fences and in your ability to monitor the drift fence on a daily basis. The design stage is especially critical for this technique because the placement of fences (in habitat, type, and orientation), the number installed (e.g., of arrays, buckets), and the trapping schedule (e.g., frequency per week and per day, continuous or seasonal) will affect the captures and the statistical analyses possible.

Equipment Checklist

Installation:

- Map of area and design plan
- Compass and/or GPS unit to mark fence location
- Flagging/stake flags for marking drift fence locations
- Equipment for digging and preparing trenches (e.g., post-hole digger, motorized trencher, shovels, pruning shears, garden hoes
- Fence material (e.g., aluminum flashing, plastic silt-fencing, chicken wire, hardware cloth)
- Posts (wooden or metal)
- Rubber mallet for securing posts around the fencing
- Hole puncher (i.e., awl; for puncturing aluminum flashing to insert cable ties)
- Cable ties for securing the posts to the fence
- Glue gun and aluminum tape (for securing ends of flashing fence)
- 20-L buckets and/or large tin cans (coffee cans are commonly used)
- Drill (to put holes in buckets so water can drain)
- Work gloves (aluminum flashing is sharp)
- Sponges and shading materials for pitfall traps
- Plastic collar (optional; to put at top of pitfall traps)

Sampling:

- Data recording materials
- Measuring and marking materials (e.g., scissors, PIT tags, metric ruler, scales)
- Containers and cloth bags (for holding animals)
- Snake stick and/or tongs
- Large bucket with lid (if venomous snakes need to be returned to the lab)
- Small nets (for scooping out creatures in pitfall traps with some water in them)
- Containers or pump for bailing water out of pitfall traps
- Equipment for recording environmental data (e.g., thermometer, raingauge)

* See Appendix IX for basic equipment lists

SAMPLING AT SNAKE HIBERNACULA

Cameron A. Young, Robert N. Reed, and Robert T. Zappalorti

Many reptiles and amphibians in temperate zones or at high elevations avoid the thermal stress of winter by hibernating below the frost-line in underground hibernacula. Communal denning in hibernacula is an important part of the seasonal cycle of many reptiles, especially snakes (Klauber 1972), but also lizards (Congdon et al. 1979) and turtles (Carpenter 1957). Reptiles may hibernate singly or may aggregate in monospecific or heterospecific groups (Parker and Brown 1973; Gregory 1974).

A hibernaculum can be described as any secure retreat (i.e., protected from sub-freezing temperature) where herpetofauna can spend the winter. Natural retreats may include rock piles (Vetas 1950; Parker and Brown 1973), sinkholes (Gregory 1974; Shine et al. 2001a), hollow trees (Kauffeld 1957), the holes left by rotting tree stumps (Viitanen 1967), caves (Drda 1968; Sexton and Hunt 1980), abandoned rodent burrows (Cohen 1948; Carpenter 1953; Viitanen 1967; Plummer 2002), crevices in shale (Bothner 1963), ant mounds (Criddle 1937; Carpenter 1953), prairie dog burrows (Klauber 1972; Holycross 1995), fox burrows (Zappalorti et al. 1983), skunk burrows (Zappalorti et al. 1983), gopher tortoise burrows (Moler 1992), crayfish burrows (Carpenter 1953; Kingsbury and Coppola 2000), and limestone crevices (Shine et al. 2000). Some of these retreats are also used by herpetofauna as refuge during the remainder of the year. Reptiles and amphibians may alternatively utilize human-made structures as hibernacula, including railroad beds (Zappalorti and Reinert 1994), old wells (Brown et al. 1974), abandoned dump sites, and buried debris from land clearing operations (Zappalorti and Reinert 1994). Artificial dens can also be purposefully constructed in suitable areas as a conservation measure (Zappalorti and Reinert 1994).

Hibernacula are potentially limiting resources for many snake species, especially in extreme habitats (Parker and Brown 1973; Burger et al. 1988). Many species have experienced declines because of the loss of hibernacula by human development or their persecution at den sites (Klauber 1972; Parker and Brown 1973; Galligan and Dunson 1979; Gregory 1984; Zappalorti and Reinert 1994). Locating and monitoring populations at hibernacula can provide information about many aspects of snake natural history and should play an important role in their conservation.

Utility

Studies conducted at hibernacula can provide data on population size and demography, activity cycles, thermal characteristics of hibernating animals, and local community richness and evenness (Blouin-Demers et al 2002; Brown and Parker 1982; Graves and Duvall 1987, 1990). Observing or capturing entire populations at communal hibernacula allows the researcher to rigorously investigate topics such as temperature relationships and tolerance (Brown et al. 1974; Jacob and Painter 1980; Sexton and Hunt 1980; Weatherhead 1989), winter mortality (Hirth 1966; Shine et al. 2001a,b), species comparisons (Parker and Brown 1973), courtship and mating behaviors (Shine et al. 2000), and movement patterns into and away from hibernacula (Reinert and Zappalorti 1988; Shine et al. 2001a,b).

Pedestrian surveys and radiotelemetry are often used to discover unknown hibernacula and retreat locations (Brown and Parker 1976; Reinert 1992; Zappalorti and Reinert 1994; Prior and Weatherhead 1996; Kingsbury and Coppola 2000). Pedestrian surveys (also known as visual encounter surveys) are conducted by looking for snakes while walking slowly through potential denning habitats (in the north temperate zone these are typically south-facing rocky hillsides). Radiotelemetry can be one of the most productive ways of locating den sites (Reinert and Zappalorti 1988), as snakes implanted with radiotransmitters during summer months can be followed to hibernacula in the fall. However, radiotelemetry is very expensive in initial equipment costs and personnel time (See Radiotelemetry section). New methods such as remote sensing and geographic information system technologies (see Chapter 4) can be used to locate potential denning sites, followed by surveys at these sites to pinpoint actual hibernacula.

Limitations

Several factors must be considered when planning surveys and monitoring programs for snake dens. Observers may have different search images, experience, or knowledge of the target species and this variation must be considered when designing and implementing a den or hibernaculum study (e.g., Rodda 1993). More importantly, only a small subset of North American snakes are known to hibernate communally in appreciable numbers. Most of these communally hibernating populations occur where suitable hibernacula are a limited resource; for example, prairie rattlesnakes (*Crotalus viridis*) hibernate in geographically dispersed rock outcrops that provide access to suitable below-ground winter temperatures (Graves and Duvall 1990). In areas with abundant thermally-suitable hibernacula, populations of these same species typically hibernate individually (Reed and Douglas 2002). Thus, while inventory and monitoring at hibernacula can be an extremely efficient means of detecting most or all individuals in a population, its utility is restricted to a handful of species. Finally, presence of traps or researchers at a hibernaculum may alert others to the location of the site, possibly resulting in poaching or willful destruction of the population. For some species or populations, therefore, it may be prudent to locate and study only those populations using hibernacula far from casual observers.

Inventory

Sampling snakes at communal hibernacula can yield information on entire populations or demes, but this requires a fairly large commitment in person-hours devoted to visual searching and/or resources and labor required for trap construction, placement, and checking. Fortunately, such intensive activities are only required during brief periods of time in spring and fall when snakes are concentrated around the dens. Entry and emergence from dens are seasonally predictable, but vary with elevation, latitude, species, and annual weather variation. Further, use of hibernacula may vary by sex and age of snakes; for example, gravid female rattlesnakes may emerge from hibernation after males, and may stay in the immediate vicinity of a den until parturition. Researchers may need several years of study to pinpoint optimal times and places to capture a representative sample of the population. If the goal of inventory is merely to document presence of a species of interest, then visual surveys in suitable habitat in spring or fall will likely suffice. If, however, an inventory is designed to include estimates of population size or ecology, trapping or intensive visual surveys will be required, often including mark-recapture analysis.

Monitoring

Monitoring populations at hibernacula will necessarily be a multi-year effort, as researchers typically have only two short periods annually during which snakes can be captured. Over time, a database of captures of known individuals can be assembled, allowing rigorous estimation of population size and other population parameters, as well as how these parameters change over time. Employing modern methods for analysis of mark-recapture data (e.g., program MARK; White and Burnham 1999) provides more robust conclusions than traditional index-based approaches (Amstrup et al. 2005; Williams et al. 2002).

One especially exciting avenue for monitoring populations at hibernacula involves employing technology for automated identification of individuals. Once a population has been captured and marked using passive integrated transponders (PIT tags; see Appendix III – Marking Amphibians and Reptiles) in the spring or fall (using any of the methods below), a drift fence with a single ingress/egress point can be erected around the hibernaculum. This gap in the fence can be fitted with a PIT tag scanner connected to a data recording device and a large battery or solar charger, enabling all marked snakes to be detected when they enter or leave the hibernaculum (A.T. Holycross, pers. comm.; also see Automated Data Acquisition in Chapter 4).

TRAPPING METHODS

Burrow traps

Many retreat sites can be trapped directly by setting a funnel or box trap in the entrance of the refuge, preferably so that the trap covers the entire opening. In this way, animals leaving the refuge site will be captured in the trap. This trapping method is most effective with medium sized holes or burrows, rather than rock piles or other types of refuges that lack a defined opening. These types of traps need to be sufficiently shaded and checked daily. A drawback of this method is that it only works when the species of interest are in the refuge and attempt to exit, not when they are entering the burrow or hole.

As an example of burrow trapping, in many regions of the Southeast, burrows of the gopher tortoise (*Gopherus polyphemus*) provide refuge for numerous amphibian and reptile species, several of which are rare or threatened (e.g., gopher frogs *Rana capito*, indigo snakes *Drymarchon corais*, and eastern diamondback rattlesnakes *Crotalus adamanteus*). Single-ended funnel traps made of flexible wire mesh have been used effectively to trap in this unique habitat (Fig. 5-17). These traps are set by covering a burrow entrance with the open funnel end of the trap and packing soil to seal the edges around the trap. Traps are best left on a burrow for several days and, as with other traps, must be shaded and checked at least twice daily to avoid inadvertently killing trapped animals.

Kurt Buhlmann

Figure 5-17. Single-ended funnel trap for trapping snakes at gopher tortoise burrows.

Confinement traps

Confinement traps can be used with known or suspected small retreats and hibernacula such as rodent burrows, crayfish burrows, or ant mounds (Carpenter 1953). These traps are used to confine snakes exiting hibernacula in a small arena so they can be differentiated from snakes entering the hibernacula. A series of fences and one-way funnels are constructed in the following manner: (1) The entire entrance hole or hole complex is surrounded by a cone of small-mesh hardware cloth or equivalent. The cone is placed over the entrance hole and buried at least 10 cm into the ground, and functions to prevent escape of snakes capable of climbing; (2) The trap is then enclosed with an exclusion fence approximately 1 m from the cone, with several small funnels at ground level to allow entry to the area between the exclusion fence and the confinement cone. Snakes returning to the hibernaculum will circle the exclusion fence and enter the interior through the funnels. This trap is effective for determining use of a known or suspected small hibernaculum but is not appropriate for large denning areas.

Drift fences

Known den sites can be surrounded with drift fences (see Terrestrial drift fences section) in the late winter to facilitate capture of snakes emerging from hibernation later in spring (Brown and Parker 1982) or in fall and spring to intercept movement to and from the den (Brown and Parker 1976). Care should be exercised that repeated visits do not alter behavior, drive away some species, or damage the den site (Brown 1993). Large drift fences encircling an entire denning complex are initially labor intensive, but once established can facilitate the collection of long-term data that provide information on the denning population. One assumption with drift fence and confinement cone studies is that no animal escapes detection or trespasses through the fence, so proper installation of the fence is necessary. A mark-recapture protocol and proper fence installation can ensure that this assumption is reasonable, but cannot completely validate it.

Several different traps can be used along drift fences to capture snakes, including funnel traps, box traps, and coverboards (see Funnel Trapping for more information on traps). Traps can be placed on both sides of the fence to determine ingress and egress of snakes. If irregular monitoring of the drift fence occurs, coverboards should be used instead of funnel or box traps. If a single discrete entrance to the den has been identified, it may be possible to place a large hose or pipe (rubber, PVC, or plastic) over the entrance. The hose or pipe will direct emerging snakes into a corral

or large trap with a one-way door (Klauber 1972). As with other confinement traps these should be checked daily and before any extreme temperature fluctuations. More detail on drift fences, pitfall traps, funnel and box traps, and coverboards can be found in those respective techniques sections of Chapter 5.

Conclusions and recommendations

Sampling snakes at hibernacula can be an efficient means of sampling entire populations, and only demands intensive field efforts during the spring and/or fall when snakes are transiting to and from hibernacula. Initial efforts to characterize a population can be expensive in terms of labor and supplies, but efforts usually decrease over time once drift fences and traps are in place and researchers learn the seasonal activity patterns of the particular population. The time and expense associated with capturing all or most snakes at a hibernaculum, however, is certainly less than what would be required to capture these individuals once they have dispersed widely to their activity ranges. Care must be taken to avoid alerting the public to the location of hibernacula so as to avoid persecution of the snakes.

Equipment Checklist

Installation:

Map of area and design plan
Flagging/stake flags for marking trap locations

Trap building materials (many can also be purchased)

See the sections on terrestrial drift fences and funnel trapping for more details

General:

- Shears for wire mesh
- Baling wire, hog rings or cable ties for connections and repairs
- Shovel, pickaxe, or mechanical ditch-digger for fence erection

Burrow traps:

- Single-ended flexible wire mesh funnel traps
- Plastic, wood, or other shade for traps if natural shade is lacking

Confinement traps and box traps:

- Frame constructed of wood, metal, or PVC, overlaid with wire mesh hardware cloth
- Single or multiple funnel entrances, depending on drift fence type and hibernaculum location
- Shade for trap as above, or refugium placed inside trap

Drift fences:

- Aluminum flashing (for semi-permanent fences)
- Plastic sheeting or erosion-control silt fence (for short-term fences)

Sampling:

Data recording materials
Measuring and marking materials (e.g., scissors, PIT tags, metric ruler)
Containers and cloth bags
Cable ties or baling wire (for repairing traps as needed)
Snake tongs, hooks, and/or tubes for handling venomous species

* See Appendix IX for basic equipment lists

TRACKING TECHNIQUES APPLICABLE TO MONITORING

Gabrielle J. Graeter

Although thread-tracking (i.e., spool-and-line tracking, spool-tracking), dye-tracking (i.e., fluorescent powder tracking, fluorescent pigments), and radio-tracking (i.e., radio-telemetry) are generally used for research studies, these techniques can also be useful for monitoring purposes, albeit in a supplementary role. Thread-tracking involves attaching a small spool of thread to the animal and allowing it to unravel as the animal moves, thereby leaving a thread trail. For turtles, the spool is affixed to the shell and the loose end of the string is secured to an object in the release location. Thread-tracking has been used with anurans by attaching the spool on their back with one of the various types of belts or backpacks designed for this purpose. Attaching a spool may not be feasible for many species, including snakes, because of the difficulty of securing it to the animal. With dye-tracking, non-toxic fluorescent powder is applied to an animal, and the powder trail created during movement is later tracked with the use of a portable UV lamp. Radio-tracking entails attachment or implantation of a radiotransmitter on the target species. Transmitters are now being manufactured that are small, relatively lightweight, and designed specifically for use with amphibians and reptiles. A receiver and antenna is needed to locate and follow the movements of individuals. When a transmitter is attached externally, a belt or backpack is necessary to secure it in place.

Gabrielle Graeter

Transmitters that weigh less than 1 g are available for use with medium sized amphibians, such as this southern toad (*Bufo terrestris*). The transmitter is attached with a belt around the toad's waist.

These tracking techniques can provide information that is necessary to meeting the goals of a program and cannot be obtained through basic population monitoring techniques. Tracking the movements of

individuals can yield data on habitat preferences, activity patterns and levels, and could prove helpful for acquiring information on interactions and diet. For example, detailed information about a species' habitat use in a particular area over time may be best gathered using one of these tracking techniques.

Utility

These techniques should be employed primarily when information is needed that cannot be acquired through simpler, less time-intensive methods. They can be very useful for determining habitat use and movement and activity patterns among areas or over time. The following references are useful sources for information about using thread-tracking (Heyer 1994; Lemckert and Brassil 2000; Buhlmann and Gibbons 2001; Tozetti and Toledo 2005), dye-tracking (Mullican 1988; Fellers and Drost 1989; Blankenship et al. 1990; Dodd 1992; Eggert et al. 1999; Stark and Fox 2000; Eggert 2002; Graeter and Rothermel 2007), and radio-tracking (Mullican 1988; White and Garrott 1990; Richards et al. 1994; Carter et al. 1999; Eggert et al. 1999; Lamoureux and Madison 1999; Lemckert and Brassil 2000; Eggert 2002; Johnston and Frid 2002) techniques for amphibians and reptiles.

Kurt Buhlmann

Thread trailing also has many advantages as a tracking technique.

Kurt Buhlmann

Fluorescent powder can be useful for tracking movements of some herpetofauna, including hatchling chicken turtles (*Deirochelys reticularia*).

Gabrielle Graeter

A southern toad, *Bufo terrestris*, was tracked to this location with green fluorescent powder.

Thread-tracking and dye-tracking are the most economical methods of the three and, for this reason, are generally preferred over radio-tracking as long as these simpler methods meet program needs. Determining whether to use dye-tracking or spool-tracking will depend on several factors, including the feasibility of using each technique on the species of interest (e.g., some amphibians may be too small to carry the spool without influencing their movement), the efficacy of each technique in the study habitat (e.g., if working in an area that receives high rainfall and has nearly constant high substrate moisture, then fluorescent powder will not work well and spool-tracking should be used), and the type of information needed [e.g., thread-tracking will yield exact distances traveled but dye-tracking is better for determining activity in a habitat (e.g., more powder indicates where an individual toad burrowed into the substrate)], among other considerations (e.g., limitations, data possible from each technique). For example, fluorescent powder may be more useful and more easily implemented for determining the movements of juvenile turtles as they disperse from their natal pond, whereas thread-tracking may be the preferred method when data are needed on individual movements and habitat use of endangered frogs (that have small home ranges) in several different habitat types (i.e., it will be difficult to distinguish between the paths when they overlap in the small areas).

Compared to dye-tracking and thread-tracking, radio-tracking yields longer-term data, which may be necessary for some monitoring studies. Thus, radio-tracking may be most useful when data are needed over a relatively long period of time about habitat use or the behavior and activity of a species. For example, if a researcher needs to know habitat use of a turtle or frog species over several seasons, radio-telemetry may be the best way to obtain this information. Radio transmitters are available from a number of vendors. Battery life of the transmitter is affected by battery size

and pulse frequency. Radio transmitter size, shape, and weight are important factors to consider before ordering and attaching the device. In many frogs, external attachment is an option, whereas insertion via surgery is often necessary for salamanders and snakes. Because the sizes and shapes of transmitters and attachment methods vary widely and surgical insertion requires hands-on experience, we strongly recommend consulting an expert herpetologist with radio-tracking experience. If possible, seek advice and guidance from someone who has worked with the same or a similar species.

In turtles, attaching radios has often been achieved with epoxy (two-part malleable products are preferred to liquid epoxies), but some researchers have used stainless steel bolts or wires. Some researchers prefer to allow radio transmitter antennas to trail behind the turtle, while others have curled and glued the antenna around the carapace marginals. The latter technique reduces signal attenuation (i.e., distance that the signal can be received). Attachment of the radio needs to be carefully considered as entanglement and drowning can occur, as well as interference with reproduction (i.e., mating).

Kurt Buhlmann

Radiotransmitter attached to shell of an adult chicken turtle, *Deirochelys reticularia*.

Gabrielle Graeter

Radiotracking equipment varies, but the Yagi antenna is the most common type of antenna used.

Limitations

These tracking techniques may require additional funds, planning, resources, and time to implement. For example, the equipment necessary for radio-tracking (e.g., transmitters, receiver, antenna) can be quite costly, and tracking individuals late into the night or through thick vegetation may require a large amount of time and enthusiasm. Although thread-tracking and dye-tracking can be implemented with relative ease, these techniques may not provide data from a long enough period of time to answer the program questions.

Another limitation of these techniques is that they may influence the behavior of the individual being tracked. Both thread-tracking and radio-telemetry involve adding an object and extra mass to the animal (Fig. 5-18), whereas the weight of fluorescent powder is negligible and without detrimental physiological effects (see Rittenhouse et al. 2006). For more information about the utility and limitations of each of these techniques, consult the references cited in this section.

Kurt Buhlmann

Figure 5-18. Consider the potential effects of tracking on the animal and use a method that addresses the study objectives with minimal effects on the study animals.

Conclusions and recommendations

Thread-tracking, dye-tracking, and radio-tracking can be useful for gathering additional information for a monitoring study, but should only be used when data cannot be obtained through simpler means. When determining whether or not to use any of these techniques, one should consult references that describe these techniques in greater detail (see those cited above). Likewise, resource managers who determine that thread-tracking, dye-tracking, or radio-tracking will help them gather necessary monitoring data should confer with a herpetologist who has experience with that technique. A careful consideration of the costs (e.g., time, funds, personnel, and resources), limitations, and the type of data obtained from a particular technique is an essential part of this decision process. These methods have important applications for monitoring projects, but complete instruction and use is beyond the scope of this section.

Equipment Checklist

Installation:

- Map of area and design plan
- Flagging/stake flags for marking study area

General materials:

- Data recording materials
- Measuring and marking materials (e.g., scissors, PIT tags, metric ruler, scales)
- Containers
- GPS unit or measuring tape reel and compass (depending on program objectives)

Thread-tracking:

- Spool and thread of appropriate size
- Spool attachment material (e.g., duct tape for turtles, waist belt for frogs)

Dye-tracking:

- Fluorescent powdered pigments in baggies or small containers
- UV-light
- Wet washcloth in plastic bag (wipe hands on cloth to reduce powder spillage)
- Stake flags or strings to mark powder trails

Radio-tracking:

- Radio-transmitters
- External attachment materials (e.g., Teflon tubing, beaded belt) or materials for internal implantation
- Receiver
- Antenna (Yagi or H)
- Stake flags to mark animal location

* See Appendix IX for basic equipment lists

SAMPLING TECHNIQUES FOR EXOTIC AMPHIBIANS AND REPTILES

Walter E. Meshaka, Jr.

How does one sample amphibians and reptiles? Generally speaking, sampling for amphibians and reptiles is not so difficult a task. Not so easy, however, is choosing the sampling method best suited to answer the question being asked. For example, what method should be used to determine the proportions and seasonal activity of several non-native species (Cuban treefrog, green treefrog, squirrel treefrog, Indo-Pacific gecko, tropical house gecko) on buildings and along trails in the Southern Florida Everglades? These questions were answered by conducting nocturnal counts throughout the year (Meshaka 2000, 2001).To do this, nights that were more than half full moon were avoided and around buildings and trails were searched by flashlight twice each month. Measuring the perimeters of the buildings and recording time spent searching, numbers of lights, and numbers of refuges all provided layers of information to better explain the patterns observed.

Since this study in the Everglades, the “Parknership” program of the Florida State Park Service has adopted this procedure for precisely the reasons that PARC is interested in it - to provide the kinds of useful information necessary to make good management decisions regarding exotic amphibians and reptiles on protected (in their case public) lands. Because the Florida State Park Service was interested in a quick snapshot of the relative abundances of their building inhabitants and because the studies were conducted in southern Florida parks, weekly counts during approximately two and one half month periods in the winter were adequate to answer their questions (Meshaka et al. 2005b; Meshaka et al. 2006b).

As one proceeds northward, these generally tropical species become evermore seasonal in their activity and methods must be adjusted accordingly. Therefore, to sample such exotic amphibians and reptiles as the Cuban treefrog and geckos, weekly nocturnal counts during June-August as a minimum can provide a useful assessment of presence/absence (i.e., detection/non-detection) and a measure of relative abundance during summer. For example, nocturnal counts were conducted during May-October to assess the status of the Mediterranean gecko in the Southeast (Nelson and Carey 1993). Standardized nocturnal counts are equally useful for northern populations of Mediterranean geckos and Hawaiian populations of nocturnal geckos, such as mourning geckos.

In other cases, diurnal counts may be most useful. Diurnal counts were used successfully in a four-year ecological study of the knight anole in southern Florida (Meshaka and Rice 2005). In that study, anoles were marked individually, but all sightings (sum of all marked lizards + sum of all unmarked lizards) were used for measurement of daily and seasonal activity. Counts also were employed for several species of anoles found together at various sites in southern Florida, with attention paid to perch heights (e.g., Meshaka 1999a,b). The relative abundances of the various species co-occurring at these sites can secondarily be quantified so as to understand the assemblage structure of the anole species at each site, thereby finding out what species of anoles dominate and in what proportions to one another. This technique, like its nocturnal counterpart, if related to unit time (e.g., number of animals per minute of searching a field, a transect, a trail, or a building), area (e.g., number of animals per building of differing perimeters), or distance (e.g., number of animals per meter of searching along a transect or trail), can be very effective for sampling diurnal exotic lizards, such as green iguanas (Fig. 5-19).

The diurnal exotic lizards tend to be heat-loving creatures and some of them, like the giant ameiva, have distinct daily activity patterns. Therefore, counts should include summer months and morning and afternoon times. Populations can be counted frequently, such as daily or a few times each week. This technique is well-suited to diurnal species elsewhere, such as the north temperate populations of the wall lizards and to day geckos in Hawaii. However, censuses can be conducted less frequently for shy populations or species. For some species, such as the spectacled caiman, both nocturnal and diurnal census techniques can be employed. The length and number of transects covered are relative to the size of the study area and can be constrained by the amount of time available for a given visit. The search distance to the left or right of the transect depends on the ease of detectability of the study animal. For example, the larger the body size and more boldly patterned the species, the farther one can effectively spot the animal, the distance of which can be enhanced by the use of binoculars for such animals as green iguanas and spectacled caimans. On the other hand, small and cryptically patterned species, such the tropical house gecko would be difficult to detect in vegetation more than approximately 2 m away.

PVC pipes provide an effective method for detecting and counting Cuban treefrogs, the diameters of which influence capture success (Bartareau 2004; also see the techniques section on PVC pipe surveys). The advantages of this technique are that it is effective and this method does not demand daily visits. The disadvantage of this method is the potential for habitat enhancement. Population sizes of the Cuban treefrog are limited by the number of refuges and wild Cuban treefrogs are generally dead within two years of life (Meshaka 2001). Consequently, whereas six months to one year of sampling can provide a useful measure of Cuban treefrog abundance at a site, longer durations of PVC pipe use can inadvertently and quite effectively provide room for more Cuban treefrogs. Consequently, use PVC pipes only for time intervals that are shorter than the generation times of the Cuban treefrog, with future re-sampling projects being conducted after a wait of at least two years.

Drift fence arrays, with funnel traps and buckets, and pitfall traps in a grid can be used to census the greenhouse frog (Meshaka and Layne 2005) and are also suitable for ground-dwelling lizards and small individuals of the Indian python. Because the greenhouse frog can climb, buckets are more effective if a collar or rim is created by cutting a large hole in the lid of the bucket. Funnel traps can even be modified for special use. For example, the greenhouse frog was detected as an inhabitant of gopher tortoise (*Gopherus polyphemus*) burrows by using one-sided traps at the mouth of the burrow to intercept individuals (Lips 1991).

Russ Mapp

Figure 5-19. Observing the behavior of Green Iguanas at Hugh Taylor Birch State Park in southern Florida.

Some species, such as the Brahminy blind snake, spend too much time underground, or occasionally in palm boots, to be sampled by the techniques mentioned above. For this species, active search through leaf-litter, under artificial damp cover such as carpet, and in palm boots close to the ground is most effective, especially when related to time or area searched (see descriptions of these techniques in Chapter 5). To extract more kinds of information on the species, animals can be marked individually, as was done in studies such as those on the knight anole (Meshaka and Rice 2005) and Mediterranean gecko (Punzo 2001b).

Consistency in any protocol is important. Increasing the duration of study, the number of sites sampled, and the numbers of techniques employed increases the likelihood of detection. It also provides useful data relating to habitat use and daily and seasonal activity patterns, all of which are necessary for understanding how successful the species is with respect to the number of colonies and individuals, why these species succeed or fail, and how to most effectively manage for them.

SEEK ADVICE AND FURTHER TRAINING

Gabrielle J. Graeter

This chapter provides an overview of the many sampling techniques available for use in inventory and monitoring programs, but the learning process should not end here. In order to have a successful project, it would be wise to have a complete understanding of the study species, study area, and the logistics, equipment, and design involved in using a particular technique. Reading the applicable sections in regional guidebooks can be helpful, as can pertinent literature about the species or techniques. Likewise, some studies may benefit from a visit to a local museum to view their reptile and amphibian specimens or records. One of the best ways to learn more is to consult a herpetologist who is an expert on your study species or has a lot of experience with specific techniques you are planning to use. They may offer pointers about a technique or about project design that are not found in a book or publication and that ultimately increase the likelihood of meeting the program objectives. They may also know of unpublished reports that contain helpful information on the topic. Another way to become better educated is to participate in a training workshop (e.g., PARC, Partners in Amphibian and Reptile Conservation, has offered training workshops). The hands-on aspects of these types of training courses can be very informative, as can the opportunity to speak with professionals about project ideas.

Jeff Beane

Museum collections

John White

Regional guidebooks can be very helpful in getting started

The importance of consulting with others and thoroughly educating yourself on the species and topic of interest cannot be overstressed. Below are several examples that demonstrate why doing so is critical to program efficiency and success.

Example 1: Location Considerations: Your sampling plan may look good on paper and seem solid according to the guidebooks and literature you have read, but a local expert on your study species may have information that is critical to the success of your project. For example, a range map in a guidebook may indicate that a species is present in a particular area, but a local expert may have more detailed knowledge about the species' range. An expert familiar with the region may know that one of the areas where you plan to sample has very few individuals of that species. Thus, the species may not be present in large enough numbers (if at all) to capture with your proposed method; they may suggest the use of alternative sampling techniques or areas. By consulting experts, you will have used your resources (time, money, etc.) more wisely and increased your chances of accomplishing the project goals.

Example 2: Techniques: Executing a technique, such as drift fences with pitfall traps, may sound simple when reading about it in a book, but actually implementing it may prove much more difficult. Attending a herpetological training course that includes instruction and hands-on practice with particular techniques, as well as time to ask questions about these techniques, can be an invaluable experience. Getting out into the field and physically checking traps and walking around an area with drift fence arrays and pitfalls will give you a more realistic idea of the resources, time, challenges, and personnel required. Likewise, consulting with researchers who have experience with the technique can be invaluable. Thus, this type of

workshop training experience is likely to improve the outcome of your study.

In summary, during the design phase of an inventory or monitoring program, one should consult with others that are doing similar work and with local experts to get additional advice or insight into the plans you have proposed. Visiting your local museum to examine records may also prove helpful, and attending hands-on training workshops about inventory and monitoring techniques can be a very useful experience. Taking the time to become educated thoroughly about all the details of a project will likely save valuable time, resources, and money, as well as help in meeting project goals.

Inventory and monitoring workshop participants getting hands-on experience with tin coverboards.

Inventory and monitoring workshop participants learning, in a hands-on manner, about the design and uses of aquatic funnel traps.

Prior to initiating a new study:

- Use the information in this book to guide you in formulating objectives, figuring out study design, and determining techniques
- Consult regional guidebooks, pertinent literature, and museum records as appropriate
- Contact herpetological experts on your study species and techniques
- Attend training workshops as appropriate

A PARC Inventory and Monitoring Workshop group, learning the techniques in the field.

VISION FOR THE SPECIES X TECHNIQUES TABLE

Kurt A. Buhlmann and Gabrielle J. Graeter

In Chapter 5 of the PARC Inventory and Monitoring book we present a large matrix that lists all amphibian and reptile species found in the U.S. and Canada. The intent of the matrix is to provide guidance about species-specific inventory and monitoring techniques. Suggested techniques are provided for two levels of inventory: 1) Short-term surveys (e.g., a two-week consulting job prior to development) and 2) Comprehensive surveys (e.g., a multi-year survey of a state park or national forest). Suggested techniques are also provided for two levels of monitoring: 1) Presence/absence monitoring (i.e., is the species still present on a property and detectible at regular intervals) and 2) Population status/demography (i.e., is the population increasing or decreasing; is reproduction and recruitment occurring). Each suggested technique is identified with a letter code and superscript. Descriptions of each code are found in Codes and Additional Information at the end of Chapter 5. If the Comment Code column contains a number, then a corresponding text elaboration is also available in Codes and Additional Information. Many individuals contributed to the knowledge compiled in the Species X Techniques Table and they are acknowledged separately at the end of this publication.

Inventory and monitoring techniques (terrestrial and aquatic) are presented for different life stages of all amphibian species. These include Adult, Breeding Adult, Eggs, and Larva. Techniques for reptile life stages are consolidated and generally the same for adults and juveniles, but distinctions have been made when surveying for nests or hibernacula. The most appropriate season(s) to perform a certain technique are also listed and may differ among regions for the same species.

Species are presented by group (Frogs/Toads, Salamanders, Snakes, Turtles, Lizards, Crocodilians) and by PARC region (SE, NE, SW, NW, and MW) and are listed alphabetically by Genus. The taxonomy of each species list is designed to allow direct comparisons with the Species x Habitat Tables found in each of PARC's Habitat Management Guidelines (HMG) series publications. A Taxonomy Synonyms Table is found at the end of Chapter 5 and allows the reader to easily identify species of interest whose scientific names have undergone revisions since PARC began production of the HMG series and I&M book.

DEFINITIONS of the 2 Inventory and 2 Monitoring categories	
INVENTORY: RAPID ASSESSMENT	A Rapid Assessment is limited in time and scope. Example: A consultant may have only two weeks to search a site that is slated for development.
INVENTORY: COMPREHENSIVE SURVEY	A Comprehensive Survey may be conducted over multiple seasons and years. Example: A National Park or National Forest wants thorough knowledge of all species present on their managed area.
MONITORING: PRESENCE/ ABSENCE	Monitoring comes after an inventory has documented the occurrence of a target species. Simple monitoring for presence may be conducted infrequently, or at low intensity.
MONITORING: POPULATION STATUS	Monitoring goals include obtaining population demography data. How big is the population, what is the sex ratio, what is the age structure of the poplulation, etc.

FROGS AND TOADS

SOUTHEAST REGION

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Acris crepitans	Northern Cricket Frog	A	SP, SU	V	D [pf]	V	V [ce]	
Acris crepitans	Northern Cricket Frog	BA	SP, SU	A [a]	A [a]	A [r]	V [ce]	
Acris crepitans	Northern Cricket Frog	T	SU, AU	DN	DN, SN	SN	DN, SN	
Acris gryllus	Southern Cricket Frog	A	SP, SU	V	D [pf]	V	V [ce]	
Acris gryllus	Southern Cricket Frog	BA	SU	A [a]	A [a]	A [r]	V [ce]	
Acris gryllus	Southern Cricket Frog	T	SU, AU	DN	DN, SN	SN	DN, SN	
Bufo americanus	American Toad	A	SU	C, V [r]	D [pf]	D [pf]	D [pf]	
Bufo americanus	American Toad	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Bufo americanus	American Toad	E	SP	V	V	V	V [ce]	
Bufo americanus	American Toad	T	SP, SU	V	DN	DN	D [pf]	
Bufo fowleri	Fowler's Toad	A	SU	C, V [r]	D [pf]	D [pf]	D [pf]	
Bufo fowleri	Fowler's Toad	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Bufo fowleri	Fowler's Toad	E	SP	V	V	V	V	
Bufo fowleri	Fowler's Toad	T	SP, SU	V	DN	DN	D [pf]	
Bufo quercicus	Oak Toad	A	SP, SU	V	D [pf]	C	C	
Bufo quercicus	Oak Toad	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Bufo quercicus	Oak Toad	E	SP	V	V	V	CT	
Bufo quercicus	Oak Toad	T	SU	DN	SN	SN	DN, SN	
Bufo terrestris	Southern Toad	A	SU	V [r]	D [pf]	D [pf]	D [pf]	
Bufo terrestris	Southern Toad	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Bufo terrestris	Southern Toad	E	SP	V	V	V	CT	
Bufo terrestris	Southern Toad	T	SP, SU	DN	SN	SN	DN, SN	
Bufo valliceps	Gulf Coast Toad	A	SU	V [r]	D [pf]	D [pf]	D [pf]	
Bufo valliceps	Gulf Coast Toad	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Bufo valliceps	Gulf Coast Toad	E	SP	V	V	V	CT	
Bufo valliceps	Gulf Coast Toad	T	SP, SU	DN	SN	SN	DN, SN	
Bufo woodhousii	Woodhouse's Toad	A	SU	V [r], C	D [pf]	D [pf]	D [pf]	
Bufo woodhousii	Woodhouse's Toad	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Bufo woodhousii	Woodhouse's Toad	E	SP	V	V	V	CT	
Bufo woodhousii	Woodhouse's Toad	T	SP, SU	DN	SN	SN	DN, SN	
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	A	SU	C	D [pf]	C	D [pf]	
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	BA	WI, SP	A [a]	A [r]	A [a]	D [pf]	
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	T	SP, SU	DN	SN	SN	DN, SN	
Gastrophryne olivacea	Great Plains Narrow-mouthed Toad	A	SU	C	D [pf]	C	D [pf]	
Gastrophryne olivacea	Great Plains Narrow-mouthed Toad	BA	WI, SP	A [a]	A [r]	A [a]	D [pf]	
Gastrophryne olivacea	Great Plains Narrow-mouthed Toad	T	SP, SU	DN	SN	SN	DN, SN	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Hyla andersonii	Pine Barrens Treefrog	A	N/A	N/A	N/A	N/A	N/A	
Hyla andersonii	Pine Barrens Treefrog	BA	SP	A [a]	A [r]	A [a]	CT	
Hyla andersonii	Pine Barrens Treefrog	T	SU	DN	DN	DN	DN	
Hyla avivoca	Bird-voiced Treefrog	A	SU	A [a]	T [pv], A [a]	T [pv]	CT	
Hyla avivoca	Bird-voiced Treefrog	BA	SP	A [a]	A [r]	A [a]	CT	
Hyla avivoca	Bird-voiced Treefrog	T	SU	DN	DN	DN	CT	
Hyla chrysoscelis	Cope's Gray Treefrog	A	SU	V [r]	A [a]	A [a]	A [a]	
Hyla chrysoscelis	Cope's Gray Treefrog	BA	SP	A [a]	A [r]	A [a]	A [a], V	
Hyla chrysoscelis	Cope's Gray Treefrog	T	SU	DN	DN	DN	DN	
Hyla cinerea	Green Treefrog	A	SU	V [r]	V [r]	T [pv], V [r]	T [pv]	
Hyla cinerea	Green Treefrog	BA	SP	A [a]	A [r]	A [a]	A [a], V	
Hyla cinerea	Green Treefrog	T	SU	DN	DN	DN	DN	
Hyla femoralis	Pine Woods Treefrog	A	SU	V [r]	A [a]	T [pv]	A [a]	
Hyla femoralis	Pine Woods Treefrog	BA	SP	A [a]	A [r]	A [a]	A [a], V	
Hyla femoralis	Pine Woods Treefrog	T	SU	DN	DN	DN	DN	
Hyla gratiosa	Barking Treefrog	A	SU	V [r]	T [pv]	A [a]	T [pv]	
Hyla gratiosa	Barking Treefrog	BA	SP	A [a]	A [r]	A [a]	A [d]	
Hyla gratiosa	Barking Treefrog	T	SU	DN	DN	DN	DN	
Hyla squirella	Squirrel Treefrog	A	SU	V [r]	A [a]	T [pv]	T [pv]	
Hyla squirella	Squirrel Treefrog	BA	SP	A [a]	A [r]	A [a]	A [a], V [ce]	
Hyla squirella	Squirrel Treefrog	T	SU	DN	DN	DN	DN	
Hyla versicolor	Gray Treefrog	A	SU	V [r]	A [a]	A [a]	A [d]	
Hyla versicolor	Gray Treefrog	BA	SP	A [a]	A [r]	A [a]	A [d]	
Hyla versicolor	Gray Treefrog	T	SU	DN	DN	DN	T [mt]	
Pseudacris brachyphona	Mountain Chorus Frog	A	N/A	N/A	N/A	N/A	N/A	
Pseudacris brachyphona	Mountain Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris brachyphona	Mountain Chorus Frog	T	SP	DN	DN	DN	T [mt]	
Pseudacris brimleyi	Brimley's Chorus Frog	A	N/A	N/A	N/A	N/A	N/A	
Pseudacris brimleyi	Brimley's Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris brimleyi	Brimley's Chorus Frog	T	SP	DN	DN	DN	DN	
Pseudacris crucifer	Spring Peeper	A	YR	V [r]	A [r]	A [r]	A [r]	
Pseudacris crucifer	Spring Peeper	BA	WI, SP	A [a]	A [r]	A [a]	D [pf]	
Pseudacris crucifer	Spring Peeper	T	SP	DN	DN	DN	T [mt]	
Pseudacris feriarum	Upland Chorus Frog	A	WI	V [r]	N/A	N/A	N/A	
Pseudacris feriarum	Upland Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris feriarum	Upland Chorus Frog	T	SP	DN	DN	DN	T [mt]	
Pseudacris illinoensis	Illinois Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris nigrita	Southern Chorus Frog	A	WI	V [r]	N/A	N/A	N/A	
Pseudacris nigrita	Southern Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris nigrita	Southern Chorus Frog	T	SP	DN	DN	DN	T [mt]	

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Pseudacris ocularis	Little Grass Frog	A	YR	N/A	N/A	N/A	N/A	
Pseudacris ocularis	Little Grass Frog	BA	SP	A [a]	A [a]	A [a]	N/A	
Pseudacris ocularis	Little Grass Frog	T	SP	N/A	N/A	N/A	N/A	
Pseudacris ornata	Ornate Chorus Frog	A	YR	N/A	N/A	N/A	N/A	
Pseudacris ornata	Ornate Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris ornata	Ornate Chorus Frog	T, M	SP	DN	DN	DN	DN, D [pf]	
Pseudacris streckeri	Strecker's Chorus Frog	A	N/A	N/A	N/A	N/A	N/A	
Pseudacris streckeri	Strecker's Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris streckeri	Strecker's Chorus Frog	T	SP	DN	DN	DN	N/A	
Pseudacris triseriata	Western Chorus Frog	A	YR	N/A	N/A	N/A	N/A	
Pseudacris triseriata	Western Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris triseriata	Western Chorus Frog	T	SP	DN	DN	DN	N/A	
Rana areolata	Crawfish Frog	A	SU	V	V [b]	V	V [ce]	1
Rana areolata	Crawfish Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Rana areolata	Crawfish Frog	E	WI	V	V	V	CT	
Rana areolata	Crawfish Frog	T	SP, SU	DN	DN, T [mt]	DN	D [pf]	
Rana blairi	Plains Leopard Frog	A	SU	V [r]	D [pf]	D [pf]	D [pf]	
Rana blairi	Plains Leopard Frog	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Rana blairi	Plains Leopard Frog	E	WI	V	V	V	CT	
Rana blairi	Plains Leopard Frog	T	SP, SU	DN	DN, T [mt]	DN	D [pf]	
Rana capito	Gopher Frog	A	SU	V [b]	N/A	N/A	N/A	1
Rana capito	Gopher Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Rana capito	Gopher Frog	E	WI	V	V	V	CT	
Rana capito	Gopher Frog	T	SP, SU	DN	DN, T [mt]	DN	D [pf]	
Rana catesbeiana	American Bullfrog	A	SP, SU, AU	V	V	V	V [ce]	
Rana catesbeiana	American Bullfrog	BA	SU	A [a]	A [r]	A [a]	A [d]	
Rana catesbeiana	American Bullfrog	T	YR	DN	DN, T [mt]	DN	DN	
Rana clamitans	Green Frog	A	SP, SU, AU	V	V	V	V [ce], D [pf]	
Rana clamitans	Green Frog	BA	SU	A [a]	A [r]	A [a]	A [d]	
Rana clamitans	Green Frog	T	YR	DN	DN, T [mt]	DN	DN	
Rana grylio	Pig Frog	A	SP, SU, AU	V, A [a]	V, A [a]	V, A [a]	V, A [d]	
Rana grylio	Pig Frog	BA	SP, SU	A [a]	A [r]	A [a]	A [d]	
Rana grylio	Pig Frog	T	YR	DN	DN, T [mt]	DN	DN, T [mt]	
Rana heckscheri	River Frog	A	SP, SU, AU	V, A [a]	V, A [a]	V, A [a]	V, A [d]	
Rana heckscheri	River Frog	BA	SP	A [a]	A [r]	A [a]	A [d]	
Rana heckscheri	River Frog	T	YR	DN, V	DN, T [mt]	DN, V	SN	98
Rana okaloosae	Florida Bog Frog	A	YR	V	V	V	V [ce]	
Rana okaloosae	Florida Bog Frog	BA	SP, SU	A [a]	A [r]	A [a]	A [a]	
Rana okaloosae	Florida Bog Frog	T	AU, WI, SP	DN	DN	DN	N/A	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Rana palustris	Pickerel Frog	A	SP, SU	V	V [r]	D [pf]	N/A	
Rana palustris	Pickerel Frog	BA	SP	A [a]	A [r]	A [a]	A [d]	
Rana palustris	Pickerel Frog	T	SU, AU, WI	DN	DN, T [mt]	DN	DN	
Rana pipiens	Northern Leopard Frog	A	SU	V	V [r]	D [pf]	N/A	
Rana pipiens	Northern Leopard Frog	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Rana pipiens	Northern Leopard Frog	E	WI	V	V	V	CT	
Rana pipiens	Northern Leopard Frog	T, M	SP, SU	DN	DN, T [mt]	DN	DN, D [pf]	
Rana sevosa	Dusky Gopher Frog	A	SU	V	V [b]	N/A	N/A	
Rana sevosa	Dusky Gopher Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Rana sevosa	Dusky Gopher Frog	E	WI	V	V	V	CT	
Rana sevosa	Dusky Gopher Frog	T	SP, SU	DN	DN, T [mt]	DN	D [pf]	
Rana sphenocephala	Southern Leopard Frog	A	SU	V	V [r]	D [pf]	N/A	
Rana sphenocephala	Southern Leopard Frog	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Rana sphenocephala	Southern Leopard Frog	E	WI	V	V	V	CT	
Rana sphenocephala	Southern Leopard Frog	T	SP, SU	DN	DN, T [mt]	DN	D [pf]	
Rana sylvatica	Wood Frog	A	SU	C	D [pf]	C	N/A	
Rana sylvatica	Wood Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Rana sylvatica	Wood Frog	E	WI	V	V	V	CT	
Rana sylvatica	Wood Frog	T	SP	DN	DN, T [mt]	DN	T [mt]	
Rana virgatipes	Carpenter Frog	A	SP, SU, AU	V	V	V	N/A	
Rana virgatipes	Carpenter Frog	BA	SP, SU	A [a]	A [r]	A [a]	A [d]	
Rana virgatipes	Carpenter Frog	T	YR	DN	DN, T [mt]	DN	DN	
Scaphiopus holbrookii	Eastern Spadefoot	A	YR	V [r]	N/A	V [r]	N/A	
Scaphiopus holbrookii	Eastern Spadefoot	BA	WI, YR	A [a]	A [r]	A [a]	D [pf]	3
Scaphiopus holbrookii	Eastern Spadefoot	E	WI, YR	V	V	V	CT	
Scaphiopus holbrookii	Eastern Spadefoot	T	WI, YR	V	V	V	T [mt]	
Scaphiopus hurterii	Hurter's Spadefoot	A	SP	V [r]	N/A	V [r]	N/A	
Scaphiopus hurterii	Hurter's Spadefoot	BA	YR	A [a]	A [r]	A [a]	A [d]	3
Scaphiopus hurterii	Hurter's Spadefoot	E	WI, YR	V	V	V	CT	
Scaphiopus hurterii	Hurter's Spadefoot	T	SP	DN, SN	T [mt]	T [mt]	D [pf]	
Spea bombifrons	Plains Spadefoot	A	SP	V [r]	N/A	V [r]	N/A	
Spea bombifrons	Plains Spadefoot	BA	YR	A [a]	A [r]	A [a]	A [r]	3
Spea bombifrons	Plains Spadefoot	E	WI, YR	V	V	V	CT	
Spea bombifrons	Plains Spadefoot	T	SP	DN, SN	T [mt]	T [mt]	T [mt]	
NORTHEAST REGION								
Acris crepitans	Northern Cricket Frog	A	SP, SU	V	D [pf]	V	V [ce]	
Acris crepitans	Northern Cricket Frog	BA	SU	A [a]	A [r]	A [r]	F	
Acris crepitans	Northern Cricket Frog	T	SU, AU	DN	DN, SN	SN	DN, SN	
Acris gryllus	Southern Cricket Frog	A	SP, SU	V	D [pf]	V	V [ce]	
Acris gryllus	Southern Cricket Frog	BA	SU	A [a]	A [r]	A [r]	F	
Acris gryllus	Southern Cricket Frog	T	SU, AU	DN	DN, SN	SN	DN, SN	

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Bufo americanus	American Toad	A	SU	C, V [r]	D [pf]	D [pf]	D [pf]	
Bufo americanus	American Toad	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Bufo americanus	American Toad	E	SP	V	V	V	CT	
Bufo americanus	American Toad	T	SP, SU	V	D [pf]	D [pf]	D [pf]	
Bufo fowleri	Fowler's Toad	A	SU	V [r], C	D [pf]	D [pf]	D [pf]	
Bufo fowleri	Fowler's Toad	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Bufo fowleri	Fowler's Toad	E	SP	V	V	V	CT	
Bufo fowleri	Fowler's Toad	T	SP, SU	V	D [pf]	D [pf]	D [pf]	
Bufo quercicus	Oak Toad	A	SP, SU	V	D [pf]	C	C	
Bufo quercicus	Oak Toad	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Bufo quercicus	Oak Toad	E	SP, SU	V	A r	V	N/A	
Bufo quercicus	Oak Toad	T	SU	DN		SN	DN, SN	
Bufo terrestris	Southern Toad	A	SU	V [r], C	D [pf]	D [pf]	D [pf]	
Bufo terrestris	Southern Toad	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Bufo terrestris	Southern Toad	E	SP	V	V	V	CT	
Bufo terrestris	Southern Toad	T	SP, SU	DN	SN	SN	DN, SN	
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	A	SU	C	D [pf]	C	D [pf]	
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	BA	WI, SP	A [a]	A [r]	A [a]	D [pf]	
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	T	SP, SU	DN	SN	SN	DN, SN	
Hyla andersonii	Pine Barrens Treefrog	A	SP, SU	A [a]	A [r]	A [a]	A [d]	
Hyla andersonii	Pine Barrens Treefrog	BA	SP	A [a]	A [r]	A [a]	A [d]	
Hyla andersonii	Pine Barrens Treefrog	T	SU	DN	DN	DN	N/A	
Hyla chrysoscelis	Cope's Gray Treefrog	A	SU	V [r]	A [a]	A [a]	A [a]	
Hyla chrysoscelis	Cope's Gray Treefrog	BA	SP	A [a]	A [r]	A [a]	A [d]	
Hyla chrysoscelis	Cope's Gray Treefrog	T	SU	DN	DN	DN	N/A	
Hyla cinerea	Green Treefrog	A	SU	V [r]	V [r]	V [r]	A [d]	
Hyla cinerea	Green Treefrog	BA	SP	A [a]	A [r]	A [a]	A [d]	
Hyla cinerea	Green Treefrog	T	SU	DN	DN	DN	N/A	
Hyla femoralis	Pine Woods Treefrog	A	SU	V [r]	A [a]	T [pv]	A [a]	
Hyla femoralis	Pine Woods Treefrog	BA	SP	A [a]	A [r]	A [a]	A [d]	
Hyla femoralis	Pine Woods Treefrog	T	SU	DN	DN	DN	N/A	
Hyla gratiosa	Barking Treefrog	A	SU	V [r]	T [pv]	A [a]	T [pv]	
Hyla gratiosa	Barking Treefrog	BA	SP	A [a]	A [r]	A [a]	A [d]	
Hyla gratiosa	Barking Treefrog	T	SU	DN	DN	DN	DN	
Hyla squirella	Squirrel Treefrog	A	SU	V [r]	A [a]	T [pv]	A [a]	
Hyla squirella	Squirrel Treefrog	BA	SP	A [a]	A [r]	A [a]	A [d]	
Hyla squirella	Squirrel Treefrog	T	SU	DN	DN	DN	N/A	
Hyla versicolor	Gray Treefrog	A	SU	V [r]	A [a]	A [a]	A [d]	
Hyla versicolor	Gray Treefrog	BA	SP	A [a]	A [r]	A [a]	A [d]	
Hyla versicolor	Gray Treefrog	T	SU	DN	DN	DN	T [mt]	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Pseudacris brachyphona	Mountain Chorus Frog	A	WI, SP	A [a]	A [a]	A [a]	A [a]	
Pseudacris brachyphona	Mountain Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris brachyphona	Mountain Chorus Frog	T	SP	DN	DN	DN	N/A	
Pseudacris brimleyi	Brimley's Chorus Frog	A	WI, SP	A [a]	A [a]	A [a]	A [a]	
Pseudacris brimleyi	Brimley's Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris brimleyi	Brimley's Chorus Frog	T	SP	DN	DN	DN	N/A	
Pseudacris crucifer	Spring Peeper	A	WI, SP	V [r]	A [r]	A [r]	A [d]	2
Pseudacris crucifer	Spring Peeper	BA	WI, SP	A [a]	A [r]	A [a]	D [pf]	
Pseudacris crucifer	Spring Peeper	T	SP	DN	DN	DN	N/A	
Pseudacris feriarum	Upland Chorus Frog	A	WI, SP	A [a]	A [a]	A [a]	A [a]	
Pseudacris feriarum	Upland Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris feriarum	Upland Chorus Frog	T	SP	DN	DN	DN	N/A	
Pseudacris kalmi	New Jersey Chorus Frog	A	WI, SP	A [a]	A [a]	A [a]	D [pf]	
Pseudacris nigrita	Southern Chorus Frog	A	WI, SP	A [a]	A [r]	A [a]	D [pf]	
Pseudacris nigrita	Southern Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris nigrita	Southern Chorus Frog	T	SP	DN	DN	DN	N/A	
Pseudacris ocularis	Little Grass Frog	A	WI, SP	A [a]	A [r]	A [a]	N/A	
Pseudacris ocularis	Little Grass Frog	BA	SP	A [a]	A [a]	A [a]	N/A	
Pseudacris ocularis	Little Grass Frog	T	SP	DN	DN	DN	N/A	
Pseudacris triseriata	Western Chorus Frog	A	WI, SP	A [a]	A [r]	A [a]	D [pf]	
Pseudacris triseriata	Western Chorus Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris triseriata	Western Chorus Frog	T	SP	DN	DN	DN	N/A	
Rana catesbeiana	American Bullfrog	A	SP, SU, AU	V	V	V	V [ce]	
Rana catesbeiana	American Bullfrog	BA	SU	A [a]	A [r]	A [a]	A [d]	
Rana catesbeiana	American Bullfrog	T	YR	DN	DN, T [mt]	DN	DN	
Rana clamitans	Green Frog	A	SP, SU, AU	V	V	V	V [ce]	
Rana clamitans	Green Frog	BA	SU	A [a]	A [r]	A [a]	A [d]	
Rana clamitans	Green Frog	T	YR	DN	DN, T [mt]	DN	DN	
Rana palustris	Pickerel Frog	A	SP, SU	V [r]	D [pf]	D [pf]	N/A	
Rana palustris	Pickerel Frog	BA	SP	A [a] , V, C	C, V, A [r]	A [a]	A [d]	
Rana palustris	Pickerel Frog	T	SU, AU, WI	DN, SN	DN, SN, T [mt]	DN, SN	DN, SN, T [mt]	
Rana pipiens	Northern Leopard Frog	A	SU	V [r]	D [pf]	D [pf]	D [pf]	
Rana pipiens	Northern Leopard Frog	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Rana pipiens	Northern Leopard Frog	E	WI	V	V	V	CT	
Rana pipiens	Northern Leopard Frog	T	SP, SU	DN	DN, T [mt]	DN	D [pf]	
Rana septentrionalis	Mink Frog	A	SU	V	A [r]	A [a]	A [d]	
Rana sphenocephala	Southern Leopard Frog	A	SU	V [r]	D [pf]	D [pf]	MR, D [pf]	
Rana sphenocephala	Southern Leopard Frog	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Rana sphenocephala	Southern Leopard Frog	E	WI	V	V	V	CT	
Rana sphenocephala	Southern Leopard Frog	T	SP, SU	DN	DN, T [mt]	DN	T [mt]	

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Rana sylvatica	Wood Frog	A	SU	C	D [pf]	C	N/A	
Rana sylvatica	Wood Frog	BA	WI	A [a]	A [r]	A [a]	D [pf]	
Rana sylvatica	Wood Frog	E	WI	V	V	V	CT	
Rana sylvatica	Wood Frog	T	SP	DN	DN, T [mt]	DN	T [mt]	
Rana virgatipes	Carpenter Frog	A	SP, SU, AU	V	V	V	V [ce]	
Rana virgatipes	Carpenter Frog	BA	SP, SU	A [a]	A [r]	A [a]	A [d]	
Rana virgatipes	Carpenter Frog	T	YR	DN	DN, T [mt]	DN	DN	
Scaphiopus holbrookii	Eastern Spadefoot	A	YR	V [r]	N/A	V [r]	N/A	3
Scaphiopus holbrookii	Eastern Spadefoot	BA	WI, SP	A [a]	A [r]	A [a]	N/A	
Scaphiopus holbrookii	Eastern Spadefoot	E	W	V	V	V	CT	
Scaphiopus holbrookii	Eastern Spadefoot	T	WI, YR	V	V	V	T [mt]	
SOUTHWEST REGION								
Acris crepitans	Northern Cricket Frog	A	SP, SU, AU	A [a]	A [a]	A [r]	V [ce]	
Acris crepitans	Northern Cricket Frog	E	SP, SU	V	V	V	CT	
Acris crepitans	Northern Cricket Frog	J	SP, SU, AU	A [a]	A [a]	A [r]	V [ce]	
Acris crepitans	Northern Cricket Frog	T	SP, SU, AU	DN	DN, SN	SN	Dn, SN	
Ascaphus truei	Coastal Tailed Frog	A, J	YR	V	V	V	V [ce]	
Bufo alvarius	Colorado River Toad	A	SU, AU	V, A [a]	V, A [a]	V, A [a]	V [ce], D [pf]	
Bufo alvarius	Colorado River Toad	E	SU	V	V	V	CT, V [ce]	
Bufo alvarius	Colorado River Toad	T	SU, AU	DN	DN	DN	V [ce]	
Bufo americanus	American Toad	A	SP, SU, WI, SP	V [r], C, A [a]	D [pf], A [r]	D [pf], A [a]	D [pf], MR	
Bufo americanus	American Toad	E	SP	V	V	V	CT	
Bufo americanus	American Toad	T	SP, SU	DN	DN, SN	SN	DN, SN	
Bufo boreas	Western Toad	A	SP, SU	V, A [a]	A [a], V [ce]	V, A [a]	D [ft], V [ce]	
Bufo boreas	Western Toad	E	SP, SU	V	V	V	CT	
Bufo boreas	Western Toad	J	SP, SU	V	V	V	D [ft], V [ce]	
Bufo boreas	Western Toad	T	SP, SU	V, DN	V, DN, T [mt]	V, DN, T [mt]	V, DN, T [mt]	
Bufo californicus	Arroyo Toad	A	SP, SU	V	V	V	V [ce]	
Bufo californicus	Arroyo Toad	T, E	SP, SU	V	V	V	V [ce]	
Bufo canorus	Yosemite Toad	A	SP, SU	V	V	V	V [ce]	
Bufo canorus	Yosemite Toad	T, E	SP, SU	V	V	V	V [ce]	
Bufo cognatus	Great Plains Toad	A	YR	V	V	V	V [ce]	8
Bufo cognatus	Great Plains Toad	E	SP,SU	V	V	V	CT	
Bufo cognatus	Great Plains Toad	J	SU, AU	V	V	V	V [ce], D [pf]	
Bufo cognatus	Great Plains Toad	T	SU	V	DN	DN, SN	DN, SN	
Bufo debilis	Green Toad	A	SP, SU, AU	V, DN, A [a]	V, DN, A [a]	V, DN, A [a]	V, DN, A [a]	
Bufo debilis	Green Toad	E	SU	V, DN	V, DN	V, DN	CT, DN	
Bufo debilis	Green Toad	J	SU, AU	V, DN, T [mt]	V, DN, T [mt]	V, DN, T [mt]	V [ce], DN, T [mt]	
Bufo debilis	Green Toad	T	SU	DN	DN	DN	DN, SN	
Bufo exsul	Black Toad	A	SP, SU	V	V	V	V [ce]	
Bufo exsul	Black Toad	T, E	SP, SU	V	V	V	V [ce]	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Bufo hemiophrys	Canadian Toad	A	SP, SU	V, A [a]	V [ce], A [a]	V, A [a], D [pf]	V [ce]	
Bufo hemiophrys	Canadian Toad	T, E	SP, SU	V, DN	V, DN	V, DN	V [ce], DN	
Bufo houstonensis	Houston Toad	A	WI, SP	A [a]	A [r]	A [r]	D [pf], MR	4
Bufo houstonensis	Houston Toad	E	WI, SP	V	V	V	CT	
Bufo houstonensis	Houston Toad	T	SP	DN	DN, SN	SN	DN, SN	
Bufo microscaphus	Arizona Toad	A	SP, SU, AU	V	V, A [a]	V, A a	D [pf], MR	
Bufo microscaphus	Arizona Toad	E	SU, SP	V	V	V	CT, V [ce]	
Bufo microscaphus	Arizona Toad	T	SU, SP	DN, V	DN, V	DN	V [ce]	
Bufo nelsoni	Amargosa Toad	A	SP, SU	V	V	V	MR	
Bufo nelsoni	Amargosa Toad	E	SP	V	V [ce]	V	CT	
Bufo nelsoni	Amargosa Toad	M	SP, SU	V	V [ce]	V	V [ce]	
Bufo nelsoni	Amargosa Toad	T	SP, SU	V, DN	V [ce], DN, T [mt]	V [ce], DN, T [mt]	V [ce], DN, T [mt]	
Bufo punctatus	Red-spotted Toad	A	SP, SU, AU	V, A [a]	V, A [a]	V, A [a]	V [ce], A [d]	
Bufo punctatus	Red-spotted Toad	E	SU, SP	V, DN	V, DN	V, DN	V [ce], CT, DN	7
Bufo punctatus	Red-spotted Toad	J	SU, AU	V, DN	V, DN	V, DN	V [ce], DN	
Bufo punctatus	Red-spotted Toad	T	SU, SP	V, DN	V, DN	DN	V [ce]	
Bufo retiformis	Sonoran Green Toad	A	SP, SU	V	V	V	V [ce]	
Bufo retiformis	Sonoran Green Toad	E, T	SP, SU	V	V	V	V [ce]	
Bufo speciosus	Texas Toad	A	SP, SU, AU	V, V [r], A [a]	V, D [pf], A [a]	V, V [r], A [a]	D [pf], MR	
Bufo speciosus	Texas Toad	E	SU, SP	V	V	V	CT	
Bufo speciosus	Texas Toad	T	SU, AU	DN	DN	DN	V [ce]	
Bufo valliceps	Gulf Coast Toad	A	SP, SU	V [r], A [a], C	D [pf], A [r], V	D [pf], A [a], V	D [pf], MR	
Bufo valliceps	Gulf Coast Toad	E	SP, SU	V	V	V	CT	
Bufo valliceps	Gulf Coast Toad	T	SP, SU	DN, V	SN, V	SN	DN, SN	
Bufo woodhousii	Woodhouse's Toad	A	SP, SU, AU	A [a]	A [r]	A [a]	D [pf]	
Bufo woodhousii	Woodhouse's Toad	E	SP	V	V	V	CT	
Bufo woodhousii	Woodhouse's Toad	J	SU, AU	V	V	V	V [ce]	
Bufo woodhousii	Woodhouse's Toad	T	SP, SU	DN	SN	SN	DN, SN	
Eleutherodactylus augusti	Barking Frog	A	SP, SU	V, V [r], A [a]	D [pf], V, V [r], A [a]	D [pf], V, V [r], A [a]	D [pf], MR, V [r], A [d]	
Eleutherodactylus augusti	Barking Frog	E	SU, SP	F	F	F	TR [rt]	5
Eleutherodactylus augusti	Barking Frog	T		N/A	N/A	N/A	N/A	11
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	A	SP, SU	C, A [a]	D [pf], A [r]	C, A [a]	D [pf], MR	
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	T	SP, SU	DN	SN	DN	SN	
Gastrophryne olivacea	Western Narrow-mouthed Toad	A	SU	V, A [a]	V, A [a]	V, A [a]	V [ce], A [a]	
Gastrophryne olivacea	Western Narrow-mouthed Toad	E	SU	V, DN	V, DN	V, DN	V [ce], DN	
Gastrophryne olivacea	Western Narrow-mouthed Toad	J	SU	V	V	V	V [ce]	
Gastrophryne olivacea	Western Narrow-mouthed Toad	T	SU, SP	DN	DN	DN	V [ce], DN	6
Hyla arenicolor	Canyon Treefrog	A	SP, SU, AU	A [a], V	A [a], V	A [a], V	A [d], V [ce]	
Hyla arenicolor	Canyon Treefrog	E	SU, SP	V	V	V	V [ce]	
Hyla arenicolor	Canyon Treefrog	J	SU	V	V	V	V [ce]	
Hyla arenicolor	Canyon Treefrog	T	SU, SP	DN, V	DN, V	DN, N	V [ce]	

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Hyla chrysoscelis *Hyla versicolor*	Cope's Gray Treefrog Gray Treefrog	A	SP, SU	V [r]	A [a]	A [a]	A [a]	99
Hyla chrysoscelis *Hyla versicolor*	Cope's Gray Treefrog Gray Treefrog	BA	SP, SU	A [a]	A [r]	A [a]	A [d]	99
Hyla chrysoscelis *Hyla versicolor*	Cope's Gray Treefrog Gray Treefrog	T	SP, SU	DN	SN, T [mt]	SN	SN, T [mt]	99
Hyla cinerea	Green Treefrog	A	SP,SU	V [r]	V, V [r]	V [r], V, T [pv]	T [pv], MR	
Hyla cinerea	Green Treefrog	BA	SP, SU	A [a]	A [r]	A [a]	A [d], MR	
Hyla cinerea	Green Treefrog	T	SP, SU	DN	DN	DN	DN, T [mt]	
Hyla squirella	Squirrel Treefrog	A	SP, SU	V [r], A [a]	V [r], A [a]	V [r], A [a]	T [pv], MR	
Hyla squirella	Squirrel Treefrog	BA	SP, SU	A [a]	A [r]	A [a]	A [d], MR	
Hyla squirella	Squirrel Treefrog	T	SU	DN	SN, T [mt]	DN, SN, T [mt]	SN, T [mt]	
Hyla wrightorum	Arizona Tree Frog	A	SU	V, A [a]	V, A [a]	V, A [a]	MR, V [ce]	
Hyla wrightorum	Arizona Tree Frog	E	SU	V	V	V	CT	
Hyla wrightorum	Arizona Tree Frog	T	SU	DN	DN	DN	V [ce]	
Hypopachus variolosus	Sheep Frog	A	SU	C	C	C	C, D [pf], MR	
Hypopachus variolosus	Sheep Frog	BA	SP, SU	A [a]	A [r]	A [a]	D [pf], MR	
Hypopachus variolosus	Sheep Frog	T	SU	DN	SN	SN	DN, SN	
Leptodactylus fragilis	Mexican White-lipped Frog	BA	SP, SU, AU	A [a]	A [r]	A [a]	A [d], MR	
Leptodactylus fragilis	Mexican White-lipped Frog	A	SU	V, V [r]	V, V [r]	V, V [r]	V, V [r]	
Leptodactylus fragilis	Mexican White-lipped Frog	E	SU	N/A	N/A	N/A	N/A	12
Leptodactylus fragilis	Mexican White-lipped Frog	T	SU, AU	DN	SN	SN	DN, SN	
Pseudacris cadaverina	California Tree Frog	A	SP, SU	V	V	V	V [ce]	
Pseudacris cadaverina	California Tree Frog	E, T	SP, SU	V	V	V	V [ce]	
Pseudacris clarkii	Spotted Chorus Frog	A	WI, SP, SU, AU	D [pf]	D [pf]	D [pf]	MR, D [pf]	
Pseudacris clarkii	Spotted Chorus Frog	BA	WI, SP, SU, AU	A [a]	A [r]	A [a]	A [d], MR	
Pseudacris clarkii	Spotted Chorus Frog	T	SP, SU, AU	DN	SN	SN	DN, SN, T [mt]	
Pseudacris crucifer	Spring Peeper	A	WI, SP	V [r]	V [r]	V [r]	D [pf]	
Pseudacris crucifer	Spring Peeper	BA	WI, SP	A [a]	A [r]	A [a]	D [pf]	
Pseudacris crucifer	Spring Peeper	T	SP	DN	DN, SN	DN	DN, SN, T [mt]	
Pseudacris hypochondriaca	Baja California Treefrog	A	SP, SU	V	V	V	V [ce]	
Pseudacris hypochondriaca	Baja California Treefrog	E, T	SP, SU	V	V	V	V [ce]	
Pseudacris maculata	Boreal Chorus Frog	A	SP, SU, AU	V	V [ce]	V	D [pf], V [ce]	
Pseudacris maculata	Boreal Chorus Frog	E	SP, SU	V	V	V	CT, V [ce]	
Pseudacris maculata	Boreal Chorus Frog	J	SU, AU	V, DN	V, DN	V, DN, T [mt]	DN, V [ce]	
Pseudacris regilla	Northern Pacific Chorus Frog	A	SP	V, A [a]	V, A [a]	A [a], V, T [pv]	V [ce], A [d], T [pv]	
Pseudacris regilla	Northern Pacific Chorus Frog	E	SP	V	V	V	CT	
Pseudacris regilla	Northern Pacific Chorus Frog	T	SP, SU	V	V	V, DN	DN, T [mt]	
Pseudacris regilla	Northern Pacific Chorus Frog	L	SP, SU	V, DN	V, DN, T [mt]	V, DN	DN, V [ce], T [mt]	
Pseudacris regilla	Northern Pacific Chorus Frog	M	SU, AU	V	V [ce]	V	V [ce]	
Pseudacris streckeri	Strecker's Chorus Frog	A	SP	V, A [a]	V, A [a]	V, A [a]	V [ce], A [d]	
Pseudacris streckeri	Strecker's Chorus Frog	BA	WI, SP	A [a]	A [r]	A [a]	D [pf]	
Pseudacris streckeri	Strecker's Chorus Frog	T	SP	DN	DN; SN	DN	DN, SN, T [mt]	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Pseudacris triseriata	Western Chorus Frog	A	SP, SU, AU, WI	A [a]	A [r]	A [a]	D [pf]	
Pseudacris triseriata	Western Chorus Frog	E	SU	V	V	V	V [ce]	
Pseudacris triseriata	Western Chorus Frog	J	SU, AU	V	V	V	V [ce]	
Pseudacris triseriata	Western Chorus Frog	T	SP, SU	DN	DN	DN		
Pseudacris triseriata	Western Chorus Frog	BA	WI, SP	A [a]	A [r]	A [a]	D [pf], MR	
Pternohyla fodiens	Lowland Burrowing Treefrog	A	SP, SU	V	V	V	V [ce]	
Pternohyla fodiens	Lowland Burrowing Treefrog	E, T	SP, SU	V	V	V	V [ce]	
Rana areolata	Crawfish Frog	A	SP	V [b]	V [b]	V [b]	D [pf]	
Rana areolata	Crawfish Frog	BA	WI, SP	A [a]	A [r]	A [a]	D [pf]	
Rana areolata	Crawfish Frog	E	WI	V	V	V	CT	
Rana areolata	Crawfish Frog	T	SP, SU	DN	DN, T [mt]	DN	T [mt], D [pf]	
Rana aurora	Northern Red-legged Frog	A	SP, SU	V	V	V	V [ce]	
Rana aurora	Northern Red-legged Frog	E, T	SP, SU	V	V	V	V [ce]	
Rana berlandieri	Rio Grande Leopard Frog	A	YR	V [r], A [a]	D [pf], A [a]	D [pf], A [a]	D [pf]	
Rana berlandieri	Rio Grande Leopard Frog	BA	WI, SP, AU	A [a]	A [r]	A [a]	D [pf], MR	
Rana berlandieri	Rio Grande Leopard Frog	E	WI, SP, AU	V	V	V	CT	
Rana berlandieri	Rio Grande Leopard Frog	T	YR	DN	DN, T [mt]	DN	V [ce]	
Rana blairi	Plains Leopard Frog	A	SP, SU, AU	V [r]	D [pf]	D [pf]	D [pf]	
Rana blairi	Plains Leopard Frog	BA	SP, SU	A [a]	A [r]	A [a]	D [pf]	
Rana blairi	Plains Leopard Frog	E	SP, SU	V	V	V	CT	
Rana blairi	Plains Leopard Frog	J	SU, AU	V	V	V	V [ce]	
Rana blairi	Plains Leopard Frog	T	YR	DN	DN, T [mt]	DN	D [pf]	
Rana boylii	Foothill Yellow-legged Frog	A	SP, SU	V	V	V	V [ce]	
Rana boylii	Foothill Yellow-legged Frog	E, T	SP, SU	V	V	V	V [ce]	
Rana cascadae	Cascades Frog	A	SP, SU	V	V	V	V [ce]	
Rana cascadae	Cascades Frog	E, T	SP, SU	V	V	V	V [ce]	
Rana catesbeiana	American Bullfrog	A	SP, SU, AU	V	V	V	V [ce]	
Rana catesbeiana	American Bullfrog	BA	SP, SU	A [a]	A [r]	A [a]	A [d]	
Rana catesbeiana	American Bullfrog	E	SU	V	V	V	V [ce]	
Rana catesbeiana	American Bullfrog	J	YR	V	V	V	V [ce]	
Rana catesbeiana	American Bullfrog	T	YR	DN	DN	DN	DN, V [ce]	
Rana chiricahuensis	Chiricahua Leopard Frog	A	SP, SU, AU	V, A [a]	V, A [a]	V, A [a]	A [d], MR, D [pf]	
Rana chiricahuensis	Chiricahua Leopard Frog	E	SU	V	V	V	CT	
Rana chiricahuensis	Chiricahua Leopard Frog	T	YR	DN	DN	DN	DN, V [ce]	
Rana clamitans	Green Frog	A	SP, SU, AU	V	V	V	V [ce], D [pf]	
Rana clamitans	Green Frog	BA	SP, SU	A [a]	A [r]	A [a]	A [d]	
Rana clamitans	Green Frog	T	YR	DN	DN, T [mt]	DN	DN, T [mt]	
Rana draytonii	California Red-legged Frog	A	SP, SU	V, V [r]	V, V [r]	V	V [ce]	
Rana draytonii	California Red-legged Frog	E, T	SP, SU	V, DN	V, DN	V, DN	DN, T [mt]	
Rana grylio	Pig Frog	A	SP, SU, AU	V, A [a]	V, A [a]	V, A [a]	V, A [d]	
Rana grylio	Pig Frog	BA	SP, SU	A [a]	A [r]	A [a]	A [d]	
Rana grylio	Pig Frog	T	YR	DN	DN, T [mt]	DN	DN, T [mt]	

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Rana luteiventris	Columbia Spotted Frog	A	SP	V	V	V	MR	
Rana luteiventris	Columbia Spotted Frog	E	SP	V	V	V	CT	
Rana muscosa	Mountain Yellow-legged Frog	A	SP, SU	V	V	V	V [ce]	
Rana muscosa	Mountain Yellow-legged Frog	E, T	SP, SU	V	V	V	V [ce], CT	
Rana onca	Relict Leopard Frog	A	YR	V	V	V	V [ce]	
Rana onca	Relict Leopard Frog	All	SP	V	V	V	V [ce]	
Rana onca	Relict Leopard Frog	E	SP	V	V	V	CT	
Rana palustris	Pickerel Frog	A	SP	V [r]	D [pf]	D [pf]	D [pf]	
Rana palustris	Pickerel Frog	BA	WI, SP	A [a] , V, C	C, V, A [r]	A [a]	A [d]	
Rana palustris	Pickerel Frog	T	SU, AU, WI	DN, SN	DN, SN, T [mt]	DN, SN	DN, SN, T [mt]	
Rana pipiens	Northern Leopard Frog	A	SP, SU, AU	V [r]	D [pf]	D [pf]	D [pf]	
Rana pipiens	Northern Leopard Frog	E	SP, SU	V	V	V	CT	
Rana pipiens	Northern Leopard Frog	J	SU, AU	V	V	V	V [ce]	
Rana pipiens	Northern Leopard Frog	T	YR	DN	DN, T [mt]	DN	DN, D [pf]	
Rana pretiosa	Oregon Spotted Frog	A	SP, SU	V	V	V	V [ce]	
Rana pretiosa	Oregon Spotted Frog	E, T	SP, SU	V, DN	V, DN	V, DN	DN, T [mt]	
Rana sierrae	Sierra Nevada Yellow-legged Frog	A	SP, SU	V	V [ce]	V	V [ce]	
Rana sierrae	Sierra Nevada Yellow-legged Frog	E, T	SP, SU	V	V [ce]	V	V [ce]	
Rana sphenocephala	Southern Leopard Frog	A	SP, SU, AU	V [r]	D [pf]	D [pf]	MR, D [pf]	
Rana sphenocephala	Southern Leopard Frog	BA	AU, WI, SP, SU	A [a]	A [r]	A [a]	D [pf]	
Rana sphenocephala	Southern Leopard Frog	E	WI	V	V	V	CT	
Rana sphenocephala	Southern Leopard Frog	T	SP, SU	DN	DN, T [mt]	DN	T [mt]	
Rana sylvatica	Wood Frog	A	SP, SU, AU	C	D [pf]	C	N/A	
Rana sylvatica	Wood Frog	E	SP, SU	V	V	V	CT	
Rana sylvatica	Wood Frog	J	SU, AU	DN	DN, T [mt]	DN	T [mt]	
Rana tarahumarae	Tarahumara Frog	A	SP, SU, AU	V, A [a]	V, A [a]	V, A [a]	T [mt], MR	
Rana yavapaiensis	Lowland Leopard Frog	A	SP, SU, AU	V, A [a]	V, A [a]	V, A [a]	T [mt], MR	
Rhinophrynus dorsalis	Mexican Burrowing Toad	A	N/A	N/A	N/A	N/A	N/A	8
Rhinophrynus dorsalis	Mexican Burrowing Toad	BA	SP, SU, AU	A [a]	A [a]	A [a]	A [d]	
Rhinophrynus dorsalis	Mexican Burrowing Toad	E	SP, SU, AU	N/A	N/A	N/A	N/A	10
Rhinophrynus dorsalis	Mexican Burrowing Toad	T	SP, SU, AU	DN	DN	DN	DN	
Scaphiopus couchii	Couch's Spadefoot	A	YR, SU, AU	V [r]	D [pf]	V [r]	D [pf]	9
Scaphiopus couchii	Couch's Spadefoot	BA	YR	A [a]	A [r]	A [a]	D [pf]	
Scaphiopus couchii	Couch's Spadefoot	E	SU, YR	V	V	V	CT	
Scaphiopus couchii	Couch's Spadefoot	J	SU, AU	V	V	V	V [ce]	
Scaphiopus couchii	Couch's Spadefoot	T	SU, YR	V, DN	V, DN	V, DN	T [mt]	
Scaphiopus hurterii	Hurter's Spadefoot	A	YR	V [r]	V	V [r]	V [ce], V [r]	
Scaphiopus hurterii	Hurter's Spadefoot	BA	YR	A [a]	A [r]	A [a]	A [d]	
Scaphiopus hurterii	Hurter's Spadefoot	E	YR	V	V	V	CT	
Scaphiopus hurterii	Hurter's Spadefoot	T	YR	V	V	V	T [mt]	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Smilisca baudinii	Mexican Treefrog	A	SP, SU, AU	V [r]	A [a]	V, V [r]	T [pv], A [d]	
Smilisca baudinii	Mexican Treefrog	BA	SP, SU, AU	A [a]	A [r]	A [a]	CT, A [d]	
Smilisca baudinii	Mexican Treefrog	E	SP, SU, AU	N/A	N/A	N/A	N/A	10
Smilisca baudinii	Mexican Treefrog	T	SP, SU, AU	DN	DN	DN	DN	
Spea bombifrons	Plains Spadefoot	A	SP, SU, AU	V	V	V	V [ce]	
Spea bombifrons	Plains Spadefoot	BA	SP, SU, AU	A [a]	A [r]	A [a]	A [d]	
Spea bombifrons	Plains Spadefoot	E	SU, AU, SP	V	V	V	CT	
Spea bombifrons	Plains Spadefoot	J	SU, AU	V	V	V	V [ce]	
Spea bombifrons	Plains Spadefoot	T	SP, SU, AU	DN, SN	T [mt]	T [mt]	T [mt]	
Spea hammondi	Western Spadefoot	A	SP, SU	V	V	V	MR, V [ce]	
Spea hammondi	Western Spadefoot	E, T	SP, SU	V	V	V	CT, V [ce]	
Spea intermontana	Great Basin Spadefoot	A	SP, SU, AU	V, A [a]	V [ce]	V, A [a]	D [pf], A [d], V [ce]	
Spea intermontana	Great Basin Spadefoot	E	SP, SU	V	V	V	CT	
Spea intermontana	Great Basin Spadefoot	J	SU, AU	V	V	V	V [ce]	
Spea intermontana	Great Basin Spadefoot	T	SP, SU	V, DN	V, DN	V, DN	V, DN, T [mt]	
Spea multiplicata	Mexican Spadefoot	A	SU, AU	V, A [a]	V [ce]	V, A [a]	D [pf], A [d], V [ce]	
Spea multiplicata	Mexican Spadefoot	E	SU,AU	V	V	V	CT	
Spea multiplicata	Mexican Spadefoot	J	SU, AU	V	V	V	V [ce]	
Spea multiplicata	Mexican Spadefoot	T	SU, AU	V, DN	V, DN	V, DN	V, DN, T [mt]	
Syrrhophus cystignathoides	Rio Grande Chirping Frog	A	SP, SU, AU	A [a]	A [r]	A [a]	F, A [d]	
Syrrhophus cystignathoides	Rio Grande Chirping Frog	E	SP	SF	TR [rt]	SF	TR [rt]	
Syrrhophus cystignathoides	Rio Grande Chirping Frog	T	N/A	N/A	N/A	N/A	N/A	11
Syrrhophus gutillatus	Spotted Chirping Frog	A	SP, SU, AU	A [a]	A [r]	C, A [a]	F, A [d]	
Syrrhophus gutillatus	Spotted Chirping Frog	E	SP	N/A	TR [rt]	N/A	TR [rt]	
Syrrhophus gutillatus	Spotted Chirping Frog	T	N/A	N/A	N/A	N/A	N/A	100
Syrrhophus marnockii	Cliff Chirping Frog	A	SP, SU, AU	A [a]	A [r]	A [a]	F, A [d]	
Syrrhophus marnockii	Cliff Chirping Frog	E	SP	N/A	TR [rt]	N/A	TR [rt]	
Syrrhophus marnockii	Cliff Chirping Frog	T	N/A	N/A	N/A	N/A	N/A	11
NORTHWEST REGION								
Ascaphus montanus	Rocky Mountain Tailed Frog	A	YR	V [ce]	V [s], V [ce]	V [s], V [ce]	V [s], V [ce]	14, 101
Ascaphus montanus	Rocky Mountain Tailed Frog	L	YR	V [ks], V [ce]	V [s], V [ce]	V [s], V [ce]	V [s], V [ce]	13, 101
Ascaphus truei	Coastal Tailed Frog	A	YR	V	V, V [s]	V	V [ce]	14, 101
Ascaphus truei	Coastal Tailed Frog	L	YR	V [ks], V [ce]	V [s], V [ce]	V [s], V [ce]	V [s], V [ce]	13, 101
Bufo baxteri	Wyoming Toad	BA	SP, SU	V, A [a]	A [a], V [ce]	V, A [a]	D [ft], V [ce]	
Bufo baxteri	Wyoming Toad	E	SP, SU	V	V [ce]	V	CT	
Bufo baxteri	Wyoming Toad	L	SP, SU	V, DN	V, DN, T [mt]	V, DN, T [mt]	V, DN, T [mt]	
Bufo baxteri	Wyoming Toad	M	SU, AU	V	V [ce]	V	D [ft], V [ce]	15
Bufo boreas	Western Toad	BA	SP, SU	V, A [a], V [r]	A [a], V [ce]	V, A [a]	D [pf], V [ce], CT	17
Bufo boreas	Western Toad	E	SP, SU	V	V, V [ce]	V	CT	
Bufo boreas	Western Toad	L	SP, SU	V, DN	V, DN, T [mt]	V, DN, T [mt]	V [ce], DN, T [mt]	
Bufo boreas	Western Toad	M	SU, AU	V	V [ce]	V	D [pf], V [ce]	

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Bufo cognatus	Great Plains Toad	BA	SP, SU	V, A [a]	V, A [a]	V, A [a]	D [ft], V [ce]	
Bufo cognatus	Great Plains Toad	E	SP, SU	V	V [ce]	V	CT	
Bufo cognatus	Great Plains Toad	L	SP, SU	V, DN	V, DN, T [mt]	V, DN, T [mt]	V [ce], DN, T [mt]	
Bufo cognatus	Great Plains Toad	M	SU, AU	V	V [ce]	V	V [ce], D [pf]	
Bufo hemiophrys	Canadian Toad	BA	SP, SU	V, A [a]	V [ce], A a	V, A [a], D [pf]	D [ft], V [ce]	
Bufo hemiophrys	Canadian Toad	E	SP, SU	V	V	V	CT	
Bufo hemiophrys	Canadian Toad	L	SP, SU	V, DN	V, DN	V, DN	V [ce], DN	
Bufo hemiophrys	Canadian Toad	M	SU, AU	V	V [ce]	V	D [ft], V [ce]	
Bufo woodhousii	Woodhouse's Toad	BA	SP	A [a]	A [r]	A [a]	D [pf]	16
Bufo woodhousii	Woodhouse's Toad	E	SP	V	V	V	CT	
Bufo woodhousii	Woodhouse's Toad	L	SP, SU	DN	SN	SN	DN, SN	
Bufo woodhousii	Woodhouse's Toad	M	SU	V	V [ce]	V	D [pf], V [ce]	16
Pseudacris maculata	Boreal Chorus Frog	BA	SP, SU	V, A [a]	V [ce]	A [a], A [d], V, T [pv]	D [pf], V [ce]	18, 102
Pseudacris maculata	Boreal Chorus Frog	E	SP, SU	V	V	V	CT, V [ce]	
Pseudacris maculata	Boreal Chorus Frog	L	SP, SU	V, DN	V, DN	V, DN, T [mt]	DN, V [ce], T [mt]	
Pseudacris maculata	Boreal Chorus Frog	M	SU, AU	V	V [ce]	V	D [pf], V [ce]	16, 102
Pseudacris regilla	Northern Pacific Chorus Frog	BA	SP, SU	V, A [a]	V, A [a]	A [a], V, T [pv]	V [ce], A [d], T [pv]	
Pseudacris regilla	Northern Pacific Chorus Frog	E	SP, SU	V	V	V	CT	
Pseudacris regilla	Northern Pacific Chorus Frog	L	SP, SU	V, DN	V, DN	V, DN	DN, T [mt]	
Pseudacris regilla	Northern Pacific Chorus Frog	M	SU, AU	V	V [ce]	V	V [ce]	
Pseudacris sierra	Sierran Chorus Frog	E	SP, SU	V	V	V	CT, V [ce]	
Pseudacris sierra	Sierran Chorus Frog	BA	SP, SU	V, A [a]	V, A [a]	A [a], V, T [pv]	V [ce], A [d], T [pv]	19
Pseudacris sierra	Sierran Chorus Frog	L	SP, SU	V, DN	V, DN, T [mt]	V, DN, T [mt]	DN, T [mt], V [ce]	
Pseudacris sierra	Sierran Chorus Frog	M	SU, AU	V	V [ce]	V	V [ce]	
Rana aurora	Northern Red-legged Frog	BA	WI, SP	V, A [a]	V	V	D [pf], V [ce]	21
Rana aurora	Northern Red-legged Frog	E	WI, SP	V	V	V	CT, V [ce]	
Rana aurora	Northern Red-legged Frog	J, A	YR	V, V [r]	V, V [r]	V, V [r]	D [pf], V [ce], V [r]	20
Rana aurora	Northern Red-legged Frog	L	SP, SU	V, DN	V, DN	V, DN, T [mt]	DN, T [mt]	
Rana boylii	Foothill Yellow-legged Frog	A	YR	V	V	V	V [ce]	
Rana boylii	Foothill Yellow-legged Frog	E	SP	V	V	V	V [ce]	
Rana boylii	Foothill Yellow-legged Frog	L	SP, SU	V, DN	V, DN	V, DN	DN, T [mt]	
Rana cascadae	Cascades Frog	BA	SP	V	V	V	V [ce]	21
Rana cascadae	Cascades Frog	E	SP	V	V	V	V [ce]	
Rana cascadae	Cascades Frog	J, A	SU, AU	V	V	V	V [ce]	20
Rana cascadae	Cascades Frog	L	SP, SU	V, DN	V, DN, T mt	V, DN, T [mt]	DN, T [mt]	
Rana catesbeiana	American Bullfrog	BA	SU	A [a], V	A [r], V	A [a], V	A [d], V	
Rana catesbeiana	American Bullfrog	E	SU	V	V	V	CT, V [ce]	
Rana catesbeiana	American Bullfrog	J, A	SP, SU, AU	V	V	V	V [ce]	
Rana catesbeiana	American Bullfrog	L	SP, SU, AU	DN, V	V, DN, T mt	V, DN, T [mt]	DN, V [ce], T [mt]	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Rana clamitans	Green Frog	BA	SU	A [a]	A [r]	A [a]	A [d]	
Rana clamitans	Green Frog	E	SU	V	V	V	CT, V [ce]	
Rana clamitans	Green Frog	J, A	SP, SU, AU	V	V	V	V [ce]	
Rana clamitans	Green Frog	L	SP, SU, AU	DN, V	DN, T [mt], V	V, DN, T [mt]	DN, T [mt], V [ce]	
Rana draytonii	California Red-legged Frog	BA	WI, SP	V, V [r]	V, V [r]	V	V [ce]	21
Rana draytonii	California Red-legged Frog	E	WI, SP	V	V	V	V [ce]	
Rana draytonii	California Red-legged Frog	J, A	YR	V, V [r]	V, V [r]	V	V [ce]	20
Rana draytonii	California Red-legged Frog	L	SP, SU	V, DN	V, DN	V, DN	DN, T [mt]	
Rana luteiventris	Columbia Spotted Frog	BA	SP	V, A [a]	V, A [a]	V, A [a]	V [ce]	21
Rana luteiventris	Columbia Spotted Frog	E	SP	V	V	V	CT, V [ce]	
Rana luteiventris	Columbia Spotted Frog	J, A	SU, AU	V	V, V [ce]	V	V [ce]	20
Rana luteiventris	Columbia Spotted Frog	L	SP, SU	V, DN	V, DN, T mt	V, DN, T mt	DN, T [mt], V [ce]	
Rana pipiens	Northern Leopard Frog	BA	SP	A [a]	A [r]	A [a]	D [pf]	
Rana pipiens	Northern Leopard Frog	E	SP	V	V	V	CT	
Rana pipiens	Northern Leopard Frog	J, A	SP, SU, AU	V [r]	D [pf]	D [pf], V	D [pf]	
Rana pipiens	Northern Leopard Frog	L	SP, SU	DN, V	DN, T [mt], V	V, DN, T mt	DN	
Rana pretiosa	Oregon Spotted Frog	BA	SP	A [a], V	A [a], V [ce]	A [a], V	V [ce]	21
Rana pretiosa	Oregon Spotted Frog	E	SP	V	V	V	V [ce], CT	
Rana pretiosa	Oregon Spotted Frog	J, A	SP, SU, AU	V	V [ce]	V	V [ce]	20
Rana pretiosa	Oregon Spotted Frog	L	SP, SU	V, DN	V, DN, T mt	V, DN, T mt	DN, V [ce], T [mt]	
Rana sylvatica	Wood Frog	BA	SP	A [a]	A [r]	A [a]	D [pf]	15
Rana sylvatica	Wood Frog	E	SP	V	V	V	CT	
Rana sylvatica	Wood Frog	L	SP	DN, V	DN, T [mt], V	V, DN, T [mt]	T [mt]	
Rana sylvatica	Wood Frog	M	SU	V	V	V	V [ce], D [pf]	
Spea bombifrons	Plains Spadefoot	BA	SP, SU	A [a]	A [r]	A [a]	D [pf], A [d]	15
Spea bombifrons	Plains Spadefoot	E	SP, SU	V	V	V	CT	
Spea bombifrons	Plains Spadefoot	L	SP, SU	DN, SN	T [mt]	T [mt]	T [mt]	
Spea bombifrons	Plains Spadefoot	M	SU, AU	VES	V	V	V [ce], D [pf]	15
Spea hammondii	Western Spadefoot	BA	SP, SU	V, A [a]	V [ce]	V, A [a]	D [pf], A [d], V [ce]	15
Spea hammondii	Western Spadefoot	E	SP, SU	V	V	V	CT	
Spea hammondii	Western Spadefoot	L	SP, SU	V, DN	V, DN	V, DN	V, DN, T [mt]	
Spea hammondii	Western Spadefoot	M	SU, AU	V	V	V	V [ce], D [pf]	15
Spea intermontana	Great Basin Spadefoot	BA	SP	V, A [a]	V [ce]	V, A [a]	D [pf], A [d], V [ce]	15
Spea intermontana	Great Basin Spadefoot	E	SP	V	V	V	CT	
Spea intermontana	Great Basin Spadefoot	L	SP	V, DN	V, DN, T mt	V, DN, T mt	V, DN, T [mt]	
MIDWEST REGION								
Acris crepitans	Northern Cricket Frog	A	SP, SU, AU	A [a]	A [a]	A [r]	V [ce]	23
Acris crepitans	Northern Cricket Frog	L	SU	DN	DN, SN	SN	Dn, SN	22
Bufo americanus	American Toad	A	SP, SU	V [r], C, A [a]	D [pf], A [r]	D [pf], A [a]	D [pf], MR	26
Bufo americanus	American Toad	E	SP	V	V	V	CT	24
Bufo americanus	American Toad	L	SP	V	DN	DN	D [pf]	25

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Bufo cognatus	Great Plains Toad	BA	SP, SU	V, A [a]	V, A [a]	V, A [a]	V [ce]	29
Bufo cognatus	Great Plains Toad	E	SP, SU	V	V [ce]	V	CT	27
Bufo cognatus	Great Plains Toad	L	SP, SU	V, DN	V, DN, T [mt]	V, DN, T [mt]	V [ce], DN, T [mt]	28
Bufo debilis	Green Toad	BA	SP, SU	V, DN, A [a]	V, DN, A [a]	V, DN, A [a]	V, DN, A [a]	29
Bufo debilis	Green Toad	E	SP, SU	V, DN	V, DN	V, DN	CT, DN	30
Bufo debilis	Green Toad	L	SP, SU	DN	DN	DN	DN, SN	31
Bufo fowleri	Fowler's Toad	A	SP, SU	V [r], C	D [pf]	D [pf]	D [pf]	34
Bufo fowleri	Fowler's Toad	E	SP	V	V	V	CT	32
Bufo fowleri	Fowler's Toad	L	SP	V	D [pf]	D [pf]	D [pf]	33
Bufo hemiophrys	Canadian Toad	A	SP, SU	V, A [a]	V [ce], A [a]	V, A [a], D [pf]	V [ce]	34
Bufo hemiophrys	Canadian Toad	E	SP	V	V	V	CT	35
Bufo hemiophrys	Canadian Toad	L	SP	V, DN	V, DN	V, DN	V [ce], DN	33
Bufo punctatus	Red-spotted Toad	BA	SP, SU	V, A [a]	V, A [a]	V, A [a]	V [ce], A [d]	29
Bufo punctatus	Red-spotted Toad	L	SP, SU	V, DN	V, DN	DN	V [ce]	36
Bufo woodhousii	Woodhouse's Toad	BA	SP, SU	A [a]	A [r]	A [a]	D [pf]	38
Bufo woodhousii	Woodhouse's Toad	E	SP, SU	V	V	V	CT	37
Bufo woodhousii	Woodhouse's Toad	L	SP, SU	DN	SN	SN	DN, SN	28
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	A	SP, SU	A [a]	A [r]	A [a]	D [pf]	40
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	L	SP	V, DN, SS	V, DN, SS	V, DN, SS	V, DN, SS	39
Gastrophryne olivacea	Western Narrow-mouthed Toad	BA	SP, SU	V, A [a]	V, A [a]	V, A [a]	V [ce], A [a]	41
Gastrophryne olivacea	Western Narrow-mouthed Toad	L	SP, SU	V	V	V	V [ce]	28
Hyla avivoca	Bird-voiced Treefrog	A	SP, SU	A [a]	A [a]	A [a]	A [d]	43
Hyla avivoca	Bird-voiced Treefrog	L	SU	V, T [mt]	V, T [mt]	V, T [mt]	V, T [mt]	42
Hyla chrysoscelis	Cope's Gray Treefrog	A	SP, SU	A [a]	A [r]	A [a]	A [d]	45
Hyla chrysoscelis	Cope's Gray Treefrog	L	SU	DN	DN	DN	N/A	44
Hyla cinerea	Green Treefrog	A	SP, SU	V [r]	A [a]	A [a]	A [a]	47
Hyla cinerea	Green Treefrog	L	SU	DN	DN	DN	DN, T [mt]	46
Hyla versicolor	Gray Treefrog	A	SP, SU	V [r]	A [a]	A [a]	A [d]	49
Hyla versicolor	Gray Treefrog	L	SU	DN	DN	DN	T [mt]	48
Pseudacris brachyphona	Mountain Chorus Frog	A	SP	A [a]	A r	A [a]	D [pf]	51
Pseudacris brachyphona	Mountain Chorus Frog	L	SU	DN	DN	DN	T [mt]	50
Pseudacris clarkii	Spotted Chorus Frog	A	SP	V	V	V	V [ce]	53
Pseudacris clarkii	Spotted Chorus Frog	L	SU	DN	SN	SN	DN, SN, T [mt]	52
Pseudacris crucifer	Spring Peeper	A	SP	V [r]	A [r]	A [r]	A [d]	55
Pseudacris crucifer	Spring Peeper	L	SU	DN	DN	DN	T [mt]	54
Pseudacris feriarum	Upland Chorus Frog	A	SP	A [a]	A r	A [a]	D [pf]	57
Pseudacris feriarum	Upland Chorus Frog	L	SU	DN	DN	DN	T [mt]	56
Pseudacris illinoensis	Illinois Chorus Frog	A	SP	V, A [a]	V, A [a]	V, A [a]	V [ce], A [d]	63
Pseudacris illinoensis	Illinois Chorus Frog	L	SU	V	V	V	V [ce], T [mt]	62
Pseudacris maculata	Boreal Chorus Frog	A	SP	V	V [ce]	V	D [pf], V [ce]	59
Pseudacris maculata	Boreal Chorus Frog	L	SU	V, DN	V, DN	V, DN, T [mt]	DN, V [ce]	58

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Pseudacris streckeri	Strecker's Chorus Frog	A	SP	V, A [a]	V, A [a]	V, A [a]	V [ce], A [d]	61
Pseudacris streckeri	Strecker's Chorus Frog	L	SU	DN	DN	DN		60
Pseudacris triseriata	Western Chorus Frog	A	SP	A [a]	A [r]	A [a]	D [pf]	65
Pseudacris triseriata	Western Chorus Frog	L	SU	DN	DN	DN	T [mt]	64
Rana areolata	Crawfish Frog	A	SP	V [b]	V [b]	V [b]	D [pf]	68
Rana areolata	Crawfish Frog	E	SP	V	V	V	CT	66
Rana areolata	Crawfish Frog	L	SP, SU	DN	DN, T [mt]	DN	T [mt], D [pf]	67
Rana blairi	Plains Leopard Frog	A	SP	A [a]	A [r]	A [a]	D [pf]	71
Rana blairi	Plains Leopard Frog	E	SP	V	V	V	CT	69
Rana blairi	Plains Leopard Frog	L	SP, SU	DN	DN, T [mt]	DN	D [pf]	70
Rana catesbeiana	American Bullfrog	A	SP, SU	A [a]	A [r]	A [a]	A [d]	74
Rana catesbeiana	American Bullfrog	E	SP, SU	V	V	V	V [ce]	72
Rana catesbeiana	American Bullfrog	L	SP, SU, AU	DN	DN	DN	DN, V [ce]	73
Rana clamitans	Green Frog	A	SP, SU	A [a]	A [r]	A [a]	A [d]	77
Rana clamitans	Green Frog	E	SP	V	V	V	CT	75
Rana clamitans	Green Frog	L	SP, SU	DN	DN, T [mt]	DN	DN, T [mt]	76
Rana palustris	Pickerel Frog	A	SP, SU	A [a] , V, C	C, V, A [r]	A [a]	A [d]	80
Rana palustris	Pickerel Frog	E	SP	V	V	V	CT	78
Rana palustris	Pickerel Frog	L	SP, SU	DN, SN	DN, SN, T [mt]	DN, SN	DN, SN, T [mt]	79
Rana pipiens	Northern Leopard Frog	A	SP, SU	A [a]	A [r]	A [a]	D [pf]	83
Rana pipiens	Northern Leopard Frog	E	SP	V	V	V	CT	81
Rana pipiens	Northern Leopard Frog	L	SP, SU	DN	DN, T [mt]	DN	D [pf]	82
Rana septentrionalis	Mink Frog	A	SP, SU	A [a]	A [a]	A [a]	A [d]	86
Rana septentrionalis	Mink Frog	E	SP	V	V	V	V [ce]	84
Rana septentrionalis	Mink Frog	L	SP, SU	V	V, T [mt]	V	V, T [mt]	85
Rana sphenocephala	Southern Leopard Frog	A	SP, SU	V [r]	D [pf]	D [pf]	MR, D [pf]	89
Rana sphenocephala	Southern Leopard Frog	E	SP	V	V	V	CT	87
Rana sphenocephala	Southern Leopard Frog	L	SP, SU	DN	DN, T [mt]	DN	T [mt]	88
Rana sylvatica	Wood Frog	A	SP, SU	C	D [pf]	C	N/A	92
Rana sylvatica	Wood Frog	E	SP	V	V	V	CT	90
Rana sylvatica	Wood Frog	L	SP, SU	DN	DN, T [mt]	DN	T [mt]	91
Scaphiopus holbrookii	Eastern Spadefoot	BA	SP, SU	A [a]	A [r]	A [a]	D [pf]	95
Scaphiopus holbrookii	Eastern Spadefoot	E	SP, SU	V	V	V	CT	93
Scaphiopus holbrookii	Eastern Spadefoot	L	SP, SU	V, DN	V, DN	V, DN	T [mt]	94
Spea bombifrons	Plains Spadefoot	BA	SP, SU	A [a]	A [r]	A [a]	A [d]	97
Spea bombifrons	Plains Spadefoot	L	SP, SU	DN, SN	T [mt]	T [mt]	T [mt]	96

SALAMANDERS

SOUTHEAST REGION

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Ambystoma annulatum	Ringed Salamander	A	N/A	N/A	N/A	N/A	N/A	
Ambystoma annulatum	Ringed Salamander	BA	AU	V, V [r]	D [pf]	V	D [pf]	1
Ambystoma annulatum	Ringed Salamander	E	AU	V	V	V	CT	
Ambystoma annulatum	Ringed Salamander	L	WI, SP	DN	DN, T [mt]	DN	DN, T [mt]	
Ambystoma barbouri	Streamside Salamander	A	N/A	N/A	N/A	N/A	N/A	2
Ambystoma barbouri	Streamside Salamander	BA	AU, WI	V, T [mt]	D [pf]	T [mt]	D [pf]	3
Ambystoma barbouri	Streamside Salamander	E	AU, WI	C	N/A	N/A	N/A	
Ambystoma barbouri	Streamside Salamander	L	WI, SP	DN	T [mt]	T [mt]	T [mt]	
Ambystoma bishopi	Reticulated Flatwoods Salamander	A	YR	N/A	N/A	N/A	N/A	
Ambystoma bishopi	Reticulated Flatwoods Salamander	BA	AU, WI	C	D	D	D [pf]	153
Ambystoma bishopi	Reticulated Flatwoods Salamander	E	AU	N/A	N/A	N/A	N/A	4
Ambystoma bishopi	Reticulated Flatwoods Salamander	L	SP	DN	DN, T [mt]	DN, T [mt]	N/A	5
Ambystoma cingulatum	Frosted Flatwoods Salamander	A	YR	N/A	N/A	N/A	N/A	
Ambystoma cingulatum	Frosted Flatwoods Salamander	BA	AU, WI	D	D	D	D	6
Ambystoma cingulatum	Frosted Flatwoods Salamander	E	AU	N/A	N/A	N/A	N/A	4
Ambystoma cingulatum	Frosted Flatwoods Salamander	L	WI, SP	DN	DN, T [mt]	DN, T [mt]	N/A	5
Ambystoma jeffersonianum	Jefferson Salamander	A	YR	N/A	N/A	N/A	N/A	
Ambystoma jeffersonianum	Jefferson Salamander	BA	WI	V	D [pf]	V	D [pf]	
Ambystoma jeffersonianum	Jefferson Salamander	E	WI	V	V	V	V	
Ambystoma jeffersonianum	Jefferson Salamander	L	SP	DN	DN, T [mt]	DN, T [mt]	T [mt]	
Ambystoma mabeei	Mabee's Salamander	A	YR	N/A	N/A	N/A	N/A	
Ambystoma mabeei	Mabee's Salamander	BA	WI	V	D [pf]	V	D [pf]	
Ambystoma mabeei	Mabee's Salamander	E	WI	N/A	N/A	N/A	N/A	
Ambystoma mabeei	Mabee's Salamander	L	SP	DN	DN, T [mt]	DN, T [mt]	D [pf]	
Ambystoma maculatum	Spotted Salamander	A	YR	C	C [w]	N/A	N/A	
Ambystoma maculatum	Spotted Salamander	BA	WI	V	D [pf]	V	D [pf]	
Ambystoma maculatum	Spotted Salamander	E	WI	V	V	V	CT	
Ambystoma maculatum	Spotted Salamander	L	SP	DN	DN, T [mt]	DN, T [mt]	D [pf]	
Ambystoma opacum	Marbled Salamander	A	YR	C, V [r]	C [w]	C, V [r]	N/A	
Ambystoma opacum	Marbled Salamander	BA	AU	C	D [pf]	C	D [pf]	
Ambystoma opacum	Marbled Salamander	E	AU, WI	C	C	C	CT	
Ambystoma opacum	Marbled Salamander	L	WI, SP	DN	DN, T [mt]	DN, T [mt]	D [pf]	
Ambystoma talpoideum	Mole Salamander	A	YR	C	C [w]	N/A	N/A	
Ambystoma talpoideum	Mole Salamander	BA	AU	C	D [pf]	C	D [pf]	
Ambystoma talpoideum	Mole Salamander	E	AU, WI	V	V	V	CT	7
Ambystoma talpoideum	Mole Salamander	L	YR	DN	DN, T [mt]	DN, T [mt]	D [pf]	8
Ambystoma talpoideum	Mole Salamander	PA	YR	DN, T [mt]	DN, T [mt]	DN, T [mt]	T [mt]	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Ambystoma texanum	Small-mouthed Salamander	A	N/A	N/A	N/A	N/A	N/A	10
Ambystoma texanum	Small-mouthed Salamander	BA	WI, SP	V, C, T mt	D pf	T mt	D pf	11
Ambystoma texanum	Small-mouthed Salamander	E	WI, SP	V	V	N/A	N/A	9
Ambystoma texanum	Small-mouthed Salamander	L	SP	DN, T mt	T mt	T mt	T mt	
Ambystoma tigrinum	Tiger Salamander	A	YR	V b	N/A	N/A	N/A	12
Ambystoma tigrinum	Tiger Salamander	BA	WI	V	D pf	V	D pf	
Ambystoma tigrinum	Tiger Salamander	E	SP	V	V	V	CT	
Ambystoma tigrinum	Tiger Salamander	L	SP, early SU	V, DN	DN, T mt	DN, T mt	D pf	
Amphiuma means	Two-toed Amphiuma	A, L	YR	T mt, T gd	T fy	T mt	T mt	13
Amphiuma pholeter	One-toed Amphiuma	A, L	YR	T mt	T fy	T mt	T mt	27
Amphiuma tridactylum	Three-toed Amphiuma	A, L	YR	T mt	T fy	T mt	T mt	
Aneides aeneus	Green Salamander	A, J	YR	V	V	V	MR	154
Aneides aeneus	Green Salamander	E	SP, SU	V	V	V	F	
Cryptobranchus alleganiensis	Hellbender	A	YR	C r, V vb	C r, V vb	C r	MR	
Cryptobranchus alleganiensis	Hellbender	E	AU	C r	C r, V s	C r	N/A	
Cryptobranchus alleganiensis	Hellbender	L	YR	C r	C r	C r	N/A	
Desmognathus abditus	Cumberland Dusky Salamander	A	YR	V, C	V, C	V, C	V ce	
Desmognathus abditus	Cumberland Dusky Salamander	L	YR	V, C	V, DN, C, T lb	V, DN, C	V ce, T lb	
Desmognathus aeneus	Seepage Salamander	A	YR	C	V	C	V ce	
Desmognathus aeneus	Seepage Salamander	L	YR	C	DN, T lb	C	T lb	
Desmognathus apalachicolae	Apalachicola Dusky Salamander	A	YR	C	C w	C	C	
Desmognathus apalachicolae	Apalachicola Dusky Salamander	L	YR	C	C	C	T lb	
Desmognathus auriculatus	Southern Dusky Salamander	A	YR	C	C	C	C w	
Desmognathus auriculatus	Southern Dusky Salamander	L	YR	C	C	C	T lb	
Desmognathus brimleyorum	Ouachita Dusky Salamander	A	YR	C	C	C	C w	
Desmognathus brimleyorum	Ouachita Dusky Salamander	L	YR	C	C	C	T lb	
Desmognathus carolinensis	Carolina Mountain Dusky Salamander	A	YR	C	C w, D pf	C	C w	
Desmognathus carolinensis	Carolina Mountain Dusky Salamander	L	YR	C	C	C	T lb	
Desmognathus conanti	Spotted Dusky Salamander	A	YR	C	C w	C	C w	
Desmognathus conanti	Spotted Dusky Salamander	L	YR	C	C	C	T lb	
Desmognathus folkertsi	Dwarf Black-bellied Salamander	A	YR	C r	C r	C r	C r	
Desmognathus folkertsi	Dwarf Black-bellied Salamander	L	YR	C r	C r	C r	C r	
Desmognathus fuscus	Northern Dusky Salamander	A	YR	C	C w	C	C w	
Desmognathus fuscus	Northern Dusky Salamander	L	YR	C	C	C	T lb	
Desmognathus imitator	Imitator Salamander	A	YR	C	V	C	C ce	
Desmognathus imitator	Imitator Salamander	L	YR	C	DN, T lb	C	T lb	
Desmognathus marmoratus	Shovel-nosed Salamander	A	YR	C r	C r	C r	C r	
Desmognathus marmoratus	Shovel-nosed Salamander	L	YR	C r	C r	C r	C r	
Desmognathus monticola	Seal Salamander	A	YR	C	V	V	V ce	
Desmognathus monticola	Seal Salamander	L	YR	C	DN, T lb	C	T lb	
Desmognathus ochrophaeus	Allegheny Mountain Dusky Salamander	A	YR	C	D pf	C	D pf	14
Desmognathus ochrophaeus	Allegheny Mountain Dusky Salamander	L	YR	C	DN, T lb	C	T lb	

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Desmognathus ocoee	Ocoee Salamander	A	YR	C	V	C	V [ce]	
Desmognathus ocoee	Ocoee Salamander	L	YR	C	DN, T [lb]	C	T [lb]	
Desmognathus orestes	Blue Ridge Dusky Salamander	A	YR	C	C [w]	C	C [w]	
Desmognathus orestes	Blue Ridge Dusky Salamander	L	YR	C	C	C	T [lb]	
Desmognathus quadramaculatus	Black-bellied Salamander	A	YR	C [r]	C [r]	C [r]	C [r]	
Desmognathus quadramaculatus	Black-bellied Salamander	L	YR	C [r]	C [r]	C [r]	C [r]	
Desmognathus santeetlah	Santeetlah Dusky Salamander	A	YR	C	C [w]	C	C [w]	
Desmognathus santeetlah	Santeetlah Dusky Salamander	L	YR	C	C	C	T [lb]	
Desmognathus welteri	Black Mountain Salamander	A	YR	C [r]	V	V	V [ce]	
Desmognathus welteri	Black Mountain Salamander	L	YR	C	DN, T [lb]	C	T [lb]	
Desmognathus wrighti	Pygmy Salamander	A	YR	C	D [pf]	C	D [pf]	
Desmognathus wrighti	Pygmy Salamander	L	YR	C	DN, T [lb]	C	T [lb]	
Eurycea aquatica	Brown-backed Salamander	A	WI, SP	C, C [r]	C, C [r]	C, C [r]	C, C [r]	155
Eurycea aquatica	Brown-backed Salamander	L	YR	DN	DN	DN	DN	155
Eurycea chamberlaini	Chamberlain's Dwarf Salamander	A	YR	C	C	C	C [w]	
Eurycea chamberlaini	Chamberlain's Dwarf Salamander	L	SP	T [lb]	T [lb]	T [lb]	T [lb]	
Eurycea cirrigera	Southern Two-lined Salamander	A	YR	C	C [w]	C	C [w]	
Eurycea cirrigera	Southern Two-lined Salamander	L	YR	C	C	C	T [lb]	
Eurycea guttolineata	Three-lined Salamander	A	SP, SU	V	C [w]	C [w]	C [w]	
Eurycea guttolineata	Three-lined Salamander	L	SU	C	C	C	T [lb]	
Eurycea junaluska	Junaluska Salamander	A	YR	C	C [w]	C	C [w]	
Eurycea junaluska	Junaluska Salamander	L	YR	C	C	C	T [lb]	
Eurycea longicauda	Long-tailed Salamander	A	SP, SU	C, V	C [w], V	C [w], V	C [w], V	15
Eurycea longicauda	Long-tailed Salamander	L	SU	C	C	C	T [lb]	
Eurycea lucifuga	Cave Salamander	A	SU	V	V	V	V [ce]	16
Eurycea lucifuga	Cave Salamander	L	SU	C	C	C	T [lb]	
Eurycea multiplicata	Many-ribbed Salamander	A	YR	C [r]	C [r]	C [r]	V [ce]	
Eurycea multiplicata	Many-ribbed Salamander	L	YR	C [r]	C [r]	C [r]	V [ce]	17
Eurycea quadridigitata	Dwarf Salamander	A	YR	C	C	C	C [w]	
Eurycea quadridigitata	Dwarf Salamander	L	SP	T [lb]	T [lb]	T [lb]	T [lb]	
Eurycea tynerensis	Oklahoma Salamander	A	YR	C	C	C	T [lb]	
Eurycea tynerensis	Oklahoma Salamander	L	SU	C	C	C	T [lb]	
Eurycea wilderae	Blue Ridge Two-lined Salamander	A	SP, SU, AU	V, C [w]	D [pf]	C [w]	C [w]	
Eurycea wilderae	Blue Ridge Two-lined Salamander	L	YR	C	C	C	T [lb]	
Gyrinophilus gulolineatus	Berry Cave Salamander	A, L	YR	V, DN, C	V, DN, C, T [mt]	V, DN, C, T [mt]	V [ce], T [mt], MR	
Gyrinophilus gulolineatus	Berry Cave Salamander	E	AU, WI	V, C [r]	N/A	V, C [r]	N/A	
Gyrinophilus palleucus	Tennessee Cave Salamander	A, L	YR	V, DN, C	V, DN, C, T [mt]	V, DN, C, T [mt]	V [ce], T [mt], MR	
Gyrinophilus palleucus	Tennessee Cave Salamander	E	AU, WI	V, C [r]	N/A	C [r]	N/A	18

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Gyrinophilus porphyriticus	Spring Salamander	A	SP	C [r]	C [r]	C [r]	V [ce]	
Gyrinophilus porphyriticus	Spring Salamander	L	YR	C [r]	C [r]	C [r]	V [ce]	
Haideotriton wallacei	Georgia Blind Salamander	A, L	YR	V, DN	V, DN	V, DN	N/A	
Hemidactylium scutatum	Four-toed Salamander	A	SP	C	C	C	MR	
Hemidactylium scutatum	Four-toed Salamander	BA	YR	V	V	V	MR	
Hemidactylium scutatum	Four-toed Salamander	E	SP	V	V	V	CT	19
Hemidactylium scutatum	Four-toed Salamander	L	WI (Florida), SP	DN	DN	DN	DN	20
Necturus alabamensis	Black Warrior Waterdog	A, L	AU, WI, SP	DN, T [mt], T [gd]	DN, T [mt], T [gd]	T [lb], T [mt]	T [lb], T [mt]	21
Necturus beyeri	Gulf Coast Waterdog	A, L	YR	DN	DN	DN	DN	21
Necturus lewisi	Neuse River Waterdog	A, L	YR	DN	DN	DN	DN	
Necturus maculosus	Mudpuppy	A, L	YR	C [r]	T [hl]	C [r]	MR	
Necturus punctatus	Dwarf Waterdog	A, L	YR	DN	DN	DN	DN	
Notophthalmus perstriatus	Striped Newt	A	YR	C	N/A	N/A	N/A	
Notophthalmus perstriatus	Striped Newt	BA	WI	V	D [pf]	V	D [pf]	
Notophthalmus perstriatus	Striped Newt	L	SP	DN	DN, T [mt]	DN, T [mt]	T [mt]	
Notophthalmus viridescens	Eastern Newt	A	SU	DN	T [mt]	T [mt]	T [mt]	22
Notophthalmus viridescens	Eastern Newt	BA	WI	DN	T [mt]	DN, T [mt]	T [mt]	
Notophthalmus viridescens	Eastern Newt	Eft	YR	C	C [w]	C [w]	C [w]	
Notophthalmus viridescens	Eastern Newt	L	SP	DN	DN	DN		
Phaeognathus hubrichti	Red Hills Salamander	A, J	SP, SU, AU	V [b]	V [b]	V [b]	V [ce], V [b]	
Plethodon ainsworthi	Bay Springs Salamander	A, J	?	?	?	?	?	156
Plethodon amplus	Blue Ridge Gray-cheeked Salamander	A, J, E	SP, SU, AU	V, C	V, C	V, C, C[w]	V[ce], V, C[w], D[pf]	23, 26
Plethodon angusticlavius	Ozark Zigzag Salamander	A, J	SP	C	C [w]	C [w]	C [w]	
Plethodon aureolus	Tellico Salamander	A, J	SP	C	C [w]	C [w]	C [w]	
Plethodon caddoensis	Caddo Mountain Salamander	A, J	SP	C	C [w]	C [w]	C [w]	
Plethodon chattahoochee	Chattahoochee Slimy Salamander	A, J	SP, AU	C, V	V, C[w,]	V, C[w]	V, C[w]	
Plethodon cheoah	Cheoah Bald Salamander	A, J, E	SP, SU, AU	V, C	V, C	V, C, C[w]	V[ce], V, C[w], D[pf]	
Plethodon cinereus	Eastern Red-backed Salamander	A, J	SP	C	C [w]	C [w]	C [w]	
Plethodon dorsalis	Northern Zigzag Salamander	A, J	SP	C	C [w]	C [w]	C [w]	
Plethodon electromorphus	Northern Ravine Salamander	A, J	SP	C	C [w]	C [w]	C [w]	
Plethodon fourchensis	Fourche Mountain Salamander	A, J	SP	C	C [w]	C [w]	C [w]	
Plethodon glutinosus	Northern Slimy Salamander	A, J	SP, AU	C, C [r]	C [w]	C [w]	C [w]	
Plethodon grobmani	Southeastern Slimy Salamander	A, J	SP, AU	C, V	C [w]	C [w]	C [w]	60
Plethodon jordani	Red-cheeked Salamander	A, J	SP	C	C [w]	C [w]	C [w]	
Plethodon kentucki	Cumberland Plateau Salamander	A, J	SP	C	C [w]	C [w]	C [w]	
Plethodon kisatchie	Louisiana Slimy Salamander	A, J	SP, AU	C	C[w]	C[w]	C[w]	
Plethodon meridianus	South Mountain Gray-cheeked Salamander	A, J, E	SP, SU, AU	V, C	V, C	V, C, C[w]	V[ce], V, C[w], D[pf]	23, 26
Plethodon metcalfi	Southern Gray-cheeked Salamander	A, J, E	SP, SU, AU	C, V	V, C[w]	V, C, C[w]	V[ce], V, D[pf], C[w]	23, 26
Plethodon mississippi	Mississippi Slimy Salamander	A, J	SP, AU	C	C[w]	C[w]	C[w]	
Plethodon ocmulgee	Ocmulgee Slimy Salamander	A, J	SP, AU	C	C[w]	C[w]	C[w]	
Plethodon ouachitae	Rich Mountain Salamander	A, J	SP	C	C [w]	C [w]	C [w]	

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Plethodon petraeus	Pigeon Mountain Salamander	A, J	SP, AU	V, C [r]	V	V, C [r]	V [ce]	15, 16, 23, 60
Plethodon richmondi	Southern Ravine Salamander	A, J	SP	C, V	C, V	C, V	C, V[ce]	25
Plethodon savannah	Savannah Slimy Salamander	A, J	SP	C	C[w]	C[w]	C[w]	
Plethodon serratus	Southern Red-backed Salamander	A, J	SP	C	C	C	C	
Plethodon shermani	Red-legged Salamander	A, J	SP, SU, AU	V, C	V, C[w]	V, C[w], C	V[ce], V, C[w]	26
Plethodon teyahalee	Southern Appalachian Salamander	A, J	SP	C	C	C	C	
Plethodon variolatus	South Carolina Slimy Salamander	A, J	SP	C	C[w]	C[w]	C[w]	
Plethodon ventralis	Southern Zigzag Salamander	A, J	SP	C, V	C, V	V, C[w]	V[ce], C[w]	
Plethodon websteri	Webster's Salamander	A, J	SP	C	C	C	V [ce]	24
Plethodon wehrlei	Wehrle's Salamander	A, J	SP, AU	C, V	C, V	C, V	C, V[ce]	25
Plethodon welleri	Weller's Salamander	A, J	SP, AU	C, V	C, V	C, V	C, V[ce]	
Plethodon yonahlossee	Yonahlossee Salamander	A, J	SP	V [b]	V [ce]	V [b]	V [ce]	26
Pseudobranchus axanthus	Southern Dwarf Siren	A, L	SP, SU	DN	T [mt]	T [mt]	T [mt]	
Pseudobranchus striatus	Northern Dwarf Siren	A, L	SP, SU	DN	T [mt]	T [mt]	T [mt]	
Pseudotriton montanus	Mud Salamander	A	SP, SU, AU	C	C [w]	C	C	
Pseudotriton montanus	Mud Salamander	L	SP, SU, AU	DN	DN	DN	T [lb]	
Pseudotriton ruber	Red Salamander	A	SP, SU, AU	C	C [w]	C	C	
Pseudotriton ruber	Red Salamander	L	SP, SU, AU	DN	DN	DN	T [lb]	
Siren intermedia	Lesser Siren	A	SP, SU	T [cr], T [mt]	T [cr], T [fy]	T [cr], T [mt]	T [cr], T [mt]	
Siren lacertina	Greater Siren	A	SP, SU	T [cr], T [mt]	T [cr], T [fy]	T [cr], T [mt]	T [cr], T [mt]	
Stereochilus marginatus	Many-lined Salamander	A, L	SP,SU	DN	DN	DN	DN	
Typhlotriton spelaeus	Grotto Salamander	A	YR	V	V	V	V [ce]	
Typhlotriton spelaeus	Grotto Salamander	L	YR	C [r]	C [r]	C [r]	V [ce], C [r]	
Urspelerpes brucei	Patch-nosed Salamander	A, L	YR	C [r], DN, V	T [lb]	T [lb], C [r]	T [lb], V [ce]	157
NORTHEAST REGION								
Ambystoma barbouri	Streamside Salamander	A	AU, WI	N/A	V	N/A	N/A	28
Ambystoma barbouri	Streamside Salamander	BA	AU, WI	V, T [mt]	D [pf], T [mt]	T [mt]	D [pf]	29
Ambystoma barbouri	Streamside Salamander	E	WI, SP	C [r]	C [r]	N/A	N/A	
Ambystoma barbouri	Streamside Salamander	L	SP	DN, T [mt]	T [mt]	T [mt]	T [mt]	
Ambystoma jeffersonianum	Jefferson Salamander	A	YR	V	C	N/A	N/A	30
Ambystoma jeffersonianum	Jefferson Salamander	BA	WI	V	D [pf]	V	D [pf]	
Ambystoma jeffersonianum	Jefferson Salamander	E	WI	V	V	V	CT	
Ambystoma jeffersonianum	Jefferson Salamander	L	SP	DN	DN, T [mt]	DN, T [mt]	T [mt]	
Ambystoma laterale	Blue-spotted Salamander	A	YR	C	N/A	N/A	N/A	
Ambystoma laterale	Blue-spotted Salamander	BA	WI, SP	N/A	DN, D [pf]	DN, D [pf]	D [pf]	31
Ambystoma laterale	Blue-spotted Salamander	E	WI	V	V	V	CT	
Ambystoma laterale	Blue-spotted Salamander	L	SP	DN	DN, T [mt]	DN, T [mt]	T [mt]	
Ambystoma mabeei	Mabee's Salamander	A	YR	N/A	N/A	N/A	N/A	34
Ambystoma mabeei	Mabee's Salamander	BA	WI	V, C	D [pf]	V	D [pf]	
Ambystoma mabeei	Mabee's Salamander	E	WI	V	N/A	V	N/A	32
Ambystoma mabeei	Mabee's Salamander	L	SP	DN	DN, T [mt]	DN, T [mt]	D [pf]	33

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Ambystoma maculatum	Spotted Salamander	A	YR	C	C	N/A	N/A	35
Ambystoma maculatum	Spotted Salamander	BA	WI	V	D pf	V	D pf	
Ambystoma maculatum	Spotted Salamander	E	WI	V	V	V	CT	
Ambystoma maculatum	Spotted Salamander	L	SP	DN	DN, T mt	DN, T mt	D pf	
Ambystoma opacum	Marbled Salamander	A	YR	C, V r	C w	C, V r	MR	
Ambystoma opacum	Marbled Salamander	BA	AU	C	D pf	C	D pf	38
Ambystoma opacum	Marbled Salamander	E	AU, WI	C	C	C	CT	36
Ambystoma opacum	Marbled Salamander	L	WI, SP	DN	DN, T mt	DN, T mt	D pf	37
Ambystoma talpoideum	Mole Salamander	A	YR	C	C w	N/A	N/A	
Ambystoma talpoideum	Mole Salamander	BA	AU	C	D pf	C	D pf	
Ambystoma talpoideum	Mole Salamander	E	WI, SP	V	V	V	N/A	39
Ambystoma talpoideum	Mole Salamander	L	YR	DN	DN, T mt	DN, T mt	D pf	40
Ambystoma talpoideum	Mole Salamander	PA	YR	DN, T mt	DN, T mt	DN, T mt	T mt	
Ambystoma texanum	Small-mouthed Salamander	A	N/A	N/A	N/A	N/A	N/A	42
Ambystoma texanum	Small-mouthed Salamander	BA	WI, SP	V, T mt	D pf	T mt	D pf	43
Ambystoma texanum	Small-mouthed Salamander	E	WI, SP	V	V	N/A	N/A	41
Ambystoma texanum	Small-mouthed Salamander	L	SP	DN, T mt	T mt	T mt	T mt	
Ambystoma tigrinum	Tiger Salamander	A	YR	N/A	N/A	N/A	N/A	
Ambystoma tigrinum	Tiger Salamander	BA	WI	V	D pf	V	D pf	51
Ambystoma tigrinum	Tiger Salamander	E	SP	V	V	V	CT	
Ambystoma tigrinum	Tiger Salamander	L	SP, early SU	V, DN	DN, T mt	DN, T mt	D pf	
Amphiuma means	Two-toed Amphiuma	A	SP, SU	T cr, T mt	T fy, T cr	T cr, T mt	T mt, MR	44
Aneides aeneus	Green Salamander	A, J	SP, AU	V	V	V	MR	45
Cryptobranchus alleganiensis	Hellbender	A	YR	C, V vb	C, V vb, V s	C	MR	46
Cryptobranchus alleganiensis	Hellbender	E	AU	C	C	C	N/A	
Cryptobranchus alleganiensis	Hellbender	L	YR	C	C, V s	C	N/A	
Desmognathus auriculatus	Southern Dusky Salamander	A	SP, SU, AU	C	C	C	N/A	
Desmognathus auriculatus	Southern Dusky Salamander	L	SP, SU	V	V	V	V ce	
Desmognathus fuscus	Northern Dusky Salamander	A	SP, SU, AU	C	C	C	MR, R	
Desmognathus fuscus	Northern Dusky Salamander	L	SU	C	C	C	MR, R	
Desmognathus marmoratus	Shovel-nosed Salamander	A	YR	C, DN	C, DN	C, DN	C, DN	
Desmognathus marmoratus	Shovel-nosed Salamander	L	YR	C, DN	C, DN	C, DN	C, DN	
Desmognathus monticola	Seal Salamander	A	YR	C	C	C	MR, R	
Desmognathus monticola	Seal Salamander	L	Jun-Sept	C	C	C	MR, R	
Desmognathus ochrophaeus	Allegheny Mountain Dusky Salamander	A	SP, SU, AU	C	D pf	N/A	N/A	48
Desmognathus ochrophaeus	Allegheny Mountain Dusky Salamander	L	SU, AU	C	C	N/A	N/A	47
Desmognathus orestes	Blue Ridge Dusky Salamander	A	SP, SU, AU	C	C	C	C w	50
Desmognathus orestes	Blue Ridge Dusky Salamander	L	SU, AU	C	C	C	T lb	49
Desmognathus quadramaculatus	Black-bellied Salamander	A	SP, SU	C r	C r	C r	C r	51
Desmognathus quadramaculatus	Black-bellied Salamander	L	SP, SU, AU	C r	C r	C r	C r	
Desmognathus welteri	Black Mountain Salamander	A	SP, SU	DN, C r	DN, C r	DN, C r	V ce	
Desmognathus welteri	Black Mountain Salamander	L	SP, SU	DN	DN	DN	T lb	

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Desmognathus wrighti	Pygmy Salamander	A	SP, SU	C	C	C	D [pf]	53
Desmognathus wrighti	Pygmy Salamander	L	SP, SU	DN, C [r]	DN, C [r]	DN, C [r]	T [lb]	52
Eurycea bislineata	Northern Two-lined Salamander	A	SP, AU	C	C	C	MR R	54
Eurycea bislineata	Northern Two-lined Salamander	L	YR	C, DN	C, DN	C, DN	MR, R	
Eurycea cirrigera	Southern Two-lined Salamander	A	SP, SU	C	C	C	C [w]	
Eurycea cirrigera	Southern Two-lined Salamander	L	SP, SU	C	C	C	T [lb]	
Eurycea guttolineata	Three-lined Salamander	A	AU	C	C	C	MR, R	
Eurycea guttolineata	Three-lined Salamander	L	YR	C	C	C	MR, R	
Eurycea longicauda	Long-tailed Salamander	A	SP, SU	C, V	C, V	C, V	MR, R	55
Eurycea longicauda	Long-tailed Salamander	L	SU	C	C	C	MR, R	
Eurycea lucifuga	Cave Salamander	A	SP, SU, AU	V	V	V	V [ce]	56
Eurycea lucifuga	Cave Salamander	L	SU	C	C	C	T [lb]	
Eurycea wilderae	Blue Ridge Two-lined Salamander	A	SP, SU, AU	V	V	V	V [ce]	58
Eurycea wilderae	Blue Ridge Two-lined Salamander	L	WI	V	V	V	V [ce]	57
Gyrinophilus porphyriticus	Spring Salamander	A	SP, SU, AU	V	V	V	N/A	59
Gyrinophilus porphyriticus	Spring Salamander	L	YR	C, DN	C, DN	C, DN	MR, R	
Gyrinophilus subterraneus	West Virginia Spring Salamander	A	N/A	V	N/A	N/A	N/A	60
Hemidactylium scutatum	Four-toed Salamander	A	SP,SU	C	D [pf]	C	D [pf]	63
Hemidactylium scutatum	Four-toed Salamander	BA	SP	C	C	C	C	
Hemidactylium scutatum	Four-toed Salamander	E	SP	V	V	V	CT	61
Hemidactylium scutatum	Four-toed Salamander	L	WI, SP	DN	DN	DN	DN	62
Necturus maculosus	Mudpuppy	A, L	WI, SP	C [r]	T [hl]	C [r]		64
Necturus punctatus	Dwarf Waterdog	A, L	WI, SP	DN	DN	DN	DN	65
Notophthalmus viridescens	Eastern Newt	A	YR	V, DN, T [mt]	V, DN, T [mt]	V, DN, T [mt]	V [ce], DN, T [mt]	66
Notophthalmus viridescens	Eastern Newt	BA	WI, SP, SU	V, DN	V, DN	V, DN	V [ce], DN	
Notophthalmus viridescens	Eastern Newt	Eft	SP, SU, AU	V, C	V, D	V, C	V [ce]	
Notophthalmus viridescens	Eastern Newt	L	SP, SU	V, DN	V, DN	V, DN	N/A	
Plethodon chlorobryonis	Atlantic Coast Slimy Salamander	A, J	SP, SU, AU	C	C	C	N/A	67
Plethodon cinereus	Eastern Red-backed Salamander	A, J	SP, SU, AU	C	C	C	C	68
Plethodon cylindraceus	White-spotted Slimy Salamander	A, J	SP, SU, AU	C	C	C	N/A	68
Plethodon electromorphus	Northern Ravine Salamander	A, J	WI, SP, AU	C	C	C	N/A	68
Plethodon glutinosus	Northern Slimy Salamander	A, J	SP, SU, AU	C	C	C	N/A	68
Plethodon hoffmani	Valley and Ridge Salamander	A, J	SP, AU	V	V	V	N/A	68
Plethodon hubricti	Peaks of Otter Salamander	A, J	SP, SU, AU	V	V	V	N/A	68
Plethodon kentucki	Cumberland Plateau Salamander	A, J	SP, AU, WI	C	C	C	N/A	68
Plethodon montanus	Northern Gray-cheeked Salamander	A, J	SP, SU, AU	C	C	C	N/A	69
Plethodon nettingi	Cheat Mountain Salamander	A, J	SP, SU, AU	C	C	C	N/A	70
Plethodon punctatus	Cow Knob Salamander	A, J	SP, SU, AU	C	C	C	N/A	71
Plethodon richmondi	Southern Ravine Salamander	A, J	SP, AU	C	C	C	N/A	68
Plethodon shenandoah	Shenandoah Salamander	A	SP, SU, AU	C	C	C	C	72
Plethodon sherando	Big Levels Salamander	A	SP, SU, AU	C	C	C	N/A	
Plethodon ventralis	Southern Zigzag Salamander	A, J	SP, SU, AU	C	C	C	C	73
Plethodon virginia	Shenandoah Mountain Salamander	A, J	SP, SU, AU	C	C	C	N/A	71
Plethodon wehrlei	Wehrle's Salamander	A, J	SP, SU, AU	C	C	C	C	74

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Plethodon welleri	Weller's Salamander	A, J	SP, SU, AU	C	C	C	C	75
Plethodon yonahlossee	Yonahlossee Salamander	A, J	SP, SU, AU	C	C	C	N/A	
Pseudotriton montanus	Mud Salamander	A	SP, SU, AU	C	C	C	C	
Pseudotriton montanus	Mud Salamander	L	SP, SU, AU	C	C	C	C	76
Pseudotriton ruber	Red Salamander	A	SP, SU, AU	C	C	C	MR, R	77
Pseudotriton ruber	Red Salamander	L	SP, SU, AU	DN, C r	DN, C	C	MR, R	
Siren intermedia	Lesser Siren	A	SP, SU	T mt, T cr	T cr, T fy	T cr, T mt	MR, T cr, T mt	78
Siren lacertina	Greater Siren	A	SP, SU	T mt, T cr	T cr, T fy	T cr, T mt	MR, T cr, T mt	
Stereochilus marginatus	Many-lined Salamander	A	SP, SU, AU	C	C	C	N/A	79
Stereochilus marginatus	Many-lined Salamander	E	SP	C	N/A	C	N/A	
SOUTHWEST REGION								
Ambystoma californiense	California Tiger Salamander	All	YR	DN, T h, SN, V r	DN, T h, SN, V r	DN, T h, SN, V r	DN, T h, SN, V r	
Ambystoma gracile	Northwestern Salamander	All	YR	DN, T h, SN, V r	DN, T h, SN, V r	DN, T h, SN, V r	DN, T h, SN, V r	
Ambystoma macrodactylum	Long-toed Salamander	All	YR	DN, T h, SN, V r	DN, T h, SN, V r	DN, T h, SN, V r	DN, T h, SN, V r	
Ambystoma maculatum	Spotted Salamander	All	YR	DN, T h, SN, V r	DN, T h, SN, V r	DN, T h, SN, V r	DN, T h, SN, V r	
Ambystoma mavortium	Barred Tiger Salamander	A	SP, SU, AU, YR	D pf, V, V r	D pf, V, V r	D pf, V, V r	D pf, V, V r	
Ambystoma mavortium	Barred Tiger Salamander	L	YR	V, DN, SN	V, DN, SN	V, DN, SN	DN, SN	
Ambystoma opacum	Marbled Salamander	A	YR	C, V r	C w	C, V r	N/A	
Ambystoma opacum	Marbled Salamander	BA	AU	C	D pf	C	D pf	82
Ambystoma opacum	Marbled Salamander	E	AU, WI	C	C	C	CT	80
Ambystoma opacum	Marbled Salamander	L	WI, SP	DN	DN, T mt	DN, T mt	D pf	81
Ambystoma talpoideum	Mole Salamander	A	YR	C	C w	N/A	N/A	
Ambystoma talpoideum	Mole Salamander	BA	AU	C	D pf	C	D pf	
Ambystoma talpoideum	Mole Salamander	E	WI, SP	C	C	C	CT	83
Ambystoma talpoideum	Mole Salamander	L	YR	DN	DN, T mt	DN, T mt	D pf	84
Ambystoma talpoideum	Mole Salamander	PA	YR	DN, T mt	DN, T mt	DN, T mt	T mt	
Ambystoma texanum	Small-mouthed Salamander	A	YR	C	C w	N/A	N/A	86
Ambystoma texanum	Small-mouthed Salamander	BA	WI, SP	V, T mt	D pf	T mt	D pf	87
Ambystoma texanum	Small-mouthed Salamander	E	AU, WI	C	C	C	N/A	85
Ambystoma texanum	Small-mouthed Salamander	L	WI, SP	DN, T mt	T mt	T mt	T mt	
Ambystoma tigrinum	Tiger Salamander	A, L	SP, SU	V	C, T mt	V	D pf	
Amphiuma tridactylum	Three-toed Amphiuma	A, L	YR	T mt	T fy	T mt	T mt	
Aneides ferreus	Clouded Salamander	J, A	SP, SU	V	V ce, C	V, C	V ce, C	88
Aneides flavipunctatus	Black Salamander	J, A	SP, SU	V	V ce, C	V	V ce, C	89
Aneides hardii	Sacramento Mountains Salamander	A	SU	V	V ce, C	V	V ce, C, MR	90
Aneides lugubris	Arboreal Salamander	J, A	SP, AU, WI	V	V ce, C	V, C	V ce, C	91
Aneides vagrans	Wandering Salamander	J, A	SP, SU	V	V ce, C	V, C	V ce, C	92
Batrachoseps attenuatus	California Slender Salamander	J, A	SP, SU	V	V ce, C	V, C	V ce, C	
Batrachoseps campi	Inyo Mountains Salamander	J, A	SP, SU	V	V ce, C	V, C	V ce, C	
Batrachoseps diabolicus	Hell Hollow Slender Salamander	J, A	SP, SU	V	V ce, C	V, C	V ce, C	

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Batrachoseps gabrieli	San Gabriel Mountains Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps gavilanensis	Gabilan Mountains Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps gregarius	Gregarious Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps incognitus	San Simeon Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps kawia	Sequoia Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps luciae	Santa Lucia Mountains Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps major	Garden Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps minor	Lesser Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps nigriventris	Black-bellied Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps pacificus	Channel Islands Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps regius	Kings River Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps relictus	Relictual Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps robustus	Kern Plateau Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps simatus	Kern Canyon Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Batrachoseps stebbinsi	Tehachapi Slender Salamander	J, A	SP, SU	V	V [ce], C	V, C	V [ce], C	
Desmognathus auriculatus	Southern Dusky Salamander	A	SP, SU, AU	C	C	C	MR, R	
Desmognathus auriculatus	Southern Dusky Salamander	L	SU, AU	C	C	C	MR, R	
Dicamptodon ensatus	California Giant Salamander	A, J	SP, SU	V	V	V	V	
Dicamptodon tenebrosus	Coastal Giant Salamander	A, J	SP, SU	V	V	V	V	
Ensatina eschscholtzii	Ensatina	All	SP, SU	C	C [w]	C	C [w]	
Eurycea chisholmensis	Salado Salamander	L, A	YR	C	T [lb]	C	T [mt], F, T [lb]	93
Eurycea latitans	Cascade Caverns Salamander	L, A	YR	V [s], C	T [mt]	T [mt]	T [mt], F, T [lb]	93
Eurycea nana	San Marcos Salamander	L, A	YR	V, C	T [lb]	V, C	T [mt], F, T [lb]	93
Eurycea naufragia	Georgetown Salamander	L, A	YR	V [s], C	T [mt], T [lb]	V [s], C	T [mt], F, T [lb]	93
Eurycea neotenes	Texas Salamander	L, A	YR	C, V	T [lb]	C, V	T [mt], F, T [lb]	93
Eurycea pterophila	Fern Bank Salamander	L, A	YR	C, V	T [mt], T [lb]	C,	T [mt], F, T [lb]	93
Eurycea quadridigitata	Dwarf Salamander	A	YR	C	C	C	C [w]	
Eurycea quadridigitata	Dwarf Salamander	L	SP	T [lb]	T [lb]	T [lb]	T [lb]	
Eurycea rathbuni	Texas Blind Salamander	L, A	YR	V	V	V	F	94
Eurycea robusta	Blanco Blind Salamander	A	YR	C	C	C	C [w]	95
Eurycea sosorum	Barton Springs Salamander	L, A	YR	V, C	T [lb]	V, C	T [mt], F, T [lb]	93
Eurycea tonkawae	Jollyville Plateau Salamander	L, A	YR	V, C	T [lb]	V, C	T [mt], F, T [lb]	93
Eurycea tridentifera	Comal Blind Salamander	L, A	YR	V	T [mt], V [s]	T [mt]	T [mt], F	94
Eurycea troglodytes	Valdina Farms Salamander	L, A	YR	V [s], C	T [lb], T [mt]	V [s], C	T [mt], F, T [lb]	93
Eurycea waterlooensis	Austin Blind Salamander	L, A	YR	V	V	V	F	94
Hydromantes brunus	Limestone Salamander	J, A	WI, SP	V	V [ce], C	V, C	V [ce], C	96
Hydromantes platycephalus	Mount Lyell Salamander	J, A	WI, SP	V	V [ce], C	V, C	V [ce], C	96
Hydromantes shastae	Shasta Salamander	J, A	WI, SP	V	V [ce], C	V, C	V [ce], C	96
Necturus beyeri	Gulf Coast Waterdog	A, L	YR	DN, SN	DN, SN	DN, SN	D	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Notophthalamus meridionalis	Black-spotted Newt	L, A	AU, WI, SP	SN	T mt, SN	SN	SN, T mt, F	97
Notophthalmus viridescens	Eastern Newt	A	YR	V, DN, T mt	V, DN, T mt	SN	SN, T mt, F	98
Notophthalmus viridescens	Eastern Newt	BA	WI, SP, SU	V, DN	V, DN	SN	SN, T mt, F	
Notophthalmus viridescens	Eastern Newt	Eft	SP, SU, AU	V, C	V, C	SN	SN, T mt, F	
Notophthalmus viridescens	Eastern Newt	L	SP, SU	V, DN, SN	V, DN	SN	SN, T mt, F	
Plethodon albagula	Western Slimy Salamander	A	SP, AU	C	C, V	V , C	C, MR	
Plethodon asupak	Scott Bar Salamander	J, A	WI, SP	V	V ce, C	V, C	V ce, C	96
Plethodon dunni	Dunn's Salamander	J, A	SP, AU	V	V ce, C	V, C	V ce, C	99
Plethodon elongatus	Del Norte Salamander	J, A	SP, AU	V	V ce, C	V, C	V ce, C	100
Plethodon kiamichi	Kiamichi Slimy Salamander	A	SU	V	V ce, C	V, C	V ce, C	
Plethodon neomexicanus	Jemez Mountains Salamander	A	SU	V	V ce	V	V ce, MR	101
Plethodon sequoyah	Sequoyah Slimy Salamander	A	SU	V	V ce, C	V, C	V ce, C	
Plethodon serratus	Southern Red-backed Salamander	A	SU	V	V ce, C	V, C	V ce, C	
Plethodon stormi	Siskiyou Mountains Salamander	J, A	SP, AU	V	V ce, C	V	V ce, C	100
Rhyacotriton variegatus	Southern Torrent Salamander	A	YR	V, DN	V ce, DN	V, DN	V ce, DN	102
Rhyacotriton variegatus	Southern Torrent Salamander	L	YR	V, DN	V ce, DN	V, DN	V ce, DN	102
Siren intermedia	Lesser Siren	A	SP, SU	T cr, T mt	T cr, T fy	T cr, T mt	T cr, T fy	
Taricha granulosa	Rough-skinned Newt	A	YR	V, V r	V, V r	V, V r	V, V r	104
Taricha granulosa	Rough-skinned Newt	All	YR	V, SN, T h	V, SN, T h	V, SN, T h	V, SN, T h	
Taricha granulosa	Rough-skinned Newt	J	AU	V	V ce	V	V ce	103
Taricha granulosa	Rough-skinned Newt	L	SU	V, DN	V, DN, T mt	V, DN	V, DN, T mt	
Taricha rivularis	Red-bellied Newt	A	YR	V, V r	V, V r	V, V r	V, SN, T h	
Taricha torosa	California Newt	All	YR	V, SN, T h	V, SN, T h	V, SN, T h	V ce, SN, T h	
NORTHWEST REGION								
Ambystoma californiense	California Tiger Salamander	A	SP, SU, AU	V	V, V r	V	V, V r	106
Ambystoma californiense	California Tiger Salamander	E	SP	V	V ce	V	CT	
Ambystoma californiense	California Tiger Salamander	L	SP, SU, AU	V, DN	V ce, DN, T mt	V, DN	V ce, DN, T mt	105
Ambystoma gracile	Northwestern Salamander	A	SP, AU	C, V, DN, V r	C, V ce, T mt	D pf, V, DN, T mt	D pf, V ce	109, 158
Ambystoma gracile	Northwestern Salamander	E	SP, SU	V	V ce, V s	V	CT	107
Ambystoma gracile	Northwestern Salamander	J	SP, AU	C, V, V r	C, V ce	D pf, V	D pf, V ce	108, 158
Ambystoma gracile	Northwestern Salamander	L	SP, AU (some sites SU, WI)	V, DN	V, DN, T mt, V s	V, DN, T mt	V, DN, T mt, V s	
Ambystoma macrodactylum	Long-toed Salamander	A	SP, AU	C, V, V r	C, V ce	D pf, V, V r	D pf, V ce	112
Ambystoma macrodactylum	Long-toed Salamander	E	WI, SP, SU?	V	V ce	V	CT	110
Ambystoma macrodactylum	Long-toed Salamander	J	SU, AU	C,V, V r	C, V ce	D pf, V	D pf, V ce	108
Ambystoma macrodactylum	Long-toed Salamander	L	SP, SU (some areas AU, W)	V, DN	V ce, DN, T mt	V, DN, T mt	V ce, DN, T mt	111
Ambystoma mavortium	Barred Tiger Salamander	A	SP, SU, AU	V	V ce, V r	V	V ce, V r	106
Ambystoma mavortium	Barred Tiger Salamander	E	SP	V	V ce	V	CT	
Ambystoma mavortium	Barred Tiger Salamander	L	SP, SU, AU	V, DN	V ce, DN, T mt	V, DN	V ce, DN, T mt	105
Aneides ferreus	Clouded Salamander	J, A	SP, AU,WI?	V	C, V ce	V, C	C, V ce	113

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Aneides flavipunctatus	Black Salamander	J, A	SP, AU, WI?	V	V ce	V	V ce	114
Aneides lugubris	Arboreal Salamander	J, A	SP,AU,WI	V	C, V ce	V, C	C, V ce	91
Aneides vagrans	Wandering Salamander	J, A	SP, AU, WI?	C, V	C, V ce	V, C, D pf	C, V ce, D pf	92
Batrachoseps attenuatus	California Slender Salamander	J, A	SP, AU, WI?	V	C, V ce	V, C	C, V ce	
Batrachoseps wrightorum	Oregon Slender Salamander	J, A	SP, AU, WI?	V	C, V ce	V, C	V ce, C	115
Dicamptodon aterrimus	Idaho Giant Salamander	L, J, A	YR	V	V ce, V s, V es	V	V ce, DN, V s, V es	116
Dicamptodon copei	Cope's Giant Salamander	A	YR (aquatic), SP, AU (terrestrial)	V, DN	V ce, DN, V s, V es	V, DN	V ce, DN, V s, V es	117
Dicamptodon copei	Cope's Giant Salamander	L	YR	V, DN	V ce, DN, V s, V es	V, DN	V ce, DN, V s, V es	
Dicamptodon ensatus	California Giant Salamander	A	SP, AU	V	V ce, DN, V s, V es	V	V ce, DN, V s, V es	118
Dicamptodon ensatus	California Giant Salamander	L	YR	V, DN	V ce, DN, V s, V es	V, DN	V ce, DN, V s, V es	
Dicamptodon tenebrosus	Coastal Giant Salamander	A	SP, AU, SU, WI?	C, V	C, V ce, DN, V s, V es	C, V, D pf	V ce, DN, V s, V es	116, 159
Dicamptodon tenebrosus	Coastal Giant Salamander	L	YR	V, DN	V ce, DN, V s, V es	V, DN	V ce, DN, V s, V es, V ks	
Ensatina eschscholtzii	Ensatina	J, A	SP, AU, WI?	C, V, V r	C, V ce	V, C, D pf	C, V ce, D pf	119
Hydromantes shastae	Shasta Salamander	J, A	WI, SP	V	C, V ce	V, C	C, V ce	96
Plethodon asupak	Scott Bar Salamander	J, A	WI, SP	V	C, V ce	V, C	C, V ce	96
Plethodon dunni	Dunn's Salamander	J, A	SP, AU, SU?, WI?	V	C, V ce	V, C	C, V ce	120
Plethodon elongatus	Del Norte Salamander	J, A	SP, AU, WI?	V	C, V ce	V, C	C, V ce	100
Plethodon idahoensis	Coeur d'Alene Salamander	J, A	SP, AU, SU?, WI?	V	C, V ce	V, C	C, V ce	120
Plethodon larselli	Larch Mountain Salamander	J, A	SP, AU, WI?	V	C, V ce	V, C	C, V ce	100
Plethodon stormi	Siskiyou Mountains Salamander	J, A	SP, AU, WI?	V	C, V ce	V, C	C, V ce	100
Plethodon vandykei	Van Dyke's Salamander	J, A	SP, AU, WI?	V	C, V ce	V, C	C, V ce	120
Plethodon vehiculum	Western Redback Salamander	J, A	SP, AU, WI?	C, V, V r	C, V ce	V, C, D pf	C, V ce, D pf	121, 158
Rhyacotriton cascadae	Cascade Torrent Salamander	A	YR	V, DN	V ce, DN	V, DN	V ce, DN	102
Rhyacotriton cascadae	Cascade Torrent Salamander	L	YR	V, DN	V ce, DN	V, DN	V ce, DN	102
Rhyacotriton kezeri	Columbia Torrent Salamander	A	YR	V, DN	V ce, DN	V, DN	V ce, DN	102
Rhyacotriton kezeri	Columbia Torrent Salamander	L	YR	V, DN	V ce, DN	V, DN	V ce, DN	102
Rhyacotriton olympicus	Olympic Torrent Salamander	A	YR	V, DN	V ce, DN	V, DN	V ce, DN	102
Rhyacotriton olympicus	Olympic Torrent Salamander	L	YR	V, DN	V ce, DN	V, DN	V ce, DN	102
Rhyacotriton variegatus	Southern Torrent Salamander	A	YR	V, DN	V ce, DN	V, DN	V ce, DN	102
Rhyacotriton variegatus	Southern Torrent Salamander	L	YR	V, DN	V ce, DN	V, DN	V ce, DN	102
Taricha granulosa	Rough-skinned Newt	A	YR	C, V, V r	C, V ce, V r	C, V, V r, D pf	C, V ce, V r, D pf	104
Taricha granulosa	Rough-skinned Newt	J	AU	C, V, V r	C, V ce	C, V, V r, D pf	V ce, D pf	103, 158
Taricha granulosa	Rough-skinned Newt	L	SP, SU, AU	V, DN	V ce, DN, T mt	V, DN, T mt	V ce, DN	
Taricha rivularis	Red-bellied Newt	A	YR	V, V r	V ce, V r	V, V r	V ce, V r	104
Taricha rivularis	Red-bellied Newt	J	AU	V	V ce	V	V ce	103
Taricha rivularis	Red-bellied Newt	L	SP, SU	V, DN	V ce, DN, T mt	V, DN	V ce, DN, T mt	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Taricha torosa	California Newt	A	YR	V, V r	V ce, V r	V, V r	V ce, V r	104
Taricha torosa	California Newt	J	AU	V	V ce	V	V ce	103
Taricha torosa	California Newt	L	SP, SU	V, DN	V ce, DN, T mt	V, DN	V ce, DN, T mt	
MIDWEST REGION								
Ambystoma annulatum	Ringed Salamander	A	AU	V, V r	D pf	V	D pf	124
Ambystoma annulatum	Ringed Salamander	E	AU	V	V	V	CT	122
Ambystoma annulatum	Ringed Salamander	L	SP, AU, WI	DN	DN, T mt	DN	DN, T mt	123
Ambystoma barbouri	Streamside Salamander	A	AU, WI, SP	V, T mt	D pf	T mt	D pf	127
Ambystoma barbouri	Streamside Salamander	E	WI, SP	C	N/A	N/A	N/A	125
Ambystoma barbouri	Streamside Salamander	L	SP, SU	DN	T mt	T mt	T mt	126
Ambystoma jeffersonianum	Jefferson Salamander	A	SP	V	D pf	V	D pf	128
Ambystoma jeffersonianum	Jefferson Salamander	E	SP	V	V	V	V	122
Ambystoma jeffersonianum	Jefferson Salamander	L	SP, SU	DN	DN, T mt	DN, T mt	T mt	126
Ambystoma laterale	Blue-spotted Salamander	A	SP	V	D pf	V	D pf	128
Ambystoma laterale	Blue-spotted Salamander	E	SP	V	V	V	CT	122
Ambystoma laterale	Blue-spotted Salamander	L	SP, SU	DN	DN, T mt	DN, T mt	T mt	126
Ambystoma maculatum	Spotted Salamander	A	SP	V	D pf	V	D pf	128
Ambystoma maculatum	Spotted Salamander	E	SP	V	V	V	CT	122
Ambystoma maculatum	Spotted Salamander	L	SP, SU	DN	DN, T mt	DN, T mt	D pf	126
Ambystoma mavortium	Barred Tiger Salamander	A	SP	V	V ce, V r	V	V ce, V r	128
Ambystoma mavortium	Barred Tiger Salamander	E	SP	V	V ce	V	CT	133
Ambystoma mavortium	Barred Tiger Salamander	L	SP, SU	V, DN	V ce, DN, T mt	V, DN	V ce, DN, T mt	126
Ambystoma opacum	Marbled Salamander	A	AU	C	D pf	C	D pf	130
Ambystoma opacum	Marbled Salamander	E	AU, WI	C	C	C	CT	129
Ambystoma opacum	Marbled Salamander	L	SP, SU	DN	DN, T mt	DN, T mt	D pf	126
Ambystoma talpoideum	Mole Salamander	A	SP	C	D pf	C	D pf	128
Ambystoma talpoideum	Mole Salamander	E	SP	V	V	V	N/A	131
Ambystoma talpoideum	Mole Salamander	L	SP, SU	DN	DN, T mt	DN, T mt	D pf	126
Ambystoma texanum	Small-mouthed Salamander	A	SP	V, T mt	D pf	T mt	D pf	128
Ambystoma texanum	Small-mouthed Salamander	E	SP	V	V	N/A	N/A	132
Ambystoma texanum	Small-mouthed Salamander	L	SP, SU	DN, T mt	T mt	T mt	T mt	126
Ambystoma tigrinum	Tiger Salamander	A	SP	V	D pf	V	D pf	128
Ambystoma tigrinum	Tiger Salamander	E	SP	V	V	V	CT	133
Ambystoma tigrinum	Tiger Salamander	L	SP, SU	V, DN	DN, T mt	DN, T mt	D pf	126
Amphiuma tridactylum	Three-toed Amphiuma	A	YR	T mt	T fy	T mt	T mt	134
Amphiuma tridactylum	Three-toed Amphiuma	L	YR	T mt	T fy	T mt	T mt	134
Aneides aeneus	Green Salamander	A	YR	V	V	V	MR	135
Aneides aeneus	Green Salamander	J	YR	V	V	V	MR	135
Cryptobranchus alleghaniensis	Hellbender	A	YR	C, V vb	C, V vb, V s	C	MR	136
Cryptobranchus alleghaniensis	Hellbender	J	YR	C	C, V s	C	N/A	136

Scientific Name	Common Name	Life Stage	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Desmognathus conanti	Spotted Dusky Salamander	A	SP, SU, AU	C	C [w]	C	C [w]	137
Desmognathus conanti	Spotted Dusky Salamander	E	SU	V	V	V	V [ce]	137
Desmognathus conanti	Spotted Dusky Salamander	L	SU, AU	C	C	C	T [lb]	137
Desmognathus fuscus	Northern Dusky Salamander	A	SP, SU, AU	C	C [w]	C	C [w]	137
Desmognathus fuscus	Northern Dusky Salamander	E	SU	C	C	C	N/A	137
Desmognathus fuscus	Northern Dusky Salamander	L	SU, AU	C	C	C	T [lb]	137
Desmognathus ochrophaeus	Allegheny Mountain Dusky Salamander	A	SP, SU, AU	C	D [pf]	C	D [pf]	138
Desmognathus ochrophaeus	Allegheny Mountain Dusky Salamander	L	SU, AU	C	DN, T [lb]	C	T [lb]	137
Eurycea bislineata	Northern Two-lined Salamander	A	SP, SU	C	C	C	MR, R	139
Eurycea bislineata	Northern Two-lined Salamander	L	SU	C, DN	C, DN	C, DN	MR, R	139
Eurycea cirrigera	Southern Two-lined Salamander	A, J	SP, SU, AU	C	C [w]	C	C [w]	142
Eurycea cirrigera	Southern Two-lined Salamander	E	SP, SU	C [r]	C [r]	C [r]	N/A	140
Eurycea cirrigera	Southern Two-lined Salamander	L	SP, SU, AU	C	C	C	T [lb]	141
Eurycea longicauda	Long-tailed Salamander	A	SP, SU	C, V	C [w], V	C [w], V	C [w], V	143
Eurycea longicauda	Long-tailed Salamander	L	SU	C	C	C	T [lb]	143
Eurycea lucifuga	Cave Salamander	A	SP, SU, AU	V	V	V	V [ce]	144
Eurycea lucifuga	Cave Salamander	L	SP, SU, AU	C	C	C	T [lb]	144
Eurycea multiplicata	Many-ribbed Salamander	A	SP, AU, WI	C [r]	C [r]	C [r]	V [ce]	145
Eurycea multiplicata	Many-ribbed Salamander	L	WI, SP, SU	C [r]	C [r]	C [r]	V [ce]	145
Eurycea tynerensis	Oklahoma Salamander	A	SP, SU	C	C	C	T [lb]	145
Eurycea tynerensis	Oklahoma Salamander	L	SP, SU	C	C	C	T [lb]	145
Gyrinophilus porphyriticus	Spring Salamander	A	SP, SU	C [r]	C [r]	C [r]	V [ce]	145
Gyrinophilus porphyriticus	Spring Salamander	L	SP, SU	C [r]	C [r]	C [r]	V [ce]	145
Hemidactylium scutatum	Four-toed Salamander	A	SP, SU, AU	C	C	C	MR	148
Hemidactylium scutatum	Four-toed Salamander	E	SP	V	V	V	CT	146
Hemidactylium scutatum	Four-toed Salamander	L	SU	DN	DN	DN	DN	147
Necturus maculosus	Mudpuppy	A	YR	C [r]	T [hl]	C [r]	MR	149
Notophthalmus viridescens	Eastern Newt	A, L	SP, SU	DN	T [mt]	T [mt]	T [mt]	150
Plethodon albagula	Western Slimy Salamander	A, J	SP, SU, AU	C	C, V	V , C	C, MR	151
Plethodon angusticlavius	Ozark Zigzag Salamander	A, J	SP, SU, AU	C	C [w]	C [w]	C [w]	151
Plethodon cinereus	Eastern Red-backed Salamander	A, J	SP, SU, AU	C	C [w]	C [w]	C [w]	151
Plethodon dorsalis	Northern Zigzag Salamander	A, J	SP, SU, AU	C	C	C	N/A	151
Plethodon electromorphus	Northern Ravine Salamander	A, J	SP, SU, AU	C	C [w]	C [w]	C [w]	151
Plethodon glutinosus	Northern Slimy Salamander	A, J	SP, SU, AU	C	C [w]	C [w]	C [w]	151
Plethodon serratus	Southern Red-backed Salamander	A, J	SP, SU, AU	C	C	C	C	151
Plethodon wehrlei	Wehrle's Salamander	A, J	SP, SU, AU	C	C	C	C	151
Pseudotriton montanus	Mud Salamander	A, J	SP, SU, AU	DN, C	DN, C [w]	DN, C	DN, C	152
Pseudotriton ruber	Red Salamander	A, J	SP, SU, AU	DN, C	C, DN	C, DN	C, T [lb]	152
Siren intermedia	Lesser Siren	A, J	YR	T [cr], T [mt]	T [cr], T [fy]	T [cr], T [mt]	T [cr], T [mt]	134
Typhlotriton spelaeus	Grotto Salamander	A, J	YR	V	V	V	V [ce]	134

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TURTLES

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
SOUTHEAST REGION							
Apalone ferox	Florida Softshell	SP, SU, AU	V bk	T h	T h	T h, MR	
Apalone mutica	Smooth Softshell	SP, SU, AU	V bk	T fy	T fy	T fy, MR	
Apalone spinifera	Spiny Softshell	SP, SU, AU	V bk	T fy	V bk	T fy, MR	
Caretta caretta	Loggerhead Sea Turtle (Nests)	SP, SU	V cr	V	V	V	
Chelonia mydas	Green Sea Turtle (Nests)	SP, SU	V cr	V	V	V	
Chelydra serpentina	Snapping Turtle	SP, SU, AU	T h	T h	T h	T h, MR	
Chrysemys dorsalis	Southern Painted Turtle	SP, SU, AU	V bk	T h, T fy	V bk	T h, MR	
Chrysemys picta	Painted Turtle	SP, SU, AU	V bk	T h, T fy	V bk	T h, MR	
Clemmys guttata	Spotted Turtle	SP	V bk	V bk, T h	V bk	MR	
Deirochelys reticularia	Chicken Turtle	SP, SU	T fy	T fy, D	D	T fy, MR	20
Dermochelys coriacea	Leatherback Sea Turtle (Nests)	SP, SU	V cr	V	V	V	
Eretmochelys imbricata	Hawksbill Sea Turtle (Nests)	SP, SU	V cr	V	V	V	
Glyptemys muhlenbergii	Bog Turtle	SP	V	V, T fa	V, T fa	MR, TR rt, V ce	1
Gopherus polyphemus	Gopher Tortoise (Nests)	SU	V b	V b	V b	V b	
Gopherus polyphemus	Gopher Tortoise (Juveniles)	AU	V b	V b	V b	MR	
Gopherus polyphemus	Gopher Tortoise (Adults)	YR	V b	V b	V b	V b, V t	
Graptemys barbouri	Barbour's Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	2
Graptemys ernsti	Escambia Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	2
Graptemys flavimaculata	Yellow-blotched Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	2
Graptemys geographica	Northern Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	2
Graptemys gibbonsi	Pascagoula Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	2
Graptemys nigrinoda	Black-knobbed Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	2
Graptemys oculifera	Ringed Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	2
Graptemys ouachitensis	Ouachita Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	2
Graptemys pearlensis	Pearl River Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	2
Graptemys pseudogeographica	False Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	2
Graptemys pulchra	Alabama Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	2
Kinosternon baurii	Striped Mud Turtle	SP, AU	T h	D	T h	MR	
Kinosternon subrubrum	Eastern Mud Turtle	SP, AU	T h	D pf	D pf	T h, D pf, MR	
Lepidochelys kempii	Kemp's Ridley Sea Turtle	SU, AU	V st	N/A	V st	N/A	
Macrochelys temminckii	Alligator Snapping Turtle	SP, SU, AU	V s	T h	V s, T h	T h, MR	
Malaclemys terrapin	Diamond-backed Terrapin	SU	V	SN, T tn	V, SN, T tn	MR, V, SN, T tn	
Pseudemys alabamensis	Alabama Red-bellied Cooter	SP, SU, AU	V bk	V bk, T bk	V bk	T bk, T fy, MR	
Pseudemys concinna	River Cooter	SP, SU, AU	V bk	V bk, T bk	V bk	T bk, T fy, MR	
Pseudemys floridana	Florida Cooter	SP, SU	V bk, T fy	V bk, T fy	V bk	T bk, T fy, MR	
Pseudemys nelsoni	Florida Red-bellied Cooter	SP, SU	V bk, T fy	V s	V bk	T fy, MR, V s	

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Pseudemys peninsularis	Peninsula Cooter	YR	V bk, T fy	V bk, T fy	V bk	T fy, MR	
Pseudemys rubriventris	Northern Red-bellied Cooter	SP, SU	V bk, T fy	T h, T fy, T bk	V bk	T fy, MR	
Pseudemys suwanniensis	Suwannee Cooter	SP, SU	V bk, T fy	V s	V bk	T fy, MR, V s	
Sternotherus carinatus	Razor-backed Musk Turtle	SP, SU	T h	T h	T h	T h, MR	
Sternotherus depressus	Flattened Musk Turtle	SP, SU	V	T h, V s	T h, V s	MR, T h, V s	
Sternotherus minor	Loggerhead Musk Turtle	SP, SU	V s, T h	V s, T h	V s, T h	MR, V s, T h	
Sternotherus odoratus	Eastern Musk Turtle	SP, SU	T h, T cr	T h	T h, T cr	T h, MR	
Terrapene carolina	Eastern Box Turtle	SP, SU	V, V r	V, V r	V, V r	MR, V, V r	
Trachemys scripta	Pond Slider	SP, SU	T h, V bk	T h, V bk	T h, V bk	T h, V bk	
NORTHEAST REGION							
Apalone mutica	Smooth Softshell	SP, SU, AU	V bk	T fy	T fy	T fy, MR	
Apalone spinifera	Spiny Softshell	SP, SU, AU	V bk	T fy	V bk	T fy, MR	
Caretta caretta	Loggerhead Sea Turtle (Nests)	SP, SU	V cr	V	V	V	
Chelydra serpentina	Snapping Turtle	SP, SU, AU	T fy, T h	T fy, T h	T fy, T h	T fy, T h	
Chrysemys picta	Painted Turtle	SP, SU, AU	V bk	T h, T fy	V bk	T h, MR	3
Clemmys guttata	Spotted Turtle	SP	V bk	V bk, T h	V bk	MR	
Deirochelys reticularia	Chicken Turtle	SP, SU	T fy	T fy, D	D	T fy, MR	
Dermochelys coriacea	Leatherback Sea Turtle	SP, SU	V cr	V	V	V	4
Emydoidea blandingii	Blanding's Turtle	SP, SU	T h, V bk	T h, V bk	T h, V bk	MR, T h, V bk	5
Eretmochelys imbricata	Hawksbill Sea Turtle	SP, SU	V cr	V	V	V	4
Glyptemys insculpta	Wood Turtle	YR	N, V, V vb	N, V, V vb	N, V, V vb	MR, N, V, V vb	6
Glyptemys muhlenbergii	Bog Turtle	SP	V	V, T fa	V, T fa	MR, TR rt, V ce	7
Graptemys geographica	Northern Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	8
Graptemys pseudogeographica	False Map Turtle	SP, SU	V bk	V bk, T bk	V bk	T bk, T fy, MR	
Kinosternon baurii	Striped Mud Turtle	SP, SU	T h	D	T h	MR	
Kinosternon subrubrum	Eastern Mud Turtle	SP, SU	T h	D pf	D pf	T h, D pf, MR	
Lepidochelys kempii	Kemp's Ridley Sea Turtle	SU, AU	V st	N/A	V st	N/A	
Malaclemys terrapin	Diamond-backed Terrapin	SP, SU	V	SN, T tn	V, SN, T tn	MR, V, SN, T tn	
Pseudemys concinna	River Cooter	SP, SU, AU	V bk	V bk, T bk	V bk	T bk, T fy, MR	
Pseudemys rubriventris	Northern Red-bellied Cooter	SP, SU, AU	V bk, T fy	T h, T fy, T bk	T h, T fy, T bk	T h, T fy, MR	
Sternotherus minor	Loggerhead Musk Turtle	SU	V s, T h	V s, T h	V s, T h	MR, V s, T h	9
Sternotherus odoratus	Eastern Musk Turtle	SP, SU, AU	T h, T cr	T h	T h, T cr	T h, MR	
Terrapene carolina	Eastern Box Turtle	SP, SU, AU	V, V r	V, V r	V, V r	MR, V, V r	10
Trachemys scripta	Pond Slider	SP, SU, AU	T h, V bk	T h, V bk	T h, V bk	T h, V bk	11
SOUTHWEST REGION							
Apalone mutica	Smooth Softshell	SP, SU, AU	V bk	T fy	V bk	T fy, MR	
Apalone spinifera	Spiny Softshell	SP, SU, AU	V bk	T fy	V bk	T fy, MR	
Caretta caretta	Loggerhead Sea Turtle (Nests)	SP, SU	V cr	V	V	V	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Chelonia mydas	Green Sea Turtle (Nests)	SP, SU	V cr	V	V	V	
Chelydra serpentina	Snapping Turtle	SP, SU, AU	T h	T h	T h	T h, MR	
Chrysemys picta	Painted Turtle	SP, SU, AU	V bk	T h, T fy	V bk	T h, MR	
Actinemys marmorata	Western Pond Turtle	SP	V bk	V bk, T h	V bk	MR	
Deirochelys reticularia	Chicken Turtle	All	T fy	T fy, D	D	T fy, MR	
Dermochelys coriacea	Leatherback Sea Turtle (Nests)	SP, SU	V cr	V	V	V	
Eretmochelys imbricata	Hawksbill Sea Turtle (Nests)	SP, SU	V cr	V	V	V	
Gopherus agassizii	Desert Tortoise	SP, SU, AU	V	V	V	V, MR	12
Gopherus berlandieri	Texas Tortoise	SP, SU, AU	V	V	V	V, MR	
Graptemys caglei	Cagle's Map Turtle	YR	V bk	V bk, T bk	V bk	T bk, T fy, MR	
Graptemys ouachitensis	Ouachita Map Turtle	YR	V bk	V bk, T bk	V bk	T bk, T fy, MR	
Graptemys pseudogeographica	False Map Turtle	YR	V bk	V bk, T bk	V bk	T bk, T fy, MR	
Graptemys versa	Texas Map Turtle	YR	V bk	V bk, T bk	V bk	T bk, T fy, MR	
Kinosternon arizonense	Arizona Mud Turtle	YR	T h	D	T h	MR	
Kinosternon flavescens	Yellow Mud Turtle	SP, SU, AU	V bk, T h	V bk, T h	V bk, T h	MR, V bk, T h	
Kinosternon hirtipes	Rough-footed Mud Turtle	YR	V, V bk, T bk	V, V bk, T bk	V, V bk, T bk	MR, V, V bk, T bk	
Kinosternon sonoriense	Sonoran Mud Turtle	SP, SU, AU	V, V bk	SN, N, V, T h	SN, N, V, T h	MR, SN, N, V, T h	
Kinosternon subrubrum	Eastern Mud Turtle	YR	T h	D pf	D pf	T h, D pf, MR	
Lepidochelys kempii	Kemp's Ridley Sea Turtle (Nests)	SU, AU	V st	N/A	V st	N/A	13
Lepidochelys kempii	Kemp's Ridley Sea Turtle	SU, AU	V st	N/A	V st	N/A	
Lepidochelys olivacea	Olive Ridley Sea Turtle	SU, AU	V st	N/A	V st	N/A	
Macroclemys temminckii	Alligator Snapping Turtle	YR	V s	T h	V s, T h	T h, MR	
Malaclemys terrapin	Diamond-backed Terrapin	YR	V	SN, T tn	V, SN, T tn	MR, V, SN, T tn	
Pseudemys concinna	River Cooter	YR	V bk	V bk, T bk	V bk	T bk, T fy, MR	
Pseudemys gorzugi	Rio Grande Cooter	SP, SU, AU	V bk	T h	V bk	T h, MR	
Pseudemys texana	Texas Cooter	YR	V bk, T fy	V s	V bk	T fy, MR, V s	
Sternotherus carinatus	Razor-backed Musk Turtle	YR	T h	T h	T h	T h, MR	
Sternotherus odoratus	Eastern Musk Turtle	YR	T h, T cr	T h	T h, T cr	T h, MR	
Terrapene carolina	Eastern Box Turtle	YR	V, V r	V, V r	V, V r	MR, V, V r	
Terrepene ornata	Ornate Box Turtle	YR	V ce	V ce	V ce	MR, V ce	14
Trachemys gaigae	Mexican Plateau Slider	SP, SU, AU	V bk	T h	V bk	T h, MR	
Trachemys scripta	Pond Slider	YR	T h, V bk	T h, V bk	T h, V bk	T h, V bk	
NORTHWEST REGION							
Apalone spinifera	Spiny Softshell	SP, SU	V bk	T fy	V bk	T fy, MR	
Caretta caretta	Loggerhead Sea Turtle	SU	V	V	V	V	
Chelonia mydas	Green Sea Turtle	SU	V	V	V	V	
Chelydra serpentina	Snapping Turtle	SP, SU, AU	T h	T h	T h	T h, MR	
Chrysemys picta	Western Painted Turtle	SP, SU, AU	V bk	T h, T fy	V bk	T h, MR	
Actinemys marmorata	Western Pond Turtle	SP, SU, AU	V bk	V bk, T h	V bk	MR	

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Dermochelys coriacea	Leather back Sea Turtle	SU	V	V	V	V	
Lepidochelys olivacea	Olive Ridley Sea Turtle	SU	V st	N/A	V st	N/A	
Terrapene ornata	Ornate Box Turtle	SP, SU, AU	V ce	V ce	V ce	MR, V ce	
Trachemys scripta	Pond Slider	SP, SU, AU	T h, V bk	T h, V bk	T h, V bk	T h, V bk	
MIDWEST REGION							
Apalone mutica	Smooth Softshell	SP, SU, AU	V bk	T fy	V bk	T fy, MR	15
Apalone spinifera	Spiny Softshell	SP, SU, AU	V bk	T fy	V bk	T fy, MR	15
Chelydra serpentina	Snapping Turtle	SP, SU, AU	T h	T h	T h	T h, MR	16
Chrysemys picta	Painted Turtle	SP, SU, AU	V bk	T h, T fy	V bk	T h, MR	17
Chrysemys dorsalis	Southern Painted Turtle	SP, SU, AU	V bk	T h, T fy	V bk	T h, MR	17
Glyptemys insculpta	Wood Turtle	SP, SU	N, V, V vb	N, V, V vb	N, V, V vb	MR, N, V, V vb	18
Clemmys guttata	Spotted Turtle	SP, SU, AU	V bk	V bk, T h	V bk	MR	19
Deirochelys reticularia	Chicken Turtle	SP, SU	T fy	T fy, D	D	T fy, MR	20
Emydoidea blandingii	Blanding's Turtle	SP, SU, AU	T h, V bk	T h, V bk	T h, V bk	MR, T h, V bk	21
Graptemys geographica	Northern Map Turtle	SP, SU, AU	V bk	V bk, T bk	V bk	T bk, T fy, MR	22
Graptemys ouachitensis	Ouachita Map Turtle	SP, SU, AU	V bk	V bk, T bk	V bk	T bk, T fy, MR	22
Graptemys pseudogeographica	False Map Turtle	SP, SU, AU	V bk	V bk, T bk	V bk	T bk, T fy, MR	22
Kinosternon flavescens	Yellow Mud Turtle	SP, SU, AU	V bk, T h	V bk, T h	V bk, T h	MR, V bk, T h	23
Kinosternon subrubrum	Eastern Mud Turtle	SP, SU, AU	T h	D pf	D pf	T h, D pf, MR	23
Macrochelys temminckii	Alligator Snapping Turtle	SP, SU, AU	V s	T h	V s, T h	T h, MR	23
Pseudemys concinna	River Cooter	SP, SU, AU	V bk	V bk, T bk	V bk	T bk, T fy, MR	23
Sternotherus odoratus	Eastern Musk Turtle	SP, SU, AU	T h, T cr	T h	T h, T cr	T h, MR	16
Terrapene carolina	Eastern Box Turtle	SP, SU, AU	V, V r	V, V r	V, V r	MR, V, V r	24
Terrapene ornata	Ornate Box Turtle	SP, SU, AU	V ce	V ce	V ce	MR, V ce	24
Trachemys scripta	Pond Slider	SP, SU, AU	T h, V bk	T h, V bk	T h, V bk	T h, V bk	17

TABLE 5-1: SPECIES X TECHNIQUES TABLE

LIZARDS

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
SOUTHEAST REGION							
Amphisbaenians							
Rhineura floridana	Florida Worm Lizard	YR	N/A	D [pf]	D [pf]	N/A	1
Lizards							
Anolis carolinensis	Green Anole	WI, SP, SU, AU	V	V, T [pv]	V, T [pv]	V [ce]	
Cnemidophorus sexlineatus	Six-lined Racerunner	SU	V	V	V	V [ce]	
Eumeces anthracinus	Coal Skink	SU	C	N/A	C	N/A	2
Eumeces egregius	Mole Skink	SP, SU	C	D [pf]	C	N/A	3
Eumeces fasciatus	Common Five-lined Skink	SP, SU	V, A [a]	C	V, A [a]	N/A	
Eumeces inexpectatus	Southeastern Five-lined Skink	SP, SU	V, A [a]	C	V, A [a]	N/A	
Eumeces laticeps	Broad-headed Skink	SP, SU	V, A [a]	C	V, A [a]	N/A	
Neoseps reynoldsi	Florida Sand Skink	SP, SU	C	D [pf]	D [pf]	N/A	
Ophisaurus attenuatus	Slender Glass Lizard	SP, SU	V, C, V [r]	V, D, [ft], C, V [r]	V, C, V [r]	MR	4
Ophisaurus compressus	Island Glass Lizard	SP, SU	V, C, V [r]	V, D, [ft], C, V [r]	V, C, V [r]	MR	4
Ophisaurus mimicus	Mimic Glass Lizard	SP, SU	V, C, V [r]	V, D, [ft], C, V [r]	V, C, V [r]	MR	4
Ophisaurus ventralis	Eastern Glass Lizard	SP, SU	V, C, V [r]	V, D, [ft], C, V [r]	V, C, V [r]	MR	4
Sceloporus undulatus	Eastern Fence Lizard	SP, SU	V	V	V	MR	
Sceloporus woodi	Florida Scrub Lizard	SP, SU	V	V	V	MR	
Scincella lateralis	Little Brown Skink	SP, SU	V	D [pf]	V	V [ce]	
Sphaerodactylus notatus	Reef Gecko	YR	V	V	V	V [ce]	
NORTHEAST REGION							
Cnemidophorus sexlineatus	Six-lined Racerunner	SU	V	V	V	V [ce]	
Eumeces anthracinus	Coal Skink	SU	C	C	C	MR	
Eumeces fasciatus	Common Five-lined Skink	SP, SU	V, C [b]	C	V, C [b]	MR	
Eumeces inexpectatus	Southeastern Five-lined Skink	SP, SU	V, C [b]	C	V, C [b]	MR	
Eumeces laticeps	Broad-headed Skink	SP, SU	V, C [b]	C	V, C [b]	MR	
Ophisaurus attenuatus	Slender Glass Lizard	SP, SU	V, C, V [r]	D [ft], V, C, V [r]	V, C, V [r]	MR	4
Ophisaurus ventralis	Eastern Glass Lizard	SP, SU	V, C, V [r]	D [ft], V, C, V [r]	V, C, V [r]	MR	4
Sceloporus undulatus	Eastern Fence Lizard	SP, SU	V	V	V	MR	
Scincella lateralis	Little Brown Skink	SP, SU	V	D [pf]	V	MR	
SOUTHWEST REGION							
Anniella pulchra	California Legless Lizard	All	C, D [pf], SF	C, D [pf], SF	C, D [pf], SF	C, D [pf], SF	
Anolis carolinensis	Green Anole	WI, SP, SU, AU	V	V, T [pv]	V, T [pv]	V [ce]	
Callisaurus draconoides	Zebra-tailed Lizard	SP, SU, AU	V, V [r]	D [pf], V	V	D [pf], V [ce], MR	
Cnemidophorus arizonae	Arizona Striped Whiptail	SP, SU, AU	V	D [pf]	V	D [pf], MR	
Cnemidophorus burti	Canyon Spotted Whiptail	SP, SU, AU	V	D [pf]	V	D [pf], MR	
Cnemidophorus dixoni	Gray Checkered Whiptail	SP, SU, AU	V	D [pf]	V	D [pf], MR	

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Cnemidophorus exsanguis	Chihuahuan Spotted Whiptail	SP, SU, AU	V	D [pf], V	V	D [pf], V [ce], MR	
Cnemidophorus flagellicauda	Gila Spotted Whiptail	SP, SU, AU	V	D [pf]	V	D [pf], V [ce], MR	
Cnemidophorus gularis	Common Spotted Whiptail	SP, SU, AU	V	D [pf]	V	D [pf], V [ce], MR	
Cnemidophorus gypsi	Little White Whiptail	SP, SU, AU	V	D [pf]	V	D [pf], V [ce], MR	
Cnemidophorus hyperythra	Orange-throated Whiptail	SP, SU, AU	V	D [pf]	V	D [pf], V [ce], MR	
Cnemidophorus inornata	Little Striped Whiptail	SP, SU, AU	V	D [pf]	V	D [pf], V [ce], MR	
Cnemidophorus laredoensis	Laredo Striped Whiptail	SP, SU, AU	V	D [pf]	V	D [pf], MR	
Cnemidophorus marmorata	Marbled Whiptail	SP, SU, AU	V	D [pf]	V	D [pf], MR	
Cnemidophorus neomexicana	New Mexico Whiptail	SP, SU, AU	V	V, D [pf]	V	D [pf], V [ce], MR	
Cnemidophorus neotesselata	Colorado Checkered Whiptail	SP, SU, AU	V	V, D [pf]	V	D [pf], V [ce], MR	
Cnemidophorus pai	Pai Striped Whiptail	SP, SU, AU	V	D [pf]	V	D [pf], MR	
Cnemidophorus scalaris	Plateau Spotted Whiptail	SP, SU, AU	D [pf], V [ce]	D [pf]	V	D [pf], MR	
Cnemidophorus sexlineatus	Six-lined Racerunner	SP, SU, AU	D [pf], V [ce]	D [pf], V [ce]	D [pf], V [ce]	D [pf], V [ce], MR	
Cnemidophorus sonorae	Sonoran Spotted Whiptail	SP, SU, AU	D [pf], V [ce]	D [pf], V [ce]	V	D [pf], MR	
Cnemidophorus tesselata	Common Checkered Whiptail	SP, SU, AU	D [pf], V [ce]	D [pf], V [ce]	V	D [pf], V [ce], MR	
Cnemidophorus tigris	Tiger Whiptail	SP, SU, AU	D [pf], V [ce]	D [pf], V [ce]	V	D [pf], V [ce], MR	
Cnemidophorus uniparens	Desert Grassland Whiptail	SP, SU, AU	D [pf], V [ce]	D [pf]	V	D [pf], V [ce], MR	
Cnemidophorus velox	Plateau Striped Whiptail	SP, SU, AU	D [pf], V [ce]	D [pf]	V	D [pf], MR	
Cnemidophorus xanthonota	Red-backed Whiptail	SP, SU, AU	D [pf], V [ce]	D [pf]	V	D [pf], MR	
Coleonyx brevis	Texas Banded Gecko	SP, SU, AU	V, V [r], C	V [r], C, D [pf]	V, V [r], C	D [pf], MR, V [ce]	
Coleonyx reticulatus	Reticulated Gecko	SP, SU, AU	V, V [r], C	V [r], C, D [pf]	V, V [r], C	D [pf], MR, V [ce]	
Coleonyx switaki	Switak's Banded Gecko	SP, SU, AU	V [r], V, D [pf]	V [r], V, D [pf]	V [r], V, D [pf]	D [pf], MR, V [ce]	
Coleonyx variegatus	Western Banded Gecko	SP, SU, AU	V, V [r], C	V, D [pf]	V, V [r], C	D [pf], MR, V [ce]	
Cophosaurus texanus	Greater Earless Lizard	SP, SU, AU	V, D	V, D	V	D, MR	
Crotaphytus bicinctores	Great Basin Collared Lizard	SP, SU, AU	V	V, D [pf]	V	D [pf], MR	
Crotaphytus collaris	Eastern Collared Lizard	SP, SU, AU	V	V, D [ft]	V	MR	
Crotaphytus nebrius	Sonoran Collared Lizard	SP, SU, AU	V	V, D [ft]	V	MR	
Crotaphytus reticulatus	Reticulate Collared Lizard	SP, SU, AU	V	V, D [ft]	V	MR	
Crotaphytus vestigium	Baja California Collared Lizard	SP, SU, AU	V	V, D [ft]	V	MR	
Dipsosaurus dorsalis	Desert Iguana	SP, SU, AU	V	V	V	MR, V [ce]	
Elgaria coerulea	Northern Alligator Lizard	SP, AU, SU	V, C	V, D [pf], C	V, C	D [pf], MR	
Elgaria kingii	Madrean Alligator Lizard	SP, SU, AU	V, C	V, D [pf], C	V	D [pf], MR	
Elgaria multicarinata	Southern Alligator Lizard	SP, AU, SU	V, C	V, D [pf], C	V, C	D [pf], MR	
Elgaria panamintina	Panamint Alligator Lizard	SP, AU, SU	V, C	V, D [pf], C	V, C	D [pf], MR	
Eumeces anthracinus	Coal Skink	SP, SU, AU	C	V, D [pf], C	C	D [pf], MR	
Eumeces callicephalus	Mountain Skink	SP, SU, AU	C	D [pf]	C	D [pf], MR	
Eumeces fasciatus	Common Five-lined Skink	SP, SU, AU	C	D [pf]	C	D [pf], MR	
Eumeces gilberti	Gilbert's Skink	SP, SU, AU	C	D [pf]	C	D [pf], MR	
Eumeces laticeps	Broad-headed Skink	SP, SU, AU	C	D [pf]	C	D [pf], MR	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Eumeces multivirgatus	Many-lined Skink	SP, SU, AU	C	D pf	C	D pf, MR	
Eumeces obsoletus	Great Plains Skink	SP, SU, AU	C	D pf	C	D pf, MR	
Eumeces septentrionalis	Prairie Skink	SP, SU, AU	C	D pf	C	D pf, MR	
Eumeces skiltonianus	Western Skink	SP, SU, AU	C	D pf	C	D pf, MR	
Eumeces tetragrammus	Four-lined Skink	SP, SU, AU	C	D pf	C	D pf, MR	
Gambelia copeii	Cope's Leopard Lizard	SP, SU, AU	V, V r	V, D pf	V	D pf, MR	
Gambelia sila	Blunt-nosed Leopard Lizard	SP, SU, AU	V, V r	V, D pf	V	D pf, MR	
Gambelia wislizenii	Long-nosed Leopard Lizard	SP, SU, AU	V, V r	V, D pf	V	D pf, MR	
Gerrhonotus infernalis	Texas Alligator Lizard	SP, SU, AU	V, C, V r	D pf, C	V, C	D pf, MR	
Heloderma suspectum	Gila Monster	SP, SU, AU	V, V r	V, D pf	V, V b	D pf, MR, TR rt	5
Holbrookia elegans	Elegant Earless Lizard	SP, SU, AU	V	D pf	V	D pf, MR	
Holbrookia lacerata	Spot-tailed Earless Lizard	SP, SU, AU	V	D pf	V	D pf, MR	
Holbrookia maculata	Common Lesser Earless Lizard	SP, SU, AU	V	D pf	V	D pf, MR	
Holbrookia propinqua	Keeled Earless Lizard	SP, SU, AU	V	D pf	V	D pf, MR	
Ophisaurus attenuatus	Slender Glass Lizard	SP, SU	V, V r, C	V, V r, C, D pf	V, V r, C	MR	4
Petrosaurus mearnsi	Banded Rock Lizard	SP, SU, AU	V	V	V	V, MR	
Phrynosoma cornutum	Texas Horned Lizard	SP, SU, AU	V, V r	D pf	V, V r	MR, D pf	
Phrynosoma coronatum	Blainville's Horned Lizard	SP, SU, AU	V, V r	D pf	V, V r	MR, D pf	
Phrynosoma douglasii	Pygmy Short-horned Lizard	SP, SU, AU	V, V r	D pf	V, V r	MR, D pf	
Phrynosoma goodei	Goode's Horned Lizard	SP, SU, AU	V	V, D pf	V	D pf, MR	
Phrynosoma hernandesi	Greater Short-horned Lizard	SP, SU, AU	V, V r	D pf	V, V r	MR, D pf	
Phrynosoma mcallii	Flat-tailed Horned Lizard	SP, SU, AU	V, V r	D pf	V, V r	MR, D pf	
Phrynosoma modestum	Round-tailed Horned Lizard	SP, SU, AU	V	V, D pf	V	D pf, MR	
Phrynosoma platyrhinos	Desert Horned Lizard	SP, SU, AU	V	V, D pf	V, V r	D pf, MR	
Phrynosoma solare	Regal Horned Lizard	SP, SU, AU	V, V r	D pf	V, V r	MR, D pf	
Phyllodactylus xanti	Peninsular Leaf-toed Gecko	SP, SU, AU	V	V	V	V, MR	
Sauromalus obesus	Common Chuckwalla	SP, SU, AU	V, V r	V	V, V r	MR	
Sceloporus arenicolus	Dunes Sagebrush Lizard	SP, SU, AU	V	D pf	V	D pf, MR	
Sceloporus bimaculosus	Twin-spotted Spiny Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	
Sceloporus clarkii	Clark's Spiny Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	
Sceloporus consobrinus	Prairie Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	
Sceloporus cowlesi	Southwestern Fence Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	
Sceloporus cyanogenys	Blue Spiny Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	
Sceloporus graciosus	Common Sagebrush Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	
Sceloporus grammicus	Graphic Spiny Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	
Sceloporus jarrovii	Yarrow's Spiny Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	
Sceloporus magister	Desert Spiny Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	
Sceloporus merriami	Canyon Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	
Sceloporus occidentalis	Western Fence Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Sceloporus olivaceus	Texas Spiny Lizard	SP, SU, AU	V	V, D [pf]	V	V [ce], MR, D [pf]	
Sceloporus orcutti	Granite Spiny Lizard	SP, SU, AU	V	V, D [pf]	V	V [ce], MR, D [pf]	
Sceloporus poinsetti	Crevice Spiny Lizard	SP, SU, AU	V	V, D [pf]	V	V [ce], MR, D [pf]	
Sceloporus slevini	Slevin's Bunchgrass Lizard	SP, SU, AU	V	V, D [pf]	V	V [ce], MR, D [pf]	
Sceloporus tristichus	Plateau Fence Lizard	SP, SU, AU	V	V, D [pf]	V	V [ce], MR, D [pf]	
Sceloporus undulatus	Eastern Fence Lizard	SP, SU	V	V, D [pf]	V	V [ce], MR, D [pf]	
Sceloporus uniformis	Yellow-backed Spiny Lizard	SP, SU, AU	V	V, D [pf]	V	V [ce], MR, D [pf]	
Sceloporus variabilis	Rose-bellied Lizard	SP, SU, AU	V	V, D [pf]	V	V [ce], MR, D [pf]	
Sceloporus virgatus	Striped Plateau Lizard	SP, SU, AU	V	V, D [pf]	V	V [ce], MR, D [pf]	
Scincella lateralis	Little Brown Skink	SP, SU, AU	V, C	V, D [pf], C	V, C	V [ce], MR, D [pf]	
Uma inornata	Coachella Fringe-toed Lizard	SP, SU, AU	V	V, D [pf]	V	MR, D [pf]	
Uma notata	Colorado Fringe-toed Lizard	SP, SU, AU	V	V, D [pf]	V	MR, D [pf]	
Uma rufopunctata	Yuman Fringe-toed Lizard	SP, SU, AU	V	V, D [pf]	V	MR, D [pf]	
Uma scoparia	Mohave Fringe-toed Lizard	SP, SU, AU	V	V, D [pf]	V	MR, D [pf]	
Urosaurus graciosus	Long-tailed Brush Lizard	SP, SU, AU	V	D [pf], C	V, V [r]	MR, D [pf]	
Urosaurus nigricaudus	Baja California Brush Lizard	SP, SU, AU	V	D [pf], C	V	MR, D [pf]	
Urosaurus ornatus	Ornate Tree Lizard	SP, SU, AU	V	D [pf], C	V	MR, D [pf]	
Uta stansburiana	Common Side-blotched Lizard	SP, SU, AU	V	V, D [pf]	V	MR, D [pf]	
Xantusia arizonae	Arizona Night Lizard	SP, SU, AU	V, C	V, C	V, C	V [ce], C	
Xantusia bezyi	Bezy's Night Lizard	SP, SU, AU	V, C	V, C	V, C	V [ce], C	
Xantusia gracilis	Sandstone Night Lizard	SP, SU, AU	V, C	V, C	V, C	V [ce], C	
Xantusia henshawi	Granite Night Lizard	SP, SU, AU	V, C	V, C	V, C	V [ce], C	
Xantusia riversiana	Island Night Lizard	SP, SU, AU	V, C	V, C	V, C	V [ce], C	
Xantusia sierrae	Sierra Night Lizard	SP, SU, AU	V, C	V, C	V, C	V [ce], C	
Xantusia vigilis	Desert Night Lizard	SP	V	V	V	V [ce], C	
Xantusia wigginsi	Wiggins' Night Lizard	SP, SU, AU	V, C	V, C	V, C	V [ce], C	
NORTHWEST REGION							
Cnemidophorus tigris	Tiger Whiptail	SP, SU, AU	D [pf], V [ce]	D [pf], V [ce]	V	D [pf], V [ce], MR	
Cnemidophorus velox	Plateau Striped Whiptail	SP, SU, AU	D [pf], V [ce]	D [pf]	V	D [pf], MR	
Crotaphytus bicinctores	Great Basin Collared Lizard	SP, SU, AU	V	V, D [pf]	V	D [pf], MR	
Elgaria coerulea	Northern Alligator Lizard	SP, SU, AU	V, C	V, D [pf], C	V, C	D [pf], MR	
Elgaria multicarinata	Southern Alligator Lizard	SP, SU, AU	V, C	V, D [pf], C	V, C	D [pf], MR	
Eumeces gilberti	Gilbert's Skink	SP, SU, AU	V, C	V, D [pf], C	V, C	C, D [pf], MR	
Eumeces multivirgatus	Many-lined Skink	SP, SU, AU	C	D [pf]	C	D [pf], MR	
Eumeces skiltonianus	Western Skink	SP, SU, AU	C	D [pf]	C	D [pf], MR	
Gambelia wislizenii	Long-nosed Leopard Lizard	SP, SU, AU	V, V [r]	V, D [pf]	V	D [pf], MR, C	
Holbrookia maculata	Common Lesser Earless Lizard	SP, SU, AU	V	D [pf]	V	D [pf], MR	
Phrynosoma coronatum	Blainville's Horned Lizard	SP, SU, AU	V, V [r]	D [pf]	V, V [r]	MR, D [pf]	
Phrynosoma douglasii	Pygmy Short-horned Lizard	SP, SU, AU	V, V [r]	D [pf]	V, V [r]	MR, D [pf]	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Phrynosoma hernandesi	Greater Short-horned Lizard	SP, SU, AU	V, V r	D pf	V, V r	MR, D pf	
Phrynosoma platyrhinos	Desert Horned Lizard	SP, SU, AU	V	V, D pf	V, V r	D pf, MR	
Podarcis muralis	European Wall Lizard	YR	V	V	V	CT, F	
Sceloporus graciosus	Common Sagebrush Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf, C	
Sceloporus occidentalis	Western Fence Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	
Sceloporus undulatus	Eastern Fence Lizard	SP, SU, AU	V	V	V	MR, C	
Urosaurus ornatus	Ornate Tree Lizard	SP, SU, AU	V	D pf, C	V	MR, D pf	
Uta stansburiana	Common Side-blotched Lizard	SP, SU, AU	V	V, D pf	V	MR, D pf	
MIDWEST REGION							
Cnemidophorus sexlineatus	Six-lined Racerunner	SP, SU, AU	D pf, V ce	D pf, V ce	D pf, V ce	D pf, V ce, MR	6
Crotaphytus collaris	Eastern Collared Lizard	SP, SU, AU	V	V, D ft	V	MR	
Eumeces anthracinus	Coal Skink	SP, SU, AU	C	V, D pf, C	C	D pf, MR	7
Eumeces fasciatus	Common Five-lined Skink	SP, SU, AU	C	D pf	C	D pf, MR	7
Eumeces laticeps	Broad-headed skink	SP, SU, AU	C	D pf	C	D pf, MR	7
Eumeces multivirgatus	Many-lined Skink	SP, SU, AU	C	D pf	C	D pf, MR	7
Eumeces obsoletus	Great Plains Skink	SP, SU, AU	C	D pf	C	D pf, MR	7
Eumeces septentrionalis	Prairie Skink	SP, SU, AU	C	D pf	C	D pf, MR	7
Holbrookia maculata	Common Lesser Earless Lizard	SP, SU, AU	V	D pf	V	D pf, MR	8
Ophisaurus attenuatus	Slender Glass Lizard	SP, SU, AU	V, V r, C	V, V r, C, D pf	V, V r, C	MR	9
Phrynosoma cornutum	Texas Horned Lizard	SP, SU, AU	V, V r	D pf	V, V r	MR, D pf	7
Phrynosoma hernandesi	Greater Short-horned Lizard	SP, SU, AU	V, V r	D pf	V, V r	MR, D pf	7
Sceloporus consobrinus	Prairie Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	7
Sceloporus graciosus	Common Sagebrush Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	7
Sceloporus undulatus	Eastern Fence Lizard	SP, SU, AU	V	V, D pf	V	V ce, MR, D pf	10
Scincella lateralis	Little Brown Skink	SP, SU, AU	V, C	V, D pf, C	V, C	V ce, MR, D pf	7

SNAKES

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
SOUTHEAST REGION							
Agkistrodon contortrix	Copperhead	AU, SU	V [r]	V [r], C, D [ft]	V [r], C	V [r], C, MR	
Agkistrodon piscivorus	Cottonmouth	SP, SU, AU	V, V [r]	V, D [ft]	V	MR	
Carphophis amoenus	Eastern Worm Snake	SU, AU	C [w]	C [w]	C [w]	D [pf], C [w]	
Carphophis vermis	Western Worm Snake	SU, AU	C [w]	C [w]	C [w]	V [ce], C, D [pf]	
Cemophora coccinea	Scarlet Snake	SU	V [r], C	C, D [pf]	C, V [r]	MR, V [r]	
Clonophis kirtlandii	Kirtland's Snake	SU	C, D [pf]	C, D [pf]	C, D [pf]	C, MR	
Coluber constrictor	North American Racer	SP, SU, AU	C, V [r]	C, V [r]	C, V [r]	MR, V [ce], C, V [r]	
Crotalus adamanteus	Eastern Diamond-backed Rattlesnake	SP	V [r], V [b]	V [r], V [b]	V [r], V [b]	MR	
Crotalus atrox	Western Diamond-backed Rattlesnake	SP, SU, AU	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	MR, D [ft], V [r]	
Crotalus horridus	Timber Rattlesnake (Adults, Juveniles)	SP, SU, AU	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	MR, D [ft], V [r]	
Crotalus horridus	Timber Rattlesnake (hibernaculum)	AU, SP	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	MR, D [ft], V [r]	
Diadophis punctatus	Ring-necked Snake	SU, AU	C	C, D [pf]	C	C, MR	
Drymarchon couperi	Eastern Indigo Snake	AU, Wi, SP	V, V [b]	V, V [b]	V, V [b]	MR, TR [rt], V [ce]	
Elaphe guttata	Red Corn Snake	SP, SU, AU	C, V [r]	C, V [r]	C, V [r]	MR, V [ce]	
Elaphe obsoleta	Eastern Rat Snake	SP, SU, AU	V [r], C	V [r], C	V [r], C	MR, D [ft]	
Elaphe slowinskii	Slowinski's Corn Snake	SP	V [r], C	V [r], C	V [r], C	MR, D [ft]	
Farancia abacura	Red-bellied Mud Snake	SP, SU, AU	V [r], T [mt]	T [mt], T [fy], SN, C	T [mt], T [fy], SN, C	T [mt], T [fy], SN, C, MR	1
Farancia erytrogramma	Rainbow Snake	SP, SU, AU	T [mt]	T [mt], T [fy]	T [mt], C	MR	
Heterodon platirhinos	Eastern Hog-nosed Snake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	V [r], MR, C	
Heterodon simus	Southern Hog-nosed Snake	SP, AU	V [r], V	V [r], V	V [r], V, C	V [r], MR, C	
Lampropeltis calligaster	Yellow-bellied Kingsnake	SP, SU	V [r], V, C, D	V [r], V, C, D	V [r], V, C, D	MR	
Lampropeltis getula	Common Kingsnake	SP, SU, AU	V [r], V, C	V [r], D [pf], C, MR	V [r],V, D [pf], C	V [ce], V [r], MR	
Lampropeltis triangulum	Milk Snake	SP	V [r], V, C	V [r], D [pf], C, MR	V [r],V, D [pf], C	V [ce], V [r], MR	
Lampropeltis t. elapsoides	Scarlet Kingsnake	SP	V [r], V, C, C [b]	V [r], D [pf], C, MR	V [r],V, D [pf], C, C [b]	V [ce], V [r], MR	
Lampropeltis t. syspila	Red Milk Snake	SP	V [r], V, C	V [r], D [pf], C, MR	V [r],V, D [pf], C	V [ce], V [r], MR	
Masticophis flagellum	Coachwhip	SU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Micrurus fulvius	Harlequin Coral Snake	SP, AU	D [pf], C	D [pf], C	D [pf], C	D [pf], C, MR	
Nerodia clarkii	Saltmarsh Watersnake	SP,SU, AU	V	V	V	V [ce]	
Nerodia cyclopion	Mississippi Green Watersnake	SU	V [bk], V	V [bk], V, T [mt]	V [bk], V, T [mt]	MR	
Nerodia erythrogaster	Plain-bellied Watersnake	SP, SU, AU	V [bk], V	V [bk], V	V [bk], V, T [mt]	MR, TR [rt]	
Nerodia fasciata	Southern Watersnake	SP, SU	V [bk], V [r]	V [bk], V, T [mt]	V [bk], V, T [mt]	T [mt], MR	
Nerodia floridana	Florida Green Watersnake	SU	V [bk], V [r]	V [bk], V, T [mt]	V [bk], V, T [mt]	T [mt], MR	
Nerodia rhombifer	Diamond-backed Watersnake	SU	V [bk], V	V [bk], V	V [bk], V	MR	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Nerodia sipedon	Northern Watersnake	SP, SU	V [bk], V [r]	V [bk], V, T [mt]	V [bk], V, T [mt]	T [mt], MR	
Nerodia taxispilota	Brown Watersnake	SU	V [bk]	V [bk]	V [bk]	MR	
Opheodrys aestivus	Rough Green Snake	SP, SU	V, V [r]	V, V [r]	V, V [r]	MR	
Opheodrys vernalis	Smooth Green Snake	SP, SU	C	C	C	MR, C	
Pituophis melanoleucus	Pine Snake (Nests)	SP, SU	V	V	V	V	
Pituophis melanoleucus	Pine Snake (Adults, Juveniles)	SP, SU	V [r]	V [r], C, D [ft]	V [r], C	V [r], C, D [ft], MR	
Pituophis ruthveni	Louisiana Pine Snake	SP, SU	V [r], V	V [r], C, D [ft]	V [r], C	D [ft], MR	2
Regina alleni	Striped Crayfish Snake	SP, SU, AU	V, T [mt]	SN, T [mt]	T [mt]	SN, MR	
Regina grahamii	Graham's Crayfish Snake	SP, SU	T [mt]	T [mt]	T [mt]	T [mt], MR	
Regina rigida	Glossy Crayfish Snake	SU	T [mt]	T [mt]	T [mt]	T [mt], MR	
Regina septemvittata	Queen Snake	SU	C, V [bk]	C, V [bk]	C, V [bk]	MR, V [ce]	3
Rhadinaea flavilata	Pine Woods Snake	SP, SU, AU	C	C	C	MR	
Seminatrix pygaea	Black Swamp Snake	SP, SU, AU	T [mt]	C, T [mt]	SN, T [mt]	T [mt], MR	
Sistrurus catenatus	Massasauga	SP, SU, AU	V, V [r], C	D [ft], C	D [ft], C	MR, TR [rt]	
Sistrurus miliarius	Pygmy Rattlesnake	SU	V [r]	V [r]	V [r]	MR, TR [rt]	
Sonora semiannulata	Western Ground Snake	SP, SU	C, V [r]	C	C	MR	
Stilosoma extenuatum	Short-tailed Snake	SP, AU	D [pf]	D [pf]	D [pf]	MR	4
Storeria dekayi	Dekay's Brown Snake	SP, SU, AU	C, D [pf]	C, D [pf]	C, D [pf]	MR, D [pf]	
Storeria occipitomaculata	Red-bellied Snake	SP, SU, AU	C [w]	C [w], D [pf]	C [w]	MR, D [pf]	
Storeria victa	Florida Brown Snake	SP, SU, AU	C [w]	C [w], D [pf]	C [w]	MR, D [pf]	
Tantilla coronata	Southeastern Crowned Snake	SP, SU, AU	C, D [pf]	C, D [pf]	C, D [pf]	MR, C, D [pf]	
Tantilla gracilis	Flat-headed Snake	SP, SU, AU	C, D [pf]	C, D [pf]	C, D [pf]	MR, C, D [pf]	
Tantilla oolitica	Rim Rock Crowned Snake	SP, SU, AU	C, D [pf]	C, D [pf]	C, D [pf]	MR, C, D [pf]	
Tantilla relicta	Florida Crowned Snake	SP, SU, AU	C, D [pf]	C, D [pf]	C, D [pf]	MR, D [pf]	
Thamnophis proximus	Western Ribbon Snake	SP, SU, AU	V, V [r], C	D [ft]	V , V [r], C	V [ce], MR	
Thamnophis sauritus	Eastern Ribbon Snake	SP, SU, AU	V	C [w]	V	V [ce], MR	5
Thamnophis sirtalis	Common Garter Snake	SP, SU, AU	V, C, V [r]	C [w]	C [w]	MR, C [w]	
Virginia striatula	Rough Earth Snake	SP, SU, AU	C	C [w]	C	MR, C [w]	
Virginia valeriae	Smooth Earth Snake	SP, SU, AU	C	C [w]	C	MR, C [w]	
NORTHEAST REGION							
Agkistrodon contortrix	Copperhead	AU, SU	V [r]	V [r], C, D [ft]	V [r], C	MR, V [r], C, D [ft]	
Agkistrodon piscivorus	Cottonmouth	SP, SU, AU	V, V [r]	V [r], C, D [ft]	V	MR	
Carphophis amoenus	Eastern Worm Snake	SU, AU	C [w]	C [w]	C [w]	D [pf], C [w]	
Cemophora coccinea	Scarlet Snake	SU	V [r], C	C, D [pf]	C, V [r]	MR, V [r]	
Clonophis kirtlandii	Kirtland's Snake	SU	C, D [pf]	C, D [pf]	C, D [pf]	C, D [pf]	
Coluber constrictor	North American Racer	SP, SU, AU	C, V [r], V	C [m]	V [r], C	MR, D [ft]	

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Crotalus horridus	Timber Rattlesnake (Adults, Juveniles)	SP, SU, AU	D ft, V, C, V r	D ft, V, C, V r	D ft, V, C, V r	MR, D ft, V r	
Crotalus horridus	Timber Rattlesnake (Hibernaculum)	AU, SP	D ft, V, C, V r	D ft, V, C, V r	D ft, V, C, V r	MR, D ft, V r	
Crotalus horridus	Timber Rattlesnake (Coastal Plain population, Adults)	SP, SU, AU	D ft, V, C, V r	D ft, V, C, V r	D ft, V, C, V r	MR, D ft, V r	6
Crotalus horridus	Timber Rattlesnake (Coastal Plain population, Hibernaculum)	WI	D ft, V, C, V r	D ft, V, C, V r	D ft, V, C, V r	MR, D ft, V r	7
Diadophis punctatus	Ring-necked Snake	SU, AU	C	C, D pf	C	C, MR	
Elaphe guttata	Red Corn Snake	SP, SU, AU	V r, C	V r, C	V r, C	MR, D ft	
Elaphe obsoleta	Eastern Rat Snake	SP, SU, AU	V r, C	V r, C	V r, C	MR, D ft	
Farancia abacura	Red-bellied Mud Snake	SP, SU, AU	V r, T mt	T mt, T fy, SN, C	T mt, T fy, SN, C	MR	1
Farancia erytrogramma	Rainbow Snake	SP, SU, AU	T mt	T mt, T fy	T mt, C	MR	
Heterodon platirhinos	Eastern Hog-nosed Snake	SP, SU, AU	V r, V, C	V r, V, C	V r, V, C	V r, MR, C	
Lampropeltis calligaster	Yellow-bellied Kingsnake	SP, SU	V r, V, C, D	V r, V, C, D	V r, V, C, D	MR	
Lampropeltis getula	Common Kingsnake	SP, SU, AU	V r, V, C	V r, D pf, C, MR	V r,V, D pf, C	V ce, V r, MR	
Lampropeltis triangulum	Milk Snake	SP	V r, V, C	V r, D pf, C, MR	V r,V, D pf, C	V ce, V r, MR	
Nerodia erythrogaster	Plain-bellied Watersnake	SP, SU, AU	V bk, V	V bk, V	V bk, V	MR, TR rt	
Nerodia sipedon	Northern Watersnake	SP, SU	V bk, V r	V bk, V, T mt	V bk, V, T mt	T mt, MR	
Nerodia taxispilota	Brown Watersnake	SU	V bk	V bk	V bk	MR	
Opheodrys aestivus	Rough Green Snake	SP, SU	V, V r	V, V r	V, V r	MR	
Opheodrys vernalis	Smooth Green Snake	SP, SU	C	C	C	MR, C	
Pituophis melanoleucus	Pine Snake (Nests)	SP, SU	V	V	V	V	
Pituophis melanoleucus	Pine Snake (Adults, Juveniles)	SP, SU	V r	V r, C, D ft	V r, C	V r, C, D ft, MR	
Regina rigida	Glossy Crayfish Snake	SU	T mt	T mt	T mt	T mt, MR	
Regina septemvittata	Queen Snake	SU	C, V bk	C, V bk	C, V bk	MR, V ce	
Sistrurus catenatus	Massasauga	SP, SU, AU	V, V r, C	D ft, C	D ft, C	MR, TR rt	
Storeria dekayi	Dekay's Brown Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, D pf	
Storeria occipitomaculata	Red-bellied Snake	SP, SU, AU	C w	C w, D pf	C w	MR, D pf	
Tantilla coronata	Southeastern Crowned Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, C, D pf	
Thamnophis brachystoma	Short-headed Garter Snake	SP, SU	V	C w	V	V ce, MR	
Thamnophis sauritus	Eastern Ribbon Snake	SP, SU, AU	V	C w	V	V ce, MR	
Thamnophis sirtalis	Common Garter Snake	SP, SU, AU	V, C, V r	V, C, V r	V, C, V r	MR, V ce, C	
Virginia striatula	Rough Earth Snake	SP, SU, AU	C	C	C	C	
Virginia valeriae	Smooth Earth Snake	SP, SU, AU	C	C	C	C	
SOUTHWEST REGION							
Agkistrodon contortrix	Copperhead	AU, SU	V r	V r, C, D ft	V r, C	V r, C, MR	
Agkistrodon piscivorus	Cottonmouth	SP, SU, AU	V, V r	V, D ft	V	MR	
Arizona elegans	Glossy Snake	SP, SU, AU	V ce, D pf, TR	V ce, D pf, TR	V ce, D pf, TR	V ce, D pf, TR, MR	
Carphophis vermis	Western Worm Snake	SU, AU	C w	C w	C w	V ce, C, D pf	
Cemophora coccinea	Scarlet Snake	SU	V r, C	C, D pf	C, V r	MR, V r	
Charina trivirgata	Rosy Boa	SP, SU, AU	V ce, D pf	V ce, D pf	V, D pf	V ce, D pf, MR	
Charina bottae	Northern Rubber Boa	SP, SU, AU	V, V r, C	V, V r, C	V, V r, C	MR, V r, C	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Chilomeniscus cinctus	Burrowing Sand Snake	SP, SU, AU	V, SF, D [pf], C	V, SF, D [pf], C	V, SF, D [pf], C	V [ce], SF, D [pf], C	
Chionactus occipitalis	Western Shovel-nosed Snake	SP, SU, AU	V, SF, D [pf], C	V, SF, D [pf], C	V, SF, D [pf], C	V [ce], SF, D [pf], C	
Chionactus palarostris	Sonoran Shovel-nosed Snake	SP, SU, AU	V, SF, D [pf], C	V, SF, D [pf], C	V, SF, D [pf], C	V [ce], SF, D [pf], C	
Coluber constrictor	North American Racer	SP, SU, AU	C, V [r]	C, V [r]	C, V [r]	MR, V [ce], C, V [r]	
Coniophanes imperialis	Regal Black-striped Snake	SP, SU, AU	V, V [r], C, SF	V, V [r], C, SF	V, V [r], C, SF	MR, V, V [r], C, SF	
Contia tenuis	Sharp-tailed Snake	SP, SU, AU	V [ce], D [pf], C	V [ce], D [pf], C	V [ce], D [pf], C	MR, V [ce], D [pf], C	
Crotalus atrox	Western Diamond-backed Rattlesnake	SP, SU, AU	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	MR, D [ft], V [r]	
Crotalus cerastes	Sidewinder	SP, SU, AU	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	MR, D [ft], V [r]	
Crotalus cerberus	Arizona Black Rattlesnake	SP, SU, AU	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	MR, D [ft], V [r]	
Crotalus lepidus	Rock Rattlesnake	SP, SU, AU	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	MR, D [ft], V [r]	
Crotalus mitchelli	Speckled Rattlesnake	SP, SU, AU	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	MR, D [ft], V [r]	
Crotalus molossus	Black-tailed Rattlesnake	SP, SU, AU	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	MR, D [ft], V [r]	8
Crotalus oreganus	Western Rattlesnake	SP, SU, AU	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	D [ft]; V, C, V [r]	MR, D [ft], V [r]	
Crotalus pricei	Twin-spotted Rattlesnake	SP, SU, AU	V [ce]	V [ce]	V	MR, V, TR [rt]	9
Crotalus ruber	Red Diamond Rattlesnake	SP, SU, AU	V, V [r], DF [ft]	V, V [r], DF [ft]	V, V [r], DF [ft]	MR, V [r], DF [ft]	
Crotalus scutulatus	Mojave Rattlesnake	SP, SU, AU	V, V [r], DF [ft]	V, V [r], DF [ft]	V, V [r], DF [ft]	MR, V [r], DF [ft]	
Crotalus stephensi	Panamint Rattlesnake	SP, SU, AU	V, V [r], DF [ft]	V, V [r], DF [ft]	V, V [r], DF [ft]	MR, V [r], DF [ft]	
Crotalus tigris	Tiger Rattlesnake	SP, SU, AU	V, V [r], DF [ft]	V, V [r], DF [ft]	V, V [r], DF [ft]	MR, V [r], DF [ft]	10
Crotalus viridus	Prairie Rattlesnake	SP, SU, AU	V, V [r], DF [ft]	V, V [r], DF [ft]	V, V [r], DF [ft]	MR, V [r], DF [ft]	
Crotalus willardi	Ridge-nosed Rattlesnake	SP, SU, AU	V, V [r], DF [ft]	V, V [r], DF [ft]	V, V [r], DF [ft]	MR, V [r], DF [ft]	11
Diadophis punctatus	Ring-necked Snake	SP, SU, AU	C	C, D [pf]	C	C, MR	
Drymarchon corais	Central American Indigo Snake	SP, SU, AU	V, V [b]	V, V [b]	V, V [b]	MR, TR [rt], V, V [b]	
Drymobius margaritiferus	Speckled Racer	SP, SU, AU	C, V, V [r]	C, V, V [r]	C, V, V [r]	V [ce], V [r], MR	
Elaphe rosaliae	Baja California Rat Snake	SP, SU, AU	V, C	V, C	V, C	V [ce], MR	
Elaphe subocularis	Trans-Pecos Rat Snake	SP, SU, AU	V, V [r], C	V, V [r], C	V, V [r], C	V, V [r], C	
Elaphe bairdi	Baird's Rat Snake	SP, SU, AU	C, V [r]	C, V [r]	C, V [r]	MR, V [ce]	
Elaphe emoryi	Great Plains Ratsnake	SP, SU, AU	C, V [r]	C, V [r]	C, V [r]	MR, V [ce]	
Elaphe guttata	Red Corn Snake	SP, SU, AU	V [r], C	V [r], C	V [r], C	MR, D [ft]	
Elaphe obsoleta	Eastern Rat Snake	SP, SU, AU	V [r], C	V [r], C	V [r], C	MR, D [ft]	
Elaphe obsoleta	Texas Rat Snake	SP, SU, AU	V [r], C	V [r], C	V [r], C	MR, D [ft]	
Elaphe triaspis	Green Rat Snake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Ficimia streckeri	Tamaulipan Hook-nosed Snake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	MR, V [r], V, C	
Gyalopion canum	Chihuahuan Hook-nosed Snake	SP, SU, AU	V [r], V, C	D	V [r], D, C	D, MR	
Gyalopion quadrangulare	Thornscrub Hook-nosed Snake	SP, SU, AU	V [r], V, C	D	V [r], D, C	D, MR	
Heterodon kennerlyi	Mexican Hog-nosed Snake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	V [r], MR, C	
Heterodon nasicus	Plains Hog-nosed Snake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	V [r], MR, C	
Heterodon platirhinos	Eastern Hog-nosed Snake	SP, AU	V [r], V, C	V [r], V, C	V [r], V, C	V [r], MR, C	12
Hypsiglena torquata	Desert Night Snake	SP, SU, AU	V [r], V, C	V [r], V, C, D [pf]	V [r], D [pf]	D [pf], MR	
Hypsiglena torquata	Chihuahuan Night Snake	SP, SU, AU	V [r], V, C	V [r], V, C, D [pf]	V [r], D [pf]	D [pf], MR	

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Hypsiglena torquata	Coast Night Snake	SP, SU, AU	V [r], V, C	V [r], V, C, D [pf]	V [r], D [pf]	D [pf], MR	
Lampropeltis alterna	Gray-banded Kingsnake	SP, SU, AU	V [r], V, C	V [r], V, C, D	V [r], V, C	MR	
Lampropeltis calligaster	Yellow-bellied Kingsnake	SP, SU, AU	V [r], V, C, D	V [r], V, C, D	V [r], V, C, D	MR	
Lampropeltis getula	Common Kingsnake	SP, SU, AU	V [r], V, C	V [r], D [pf], C, MR	V [r], D [pf], C	V [ce], V [r], MR	
Lampropeltis pyromelana	Sonoran Mountain Kingsnake	SP, SU, AU	V [r], V, C	V [r], D [pf], C, MR	V [r],V, D [pf], C	V [ce], V [r], MR	
Lampropeltis triangulum	Milk Snake	SP, SU, AU	V [r], V, C	V [r], D [pf], C, MR	V [r],V, D [pf], C	V [ce], V [r], MR	
Lampropeltis zonata	California Mountain Kingsnake	SP, SU, AU	V [r], V, C	V [r], D [pf], C, MR	V [r],V, D [pf], C	V [ce], V [r], MR	
Leptodeira septentrionalis	Cat-eyed Snake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	V [ce], MR	
Leptotyphlops dissectus	New Mexico Thread Snake	SP, SU, AU	V [r], C	C, D [pf], V [r]	V [r], C, D	D, C, MR	
Leptotyphlops dulcis	Texas Thread Snake	SP, SU, AU	V [r], C	C, D [pf], V [r]	V [r], C, D	D, C, MR	
Leptotyphlops humilis	Western Thread Snake	SP, SU, AU	V [r], C	C, D [pf], V [r]	V [r], C, D	D, C, MR	
Masticophis bilineatus	Sonoran Whipsnake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Masticophis flagellum	Coachwhip	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Masticophis fuliginosus	Baja California Coachwhip	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Masticophis lateralis	Striped Racer	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Masticophis schotti	Schott's Whipsnake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Masticophis taeniatus	Striped Whipsnake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Micruroides euryxanthus	Sonoran Coral Snake	SP, SU, AU	D [pf], C	D [pf], C	D [pf], C	D [pf], C, MR	
Micrurus tener	Texas Coral Snake	SP, AU	D [pf], C	D [pf], C	D [pf], C	D [pf], C, MR	
Nerodia clarkii	Saltmarsh Watersnake	SP, SU, AU	V	V	V	V [ce]	
Nerodia cyclopion	Mississippi Green Watersnake	SU	V [bk], V	V [bk], V, T [mt]	V [bk], V, T [mt]	MR	
Nerodia erythrogaster	Plain-bellied Watersnake	SP, SU, AU	V [bk], V	V [bk], V	V [bk], V	MR, TR [rt]	
Nerodia fasciata	Southern Watersnake	SP, SU	V [bk], V [r]	V [bk], V, T [mt]	V [bk], V, T [mt]	T [mt], MR	
Nerodia harteri	Brazos River Watersnake	SP, SU	V, V [r]	T [mt], V, V [r]	V, V [r]	T [mt], MR	
Nerodia paucimaculata	Concho Watersnake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	V [r], MR, C	
Nerodia rhombifer	Diamond-backed Watersnake	SU	V [bk], V	V [bk], V	V [bk], V	MR	
Nerodia sipedon	Northern Watersnake	SP, SU, AU	V [bk], V [r]	V [bk], V, T [mt]	V [bk], V, T [mt]	T [mt], MR	
Opheodrys aestivus	Rough Green Snake	SP, SU	V, V [r]	V, V [r]	V, V [r]	MR	
Opheodrys vernalis	Smooth Green Snake	SP, SU, AU	C	C	C	MR, C	
Oxybelis aeneus	Brown Vine Snake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Phyllorhynchus browni	Saddled Leaf-nosed Snake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Phyllorhynchus decurtatus	Spotted Leaf-nosed Snake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	V [r], V, C, MR	
Pituophis catenifer	Gopher Snake	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Pituophis ruthveni	Louisiana Pine Snake	SP, SU	V [r], V	V [r], C, D [ft]	V [r], C	D [ft], MR	2
Regina grahamii	Graham's Crayfish Snake	SP, SU	T [mt]	T [mt]	T [mt]	T [mt], MR	
Regina rigida	Glossy Crayfish Snake	SP, SU	T [mt]	T [mt]	T [mt]	T [mt], MR	
Rhinocheilus lecontei	Long-nosed Snake	SP, SU, AU	V [r], D [ft]	V [r], D [ft]	V [r], D [ft]	MR	
Salvadora grahamiae	Eastern Patch-nosed Snake	SP, SU, AU	V [r], D [ft]	V [r], D [ft]	V [r], D [ft]	MR	
Salvadora hexalepis	Western Patch-nosed Snake	SP, SU, AU	V [r], D [ft]	V [r], D [ft]	V [r], D [ft]	MR	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Sistrurus catenatus	Massasauga	SP, SU, AU	V, V r, C	D ft, C	D ft, C	MR, TR rt	
Sistrurus miliarius	Pygmy Rattlesnake	SU	V r	V r	V r	MR, TR rt	
Sonora semiannulata	Western Ground Snake	SP, SU, AU	C, V r	C	C	MR	
Storeria dekayi	Dekay's Brown Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, D pf	
Storeria occipitomaculata	Red-bellied Snake	SP, SU, AU	C w	C w, D pf	C w	MR, D pf	
Tantilla atriceps	Mexican Black-headed Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, C, D pf	
Tantilla cucullata	Trans-Pecos Black-headed Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, C, D pf	
Tantilla gracilis	Flat-headed Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, C, D pf	
Tantilla hobartsmithi	Smith's Black-headed Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, C, D pf	
Tantilla nigriceps	Plains Black-headed Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, C, D pf	
Tantilla planiceps	Western Black-headed Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, C, D pf	
Tantilla wilcoxi	Chihuahuan Black-headed Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, D pf	
Tantilla yaquia	Yaqui Black-headed Snake	SP, SU, AU	V r, C	C, D pf	V r, C	MR, D pf	
Thamnophis atratus	Aquatic Garter Snake	SP, SU, AU	V, V r, C	D ft	V, V r, C	MR	
Thamnophis couchii	Sierra Garter Snake	SP, SU, AU	V, C, V r	V, C, V r	V, C, V r	V, C, V r	
Thamnophis cyrtopsis	Black-necked Garter Snake	SP, SU, AU	V, D ft	V, D ft	V, D ft	MR	13
Thamnophis elegans	Terrestrial Garter Snake	SP, SU, AU	V, V r, C	D ft	V, V r, C	V ce, MR	
Thamnophis eques	Mexican Garter Snake	SP, SU, AU	V, V r, C	D ft	V, V r, C	V ce, MR	
Thamnophis gigas	Giant Garter Snake	SP, SU, AU	V, V r, C	V, D ft	V, V r, C	V ce, MR	
Thamnophis hammondii	Two-striped Garter Snake	SP, SU, AU	V, V r, C	V, D ft	V, V r, C	V ce, MR	
Thamnophis marcianus	Checkered Garter Snake	SP, SU, AU	V, V r, C	V r, D ft	V r, D ft	V ce, MR	
Thamnophis ordinoides	Northwestern Garter Snake	SP, SU, AU	V, V r, C	D ft	V, V r, C	V ce, MR	
Thamnophis proximus	Western Ribbon Snake	SP, SU, AU	V, V r, C	D ft	V , V r, C	V ce, MR	
Thamnophis radix	Plains Garter Snake	SP, SU, AU	V, V r, C	V r, D ft	V, V r, C	V ce, MR	
Thamnophis rufipunctatus	Narrow-headed Garter Snake	SP, SU, AU	V, C	V, C	V, C	V ce, MR	
Thamnophis sirtalis	Common Garter Snake	SP, SU, AU	V, C, V r	V, C, V r	V, C, V r	MR, V ce, C	
Trimorphodon biscutatus	Western Lyre Snake	SP, SU, AU	V, C, V r	V, C, V r	V, C, V r	MR, V ce, C	14
Trimorphodon vilkinsonii	Chihuahuan Lyre Snake	SP, SU, AU	V, C, V r	V, C, V r	V, C, V r	MR, V ce, C	
Tropidoclonion lineatum	Lined Snake	SP, SU, AU	V, C, V r	V, C, V r	V, C, V r	MR, V ce, C	
Virginia striatula	Rough Earth Snake	SP, SU, AU	C	C	C	C	
Virginia valeriae	Smooth Earth Snake	SP, SU, AU	C	C	C	C	
NORTHWEST REGION							
Charina bottae	Northern Rubber Boa	SP	V, V r, C	V, V r, C	V, V r, C	MR, V r, C	
Coluber constrictor	North American Racer	SP, SU, AU	C, V r	C, V r	C, V r	MR, V ce, C, V r, D ft	
Contia tenuis	Sharp-tailed Snake	SP, SU, AU	V ce, D pf, C	V ce, D pf, C	V ce, D pf, C	MR, V ce, D pf, C	
Crotalus oreganus	Western Rattlesnake	SP, SU, AU	D ft; V, C, V r	D ft; V, C, V r	D ft; V, C, V r	MR, D ft, V r	
Crotalus viridis	Prairie Rattlesnake	SP, SU, AU	V, V r, DF ft	V, V r, DF ft	V, V r, DF ft	MR, V r, DF ft	
Diadophis punctatus	Ring-necked Snake	SP, SU, AU	C	C, D pf	C	C, MR	
Heterodon nasicus	Plains Hog-nosed Snake	SP, AU	V r, V, C	V r, V, C	V r, V, C	V r, MR, C	

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Hypsiglena torquata	Desert Night Snake	SP, SU, AU	V [r], V, C	V [r], V, C, D [pf]	V [r], D [pf]	D [pf], MR	
Lampropeltis getula	Common Kingsnake	SP, SU, AU	V [r], V, C	V [r], D [pf], C, MR	V [r],V, D [pf], C	V [ce], V [r], MR	
Lampropeltis g. californiae	California Kingsnake	SP, SU, AU	V [r], V, C	V [r], D [pf], C, MR	V [r],V, D [pf], C	V [ce], V [r], MR	
Lampropeltis pyromelana	Sonoran Mountain Kingsnake	SP, SU, AU	V [r], V, C	V [r], D [pf], C, MR	V [r],V, D [pf], C	V [ce], V [r], MR	
Lampropeltis triangulum	Milk Snake	SP, SU, AU	V [r], V, C	V [r], D [pf], C, MR	V [r],V, D [pf], C	V [ce], V [r], MR	
Lampropeltis zonata	California Mountain Kingsnake	SP, SU, AU	V [r], V, C	V [r], D [pf], C, MR	V [r], D [pf], C	V [ce], V [r], MR	
Opheodrys vernalis	Smooth Green Snake	SP, SU, AU	C	C	C	MR, C	
Masticophis lateralis	Striped Racer	SP, SU, AU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Masticophis taeniatus	Striped Whipsnake	SP, SU, AU	V [r], V, C	V [r], V, C, D [pf]	V [r], V, C, D [pf]	MR, D [ft]	
Nerodia fasciata	Southern Watersnake	SP, SU	V [bk], V [r]	V [bk], V, T [mt]	V [bk], V, T [mt]	T [mt], MR	
Nerodia rhombifer	Diamond-backed Watersnake	SP, SU	V [bk], V	V [bk], V	V [bk], V	MR	
Pituophis catenifer	Gopher Snake	SP, SU	V [r], V, C	V [r], V, C	V [r], V, C	MR, D [ft]	
Pituophis c. catenifer	Pacific Gopher Snake	SP, SU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Pituophis c. deserticola	Great Basin Gopher Snake	SP, SU	V [r], V, C	V [r], V, C	V [r], V, C	MR	
Rhinocheilus lecontei	Long-nosed Snake	SP, SU, AU	V [r], D [ft]	V [r], D [ft]	V [r], D [ft]	MR, D [ft]	
Salvadora hexalepis	Western Patch-nosed Snake	SP, SU, AU	V [r], D [ft]	V [r], D [ft]	V [r], D [ft]	MR	
Salvadora h. virgultea	Coast Patchnose Snake	SP, SU, AU	V [r], D [ft]	V [r], D [ft]	V [r], D [ft]	MR	
Sonora semiannulata	Western Ground Snake	SP, SU, AU	C, V [r]	C	C	MR, D [ft]	
Thamnophis atratus	Aquatic Garter Snake	SP, SU, AU	V, V [r], C	D [ft]	V, V [r], C	MR	
Thamnophis couchii	Sierra Garter Snake	SP, SU, AU	V, C, V [r]	V, C, V [r]	V, C, V [r]	V, C, V [r]	
Thamnophis elegans	Terrestrial Garter Snake	SP, SU, AU	V, V [r], C	D [ft]	V, V [r], C	V [ce], MR, D [ft]	
Thamnophis gigas	Giant Garter Snake	SP, SU, AU	V, V [r], C	V, D [ft]	V, V [r], C	V [ce], MR	
Thamnophis ordinoides	Northwestern Garter Snake	SP, SU, AU	V, V [r], C	V [r], D [ft]	V [r], D [ft]	V [ce], MR	
Thamnophis radix	Plains Garter Snake	SP, SU, AU	V, V [r], C	V [r], D [ft]	V, V [r], C	V [ce], MR	
Thamnophis sirtalis	Common Garter Snake	SP, SU, AU	V, C, V [r]	V, C, V [r]	V, C, V [r]	MR, V [ce], C	
MIDWEST REGION							
Agkistrodon contortrix	Copperhead	SP, SU, AU	V, V [r]	V, V [r]	V, V [r]	V, V [r]	
Agkistrodon piscivorus	Cottonmouth	SP, SU, AU	V, V [r]	V, V [r]	V, V [r]	V, V [r]	15
Arizona elegans	Glossy Snake	SP, SU, AU	V, C, V [r]	V, C, V [r]	V, C, V [r]	V, C, V [r]	
Carphophis amoenus	Eastern Worm Snake	SP, SU, AU	C [w]	C [w]	C [w]	D [pf], C [w]	16
Carphophis vermis	Western Worm Snake	SP, SU, AU	C [w]	C [w]	C [w]	D [pf], C [w]	16
Cemophora coccinea	Scarlet Snake	SP, SU, AU	V [r], C	C, D [pf]	C, V [r]	MR, V [r]	
Clonophis kirtlandii	Kirtland's Snake	SP, SU, AU	C	C	C	C	17
Coluber constrictor	North American Racer	SP, SU, AU	C, V [r]	C, V [r]	C, V [r]	MR, V [ce], C, V [r]	
Crotalus horridus	Timber Rattlesnake	SP, SU, AU	D [ft], V, C, V [r]	D [ft], V, C, V [r]	D [ft], V, C, V [r]	MR, D [ft], V [r]	
Crotalus viridis	Prairie Rattlesnake	SP, SU, AU	V, V [r], DF [ft]	V, V [r], DF [ft]	V, V [r], DF [ft]	MR, V [r], DF [ft]	
Diadophis punctatus	Ring-necked Snake	SP, SU, AU	C	C, D [pf]	C	C, MR	18
Elaphe obsoleta	Eastern Rat Snake	SP, SU, AU	V [r], C	V [r], C	V [r], C	MR, D [ft]	
Elaphe emoryi	Great Plains Rat Snake	SP, SU, AU	C, V [r]	C, V [r]	C, V [r]	MR, V [ce]	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

TABLE 5-1: SPECIES X TECHNIQUES TABLE

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Elaphe gloydi	Eastern Fox Snake	SP, SU, AU	C, V r	C, V r	C, V r	MR, V ce	
Elaphe guttata	Red Corn Snake	SP, SU, AU	V r, C	V r, C	V r, C	MR, D ft	
Elaphe spiloides	Gray Rat Snake	SP, SU, AU	V r, C	V r, C	V r, C	MR, D ft	19
Elaphe vulpina	Western Fox Snake	SP, SU, AU	V r, C	V r, C	V r, C	MR, D ft	
Farancia abacura	Red-bellied Mud Snake	SP, SU, AU	V r, T mt	T mt, T fy, SN, C	T mt, T fy, SN, C	MR	20
Heterodon gloydi	Dusty Hog-nosed Snake	SP, SU, AU	V r, V, C	V r, V, C	V r, V, C	V r, MR, C	
Heterodon nasicus	Plains Hog-nosed Snake	SP, SU, AU	V r, V, C	V r, V, C	V r, V, C	V r, MR, C	
Heterodon platirhinos	Eastern Hog-nosed Snake	SP, SU, AU	V r, V, C	V r, V, C	V r, V, C	V r, MR, C	21
Hypsiglena torquata	Chihuahuan Night Snake	YR	V r, V, C	V r, V, C, D pf	V r, D pf	D pf, MR	22
Lampropeltis calligaster	Yellow-bellied Kingsnake	SP, SU, AU	V r, V, C, D	V r, V, C, D	V r, V, C, D	MR	
Lampropeltis getula	Common Kingsnake	SP, SU, AU	V r, V, C	V r, D pf, C, MR	V r, D pf, C, MR	V ce, V r, MR	
Lampropeltis triangulum	Milk Snake	SP, SU, AU	V r, V, C	V r, D pf, C, MR	V r,V, D pf, C	V ce, V r, MR	
Leptotyphlops dulcis	Texas Thread Snake	SP, SU, AU	V r, C	C, D pf, V r	V r, C, D	D, C, MR	
Masticophis flagellum	Coachwhip	SP, SU, AU	V r, V, C	V r, V, C	V r, V, C	MR	
Nerodia cyclopion	Mississippi Green Watersnake	SP, SU, AU	V bk, V	V bk, V, T mt	V bk, V, T mt	MR	
Nerodia erythrogaster	Plain-bellied Watersnake	SP, SU, AU	V bk, V	V bk, V	V bk, V	MR, TR rt	
Nerodia fasciata	Southern Watersnake	SP, SU, AU	V bk, V r	V bk, V, T mt	V bk, V, T mt	T mt, MR	
Nerodia rhombifer	Diamond-backed Watersnake	SP, SU, AU	V bk, V	V bk, V	V bk, V	MR	
Nerodia sipedon	Northern Watersnake	SP, SU, AU	V bk, V r	V bk, V, T mt	V bk, V, T mt	T mt, MR	
Opheodrys aestivus	Rough Green Snake	SP, SU, AU	V, V r	V, V r	V, V r	MR	23
Opheodrys vernalis	Smooth Green Snake	SP, SU, AU	C	C	C	MR, C	
Pituophis catenifer	Gopher Snake	SP, SU, AU	V r, V, C	V r, V, C	V r, V, C	MR	
Regina grahamii	Graham's Crayfish Snake	SP, SU, AU	T mt	T mt	T mt	T mt, MR	24
Regina septemvittata	Queen Snake	SP, SU, AU	C, V bk	C, V bk	C, V bk	MR, V ce	25
Rhinocheilus lecontei	Long-nosed Snake	SP, SU, AU	V r, D ft	V r, D ft	V r, D ft	MR	
Sistrurus catenatus	Massasauga	SP, SU, AU	V, C	V, C	V, C	MR, TR rt	26
Sistrurus miliarius	Pygmy Rattlesnake	SU	V r	V r	V r	MR, TR rt	
Sonora semiannulata	Western Ground Snake	SP, SU, AU	C, V r	C	C	MR	
Storeria dekayi	Dekay's Brown Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, D pf	
Storeria occipitomaculata	Red-bellied Snake	SP, SU, AU	C w	C w, D pf	C w	MR, D pf	
Tantilla coronata	Southeastern Crowned Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, C, D pf	
Tantilla gracilis	Flat-headed Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, C, D pf	27
Tantilla nigriceps	Plains Black-headed Snake	SP, SU, AU	C, D pf	C, D pf	C, D pf	MR, C, D pf	
Thamnophis butleri	Butler's Garter Snake	SP, SU, AU	C, V	C, V	C, V	MR	
Thamnophis elegans	Terrestrial Garter Snake	SP, SU, AU	V, V r, C	D ft	V, V r, C	V ce, MR	
Thamnophis marcianus	Checkered Garter Snake	SP, SU, AU	V, V r, C	V r, D ft	V r, D ft	V ce, MR	
Thamnophis proximus	Western Ribbon Snake	SP, SU, AU	V, V r, C	V, V r, C	V, V r, C	V ce, MR	
Thamnophis radix	Plains Garter Snake	SP, SU, AU	V, V r, C	V r, D ft	V, V r, C	V ce, MR	
Thamnophis sauritus	Eastern Ribbon Snake	SP, SU, AU	V	C w	V	V ce, MR	28

Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
Thamnophis sirtalis	Common Garter Snake	SP, SU, AU	V, C, V [r]	V, C, V [r]	V, C, V [r]	MR, V [ce], C	
Tropidoclonion lineatum	Lined Snake	SP, SU, AU	V, C, V [r]	V, C, V [r]	V, C, V [r]	MR, V [ce], C	
Virginia striatula	Rough Earth Snake	SP, SU, AU	C	C	C	C	
Virginia valeriae	Smooth Earth Snake	SP, SU, AU	C	C	C	C	

TABLE 5-1: SPECIES X TECHNIQUES TABLE

CROCODILIANS							
Scientific Name	Common Name	Season	INVENTORY: Rapid Assessment	INVENTORY: Comprehensive Survey	MONITORING: Presence / Absence	MONITORING: Population Status	Comment Code
SOUTHEAST REGION							
Alligator mississippiensis	American Alligator	SP, SU, AU	V ey	V ey	V ey	MR, V n	
Crocodylus acutus	American Crocodile	YR	V ey	V ey	V ey	MR, V n	
SOUTHWEST REGION							
Alligator mississippiensis	American Alligator	SP, SU, AU	V ey	V ey	V ey	MR, V n	

Codes and Additional Information for the *Species x Techniques Table*:

1. **Seasons**: codes for each season of the year
2. **Life Stage**: key to codes for life stages
3. **Techniques**: key to codes for techniques referenced in table
4. **Comments:** key to comments, organized by taxa
5. **Taxonomic Synonyms:** provides information about official changes in scientific and common names of a given species (Crother et al. 2008)
6. **References for Techniques Table:** a list of books and articles referenced in the table, organized by PARC region

---

1. SEASONS: The seasons that a given species can be detected and/or studied are listed in this column in the *Species x Techniques Table*.

Code	Season
SP	Spring
SU	Summer
AU	Autumn
WI	Winter
YR	All Year

2. LIFE STAGE: Information is provided specific to the life stage level for many of the frogs and salamanders in the *Species x Techniques Table*. The codes used in the table for these life stages are shown below. Unless otherwise noted, all entries for snakes imply juveniles and adults, all entries for turtles imply hatchlings, juveniles, and adults, and all entries for lizards imply juveniles and adults.

Code	Life Stage
A	Adults
BA	Breeding Adults
PA	Paedomorphic Adults
J	Juveniles
M	Metamorphs
L	Larvae
T	Tadpoles
E	Eggs

3. TECHNIQUES: The codes and descriptions of all techniques referenced in the *Species x Techniques Table* are shown below.

Technique Name	Code	Description of Technique
Not Applicable	N/A	No technique known
Auditory survey (active)	A ^{a}	Listening for calls
Auditory survey (recording)	A ^{r}	Recording calls
Auditory survey (density estimate)	A ^{d}	Estimating density or number of calling individuals
Cover objects (general)	C	Searching under assorted cover objects
Cover (metal)	C ^{m}	Metal coverboards
Cover (wood)	C ^{w}	Wood coverboards
Cover (bark)	C ^{b}	Peeling bark from standing or down trees
Cover (rock)	C ^{r}	Searching under rocks and stones
Count	CT	Counting individuals or egg masses
Dip netting	DN	Sampling via dip netting
Drift fence	D	Surveying along a drift fence
Drift fence (aquatic)	D ^{aq}	Surveying along a drift fence in a wetland
Drift fence (pit falls)	D ^{pf}	A drift fence with bucket traps
Drift fence (funnel traps)	D ^{ft}	A drift fence with funnel traps
Female reproductive assessment	F	Assessing the reproductive status of captured females
Habitat assessment	H	Assessing the suitability of habitat for a focal species
Mark recapture	MR	Mark - recapture studies
Temporary Removal Study	R	Temporary removal of individuals to estimate abundance
Noodling	N	Hand capture of aquatic organisms in the riverine substrate, caves, or overhangs
Sand sifting	SF	Straining sand
Seining	SN	Pulling a seine net through aquatic habitat
Sweep sampling	SS	Sweeping a net through aquatic habitat
Tracking (radio telemetry)	TR ^{rt}	Radio telemetry
Tracking (thread)	TR ^{t}	Tracking with the use of thread bobbins
Trapping (basking trap)	T ^{bk}	self explanatory
Trapping (crawfish traps)	T ^{cr}	self explanatory
Trapping (fishing hook and line)	T ^{hl}	self explanatory
Trapping (funnel traps aquatic)	T ^{fa}	self explanatory

Technique Name	Code	Description of Technique
Trapping (fyke nets)	T fy	Hoop nets with seine (or lead nets) attached
Trapping (goin dredge)	T gd	self explanatory
Trapping (hoop nets)	T h	self explanatory
Trapping (leaf bags)	T lb	self explanatory
Trapping (minnow traps)	T mt	self explanatory
Trapping (trammel net)	T tn	self explanatory
Trapping (pvc pipes)	T pv	Plastic (PVC) pipe sections installed on trees
Visual encounter surveys (general)	V	Actively searching an area
Visual encounter surveys (controlled effort)	V ce	Searching while quantifying effort (i.e. time, area)
Visual encounter surveys (basking)	V bk	Searching for basking individuals
Visual encounter surveys (burrows)	V b	Searching for individuals in or at the mouths of burrows (e.g., gopher tortoise or crayfish)
Visual encounter surveys (crawls)	V cr	Searching for sea turtle tracks on nesting beaches
Visual encounter surveys (electroshocking)	V es	Using electroshock fishing to sample aquatic habitats
Visual encounter surveys (eye shine)	V ey	Using lights to search for eye shine
Visual encounter surveys (hibernacula)	V h	Searching for individuals at hibernation sites
Visual encounter surveys (kick sampling)	V ks	Disturbing aquatic substrate to make organisms visible
Visual encounter surveys (road cruising)	V r	Driving or walking roads and surveying for animals
Visual encounter surveys (snorkeling)	V s	Snorkeling or scuba diving
Visual encounter surveys (strandings)	V st	Looking for dead or dying animals on the beach
Visual encounter surveys (viewbox)	V vb	Using a device to look underwater to survey aquatic organisms

4. COMMENTS: The codes for comments referenced in the *Species x Techniques Table* are organized by taxonomic group in the tables below.

FROGS AND TOADS: Comment Codes	
Code	Comments
1	Associated with gopher tortoise burrows
2	Sometimes will start calling again on rainy, cool nights in the fall
3	Explosive breeder, difficult to detect outside of breeding events
4	Fossorial, should be inventoried during breeding season
5	Unlikely to find eggs, direct development of eggs laid under rocks
6	Fast developing tadpoles limit windows of opportunity for monitoring
7	Eggs are single and difficult to inventory
8	Extremely fossorial--cannot be surveyed except during ideal breeding conditions
9	Terrestrial trapping efforts should concentrate around known breeding ponds
10	Probably not easily detected
11	NA--no free-living larval stage
12	Eggs are laid in burrows in foam nests and would be difficult to inventory
13	Electrofishing is also effective
14	Eye-shine night surveys (L. Diller, Green Diamond Corp., California: put light on nose, not forehead; look in vegetation as well as on ground)
15	Pitfall traps and drift fences are also effective but can incur mortality (some other species are highly vulnerable); this approach may be more relevant to research studies.
16	Pitfall traps and drift fences are also effective but can incur mortality (some other species are highly vulnerable); this approach may be more relevant to research studies. Small hoop nets may be effective.
17	Quiet calls are not always usable in surveys. Some populations or individual toads may not call. Pitfall traps and drift fences are also effective but can incur mortality (some other species are highly vulnerable); this approach may be more relevant to research studies. Nocturnal surveys. Radio-tracking a research technique.
18	Pitfall traps and drift fences are also effective but can incur mortality (some other species are highly vulnerable); this approach may be more relevant to research studies. PVC pipes may be considered
19	Formerly *P. regilla*, Use range to distinguish species. PVC pipes may be considered.
20	Can be found at edges of streams & in wetlands, especially in summer
21	Quiet calls not always usable in surveys
22	Aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis).
23	Auditory surveys (inc. automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset; Shoreline searches at breeding ponds are also productive, counting frogs as they hop into the water.
24	Eggs are readily observed along shorelines, typically entwined in emergent vegetation. Where other toad species are present identification to species can be a problem. Egg stage is of short duration (3-12 days) and egg surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
25	Tadpoles are easily observed in visual searches, and readily captured using dip-nets, fine-mesh minnow traps, or other improvised aquatic funnel traps in shallows near shorelines. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis). Where other toad species are present tadpole identification to species can be a problem.

FROGS AND TOADS: Comment Codes	
Code	Comments
26	Spring sampling most effective (while breeding). Effective summer and autumn sampling limited to visual search and trapping techniques, with detection probabilities much higher during and immediately following rain events. Night road cruising during and immediately after rains is effective in summer. Moist artificial cover is used, and cover should be checked during daytime, as toads forage at night. Breeding is explosive or of short-duration usually on warm days in spring. Breeding choruses are often concentrated and in smaller depressions or pools.
27	Where other toad species are present egg identification to species can be a problem. Egg stage is of short duration (normally 2-7 days) and surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
28	Tadpoles can be observed in visual searches, and readily captured using dip-nets, fine-mesh minnow traps, or other improvised aquatic funnel traps. Where other toad species are present tadpole identification to species can be a problem. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis).
29	All sampling should be during breeding and during rain events. Late spring, early summer rains trigger explosive breeding.
30	Eggs are poorly known and identification to species should be verified by other life stages. Egg stage is of short duration (1-6 days) and surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
31	Tadpoles are easily observed in visual searches, and readily captured using dip-nets, fine-mesh minnow traps, or other improvised aquatic funnel traps. Where other toad species are present tadpole identification to species can be a problem. Surveys for larvae are valid only when larvae are present (between egg hatching and metamorphosis).
32	Eggs are readily observed along shorelines, typically entwined in emergent vegetation. Where other toad species are present identification to species can be a problem. Egg stage is of short duration (about 7 days) and egg surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
33	Tadpoles are easily observed in visual searches, and readily captured using dip-nets, fine-mesh minnow traps, or 2L pop bottle funnel traps in shallows near shorelines. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis). Where other toad species are present tadpole identification to species can be a problem.
34	Spring sampling most effective (while breeding). Effective summer and autumn sampling limited to visual search and trapping techniques, with detection probabilities much higher during and immediately following rain events. Night road cruising during and immediately after rains is effective in summer. Moist artificial cover is used, and cover should be checked during daytime, as toads forage at night. Breeding is explosive or of short-duration usually on warm days in spring. Breeding choruses are often concentrated and in smaller depressions or pools.
35	Eggs are readily observed along shorelines, typically entwined in emergent vegetation. Where other toad species are present identification to species can be a problem. Egg stage is of short duration (about 5 days) and egg surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
36	Tadpoles can be observed in visual searches, and captured using dip-nets, fine-mesh minnow traps, or other improvised aquatic funnel traps. Where other toad species are present tadpole identification to species can be a problem. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis).
37	Where other toad species are present egg identification to species can be a problem. Egg stage is of short duration and surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.

FROGS AND TOADS: Comment Codes	
Code	Comments
38	All sampling should be during breeding and during rain events. Late spring, early summer rains often trigger breeding, but breeding occur on rainless, warm, spring nights as well. Familiarity with local breeding habits important.
39	Effective sampling techniques for this species warrant further investigation. Tadpoles can be captured using dip-nets, fine-mesh minnow traps, or SNs. Surveys for larvae are valid only during the short larval period (6-10 days), so should be used as a supplement to surveys for adults. Tadpole identification to species can be a problem.
40	Effective sampling techniques for this species warrant further investigation.
41	All sampling should be during breeding and during or immediately following rain events. Late spring, early summer rains trigger explosive breeding. Eggs hatch in 2 days so eggs are not a good life stage for surveys.
42	Aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis), and identification of larvae to species may be problematic.
43	Auditory surveys (including automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset. Visual search methods should be confined to active breeding times and areas. Polyvinyl chloride (PVC) pipe surveys can be effective post-breeding.
44	Aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis), and identification of larvae to species may be problematic.
45	Auditory surveys (including automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset. Visual search methods should be confined to active breeding times and areas. Polyvinyl chloride (PVC) pipe surveys, and nocturnal road cruising during or immediately following rains, can be effective post-breeding.
46	Aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis), and identification of larvae to species may be problematic.
47	Auditory surveys (including automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset. Visual search methods should be confined to active breeding times and areas. Polyvinyl chloride (PVC) pipe surveys, and nocturnal road cruising during or immediately following rains, can be effective post-breeding.
48	Aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis), and identification of larvae to species may be problematic.
49	Auditory surveys (including automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset. Visual search methods should be confined to active breeding times and areas. Polyvinyl chloride (PVC) pipe surveys, and nocturnal road cruising during or immediately following rains, can be effective post-breeding.
50	Aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis), and identification of larvae to species may be problematic.
51	Auditory surveys (including automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset.

FROGS AND TOADS: Comment Codes	
Code	Comments
52	Aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis), and identification of larvae to species may be problematic.
53	Auditory surveys (including automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset.
54	Fine mesh aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis), and identification of larvae to species may be problematic.
55	Auditory surveys (including automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset. Recent metamorphs can be observed along muddy shorelines of receding breeding depressions and can be quantified by counting individuals using time- or area-constrained VES. Recent metamorphs as well as adults often found under cover-boards along shorelines.
56	Aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis), and identification of larvae to species may be problematic.
57	Auditory surveys (including automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset.
58	Aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis), and identification of larvae to species may be problematic.
59	Auditory surveys (including automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset.
60	Aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis), and identification of larvae to species may be problematic.
61	Auditory surveys (including automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset.
62	Aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis), and identification of larvae to species may be problematic.
63	Auditory surveys (including automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset.
64	Fine mesh aquatic funnel traps are probably most effective and easiest to standardize for effort. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis), and identification of larvae to species may be problematic.

FROGS AND TOADS: Comment Codes	
Code	Comments
65	Auditory surveys (including automated recording systems) may be the most efficient means of detecting this species, but are limited to the calling season and should be conducted in darkness shortly after sunset. Recent metamorphs can be observed along muddy shorelines of receding breeding depressions and can be quantified by counting individuals using time- or area-constrained VES. Recent metamorphs as well as adults often found under cover-boards along shorelines.
66	Eggs are readily observed but identification to species can be a problem. Egg stage is of short duration (7-15 days) and egg surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
67	Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis). Tadpole identification to species can be a problem. Genetic analysis using larval tissue samples may be effective for identification.
68	Visual searches and trapping should be at breeding sites and during breeding season. Playback auditory surveys deserve testing. Research into effective survey techniques needed. Breeding appears to be correlated with warmer spring rains.
69	Eggs are readily observed but identification to species can be a problem. Egg stage is of short duration (3-21 days) and egg surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
70	Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis). Tadpole identification to species can be a problem.
71	Visual searches, cover objects, and trapping should be at breeding sites and during breeding season. Cover boards along shoreline are also effective at capturing recent metamorphs leaving breeding ponds.
72	Eggs are readily observed but identification to species can be a problem. Egg stage is of short duration (3-5 days) and egg surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
73	Larval surveys are valid year-round. Tadpole identification to species can be a problem.
74	Visual searches should be along shorelines. Fish landing nets and cane-pole with jig can be used to capture adults and larger juveniles.
75	Eggs are readily observed but identification to species can be a problem. Egg stage is of short duration (a few days) and egg surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
76	Aquatic funnel traps very effective for larvae. Tadpole identification to species can be a problem.
77	Visual searches should be along shorelines. Fish landing nets and cane-pole with jig can be used to capture adults and larger juveniles. Night road cruising during or immediately after rains effective in summer, especially for recent metamorphs.
78	Eggs are readily observed but identification to species can be a problem. Egg stage is of short duration (10-24 days) and egg surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
79	Tadpole identification to species can be a problem.
80	Visual searches should be along shorelines.
81	Eggs are readily observed but identification to species can be a problem. Egg stage is of short duration (2-17 days) and egg surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
82	Tadpole identification to species can be a problem.

FROGS AND TOADS: Comment Codes	
Code	Comments
83	Visual searches should be along shorelines. Cover boards along shoreline may be effective at detecting recent metamorphs leaving breeding ponds.
84	Eggs are readily observed but identification to species can be a problem. Egg stage is of short duration (time to hatching unknown) and egg surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
85	Tadpole identification to species can be a problem.
86	Visual searches should be along shorelines. Auditory surveys should include afternoon and early morning (midnight - 3 AM) samples, as well as the usual just after dark period.
87	Eggs are readily observed but identification to species can be a problem. Egg stage is of short duration (3-15 days) and egg surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
88	Tadpole identification to species can be a problem.
89	Visual searches should be along shorelines.
90	The large eggs are readily observed and usually identification to species is not a problem. Egg stage is of relatively longer duration (typically 2-3 weeks) and egg surveys are valid only during this period.
91	Tadpole identification to species can be a problem.
92	Visual searches should be at breeding ponds during breeding period.
93	Egg stage is of short duration (1-15 days) and egg surveys are valid only during this period. Egg surveys should be used only to supplement surveys for other life stages.
94	Tadpoles can be captured using dip-nets, fine-mesh minnow traps, or other improvised aquatic funnel traps. Tadpole identification to species can be a problem. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis).
95	All sampling should be during breeding and during or immediately following rain events. Late spring, early summer rains trigger explosive breeding.
96	Tadpoles can be captured using dip-nets, fine-mesh minnow traps, or other improvised aquatic funnel traps. Tadpole identification to species can be a problem. Surveys for larvae are valid only during the larval period (between egg hatching and metamorphosis).
97	All sampling should be during breeding and during or immediately following rain events. Late spring, early summer rains trigger explosive breeding. The short duration of the egg stage (a few days) makes egg surveys unreliable.
98	Tadpoles will school
99	Adults and tadpoles of the two grey treefrog species cannot be visually distinguished; need to listen to breeding calls
100	No free-living larval stage
101	Species is present all year, but is more efficient and effective to survey during low flows.
102	A drift fence with pitfall traps may not be very effective for this species; they may be hard to catch and may climb out of buckets.

SALAMANDERS: Comment Codes	
Code	Comments
1	Breed in Sept and Oct after rains following drought
2	Rarely seen outside of breeding season
3	An early breeder. Start surveys in November or December and continue through January and February. Best results during rain events.
4	Rarely found, but possible (@ base of wire grass--difficult!). Train dogs to look for eggs??
5	Citation that tests dipnetting vs minnow trapping, winter is best, in spring early is better, not June

SALAMANDERS: Comment Codes	
Code	Comments
6	Pitfall trap good technique, but water table often floods buckets or pushes buckets out of ground.
7	Single eggs in SC not easily surveyed, but egg clusters laid in TN
8	Possibility of paedomorphs
9	Females will deposit eggs in minnow traps
10	Rarely seen outside of breeding season
11	An early breeder. Conduct surveys in January and February. Best results during rain events.
12	Prairie dog burrows
13	Eel pots?
14	Most terrestrial of the *Desmognathus*
15	In caves
16	Horizontal cracks near caves
17	Forests and caves
18	Flip rocks in stream in fall/winter. Search in one direction while surveying cave.
19	Eggs in sphagnum moss
20	Short larval period
21	see *Siren* and *Pseudobranchus* papers for more detail on dredge technique
22	Bait traps with glow stick
23	Rock outcroppings
24	March
25	Weathered talus
26	Rainy nights
27	Commercial crayfish traps have been shown to be more successful than minnow traps in capturing Amphiumas.
28	Rarely seen outside of breeding season
29	An early breeder. Start surveys in November or December and continue through January and February. Best results during rain events.
30	May be found in caves
31	Very difficult to find outside of breeding season
32	Search for egg masses in breeding sites.
33	Night surveys of breeding sites in late winter
34	Very difficult to find outside of breeding season
35	Very difficult to find outside of breeding season
36	Females on nest during October
37	Larvae overwinter and can be easily captured in minnow traps.
38	Watch for mating adults in dried ephemeral pools with black willow in late September and early October.
39	Single eggs in SC not easily surveyed, but egg clusters laid in TN.…
40	Possibility of paedomorphs
41	Females will deposit eggs in minnow traps
42	Rarely seen outside of breeding season
43	An early breeder. Conduct surveys in January and February. Best results during rain events.
44	Commercial crayfish traps have been shown to be more successful thans minnow traps in capturing Amphiumas.
45	Best results in April, May, and October. TK Pauley

SALAMANDERS: Comment Codes	
Code	Comments
46	Williams, R.D., J.E. Gates, and C.H. Hocutt. 1981. An evaluation of known and potential sampling techniques for Hellbender, *Cryptobranchus alleghaniensis*. Journal of Herpetology 15(1): 23-27. THESE AUTHORS SUGGEST ELECTRO-SHOCKING IN CONJUNCTION WITH DIPNET OR SEINE SET DOWNSTREAM OF THE ROCK SHOCKED. Humphries, W.J. and T.K. Pauley. 2005. Life history of the Hellbender, *Cryptobranchus alleghaniensis*, in a West Virginia stream. Am. Midl. Nat. 154: 135-142. THESE AUTHORS USED NOCTURNAL SURVEYS WITH HEADLAMPS TO FIND HELLBENDERS WITHOUT MOVING ROCKS.
47	May last a few days to a few weeks
48	Most terrestrial of the *Desmognathus*
49	Eggs are deposited in grape-like clusters in depressions under logs, rocks, leaf litter or moss mats which are close to streams or seeps.
50	In Virginia, it is confined to the mountainous, forested habitats in the Blue Ridge Province from Mt. Rogers and Floyd County.
51	Nocturnal surveys are most effective
52	The eggs are laid in small cavities amid the rocks of spring seeps in pockets of gravel and mud.
53	This salamander inhabits streams, stream banks, and seeps and is generally found in seeps, which are used for nesting and hibernation. This species lives in moss and leaf litter on the forest floor. This species occurs in the spruce-fir forest of Whitetop Mountain, Mount Rogers, and Pine Mountain, where Grayson, Washington, and Smyth counties meet. The entire Virginia range occurs within the Mount Rogers National Recreation Area. On foggy, rainy nights they may be found up to 7 feet above ground level on tree trunks. The greatest abundance are in the fraser fir-red spruce forest. They are present but not as common below the spruce-fir forest.
54	Can be found during summer above 3,500 ft.
55	Adults frequently found in dry habitats such as strip mines, pastures, etc. especially under logs.
56	Look for cave crickets
57	Courtship occurs in the fall and eggs are laid in the winter in running water.
58	This salamander inhabits areas near springs, seepages, and streams in hardwood forests and swamps. They are most numerous in the mountains.
59	Habitat is variable throughout the Appalachian Mountains, but is closely associated with the Appalachian uplift
60	Visual searches in caves
61	Cypress knees--search sphagnum on top; under leaves near water.
62	Short larval period
63	Can be difficult to find outside of breeding season
64	Adults are most active during the winter months (Petranka 1998)
65	Adults are most active during the winter months (Petranka 1998)
66	Will move to temporary pools (including road ruts) during summer (nonbreeding season)
67	Primarily found in and around ephemeral wetlands
68	Nocturnal surveys after rain are very effective
69	Inhabits hardwood forests and is active on the surface at night when the humidity is high and temperatures are mild. During the day it occupies burrows under stones and logs, or tunnels formed by rotted roots. This species is normally found above 3000 feet.
70	Nocturnal surveys after rain are very effective. May also occur in deciduous forests at proper elevations.
71	Nocturnal surveys after rain are very effective. This is particularly effective in summer

SALAMANDERS: Comment Codes	
Code	Comments
72	Talus. In general, the probability of detection for most terrestrial *plethodon* will be greater at night after or during rain events in the spring and fall. You need to also consider the area and shape of the sampling framework, frequency of repeat visits, number of sites, etc. (see *P. shenandoah* as an example). I would suggest though that the techniques for the inventorying and monitoring of any *plethodon* (and likely any species for that matter) should really be based on the goal and objectives of the study. An accurate rapid inventory, for example, may be hard to do if you have a limited timeframe. The detection of different species is related to season, technique, habitat conditions, etc. You will likely always be underestimating. Also, it has been shown that the detection needs to be factored into estimates of occupancy and abundance. I would just be cautious on how people go about conducting studies. In my opinion, there is no cookie cutter way to do it.
73	Associated with seepages near rock outcrops on slopes in mixed hardwood forests. It lives under leaf litter, rocks, or rotten logs. This species has never been found above 2500 feet elevation and usually occurs at elevations below 1100 feet.
74	Nocturnal surveys after rain are effective. This is particularly effective in summer above 3,500 ft. Only active on surface during summer above 3,500 ft..
75	Reaches northern-most extent of its range in southwest Virginia. Known from the higher elevations of only three Mountains: Whitetop, Mount Rogers, and Pine Mountain, at elevations from 4400 to 5729 feet in the Mount Rogers area. It is usually found in red spruce forests and not within 200 feet of water. Highest abundance seems to be correlated with abundant ground cover of rocks and downed wood. Rotting logs on the forest floor are known to be nest sites.
76	Eggs rarely found
77	Also found in fens and bogs.
78	Commercial crayfish traps have been shown to be more successful than minnow traps in capturing Amphiumas.
79	Virginia is the northern-most extent of its range. This salamander is widespread and abundant in the Coastal Plain south of the James River. It inhabits swamps and shallow cypress or gum ponds in savannahs. It is most abundant in pools, slow streams, and swampy woods.
80	Females on nest during October
81	Larvae overwinter and can be easily captured in minnow traps.
82	Watch for mating adults in dried ephemeral pools with black willow in late September and early October.
83	Single eggs in SC not easily surveyed, but egg clusters laid in TN….
84	Possibility of paedomorphs
85	Females will deposit eggs in minnow traps
86	Rarely seen outside of breeding season
87	An early breeder. Conduct surveys in January and February. Best results during rain events.
88	Down wood associate, arboreal tendencies becoming known and animal is found in red tree vole nests high in forest canopy
89	Apparent riparian association in interior dry portion of range
90	Requires summer monsoonal rains
91	Down wood associate, arboreal tendencies
92	Down wood associate, arboreal tendencies becoming known and animal has been found to 90 m in canopy
93	Neotenic
94	Neotenic; cave-dwelling
95	Completely subterranean; not seen since 1951

SALAMANDERS: Comment Codes	
Code	Comments
96	Rock associate
97	Eft stage is doubtful in the wild
98	Will move to temporary pools (including road ruts) during summer (nonbreeding season)
99	Stream bank surveys during stream monitoring.
100	Talus associate
101	Requires summer monsoonal rains
102	Small streams, seeps
103	Cover objects around pond perimeter. Pitfall traps and drift fences are also effective but can incur mortality (some other species are highly vulnerable); this approach may be more relevant to research studies.
104	Cover objects in forest or rainy periods. Pitfall traps and drift fences are also effective but can incur mortality (some other species are highly vulnerable); this approach may be more relevant to research studies.
105	Pitfall traps and drift fences are also effective but can incur mortality (some other species are highly vulnerable); this approach may be more relevant to research studies. Use flashlight/spotlight to illuminate the bottom of breeding ponds in the summer for larvae.
106	Warm, rainy nights. Pitfall traps and drift fences are also effective but can incur mortality (some other species are highly vulnerable); this approach may be more relevant to research studies. Use flashlight/spotlight to illuminate the bottom of breeding ponds in the spring for adults.
107	Persistent egg masses can be counted after hatching
108	Look under cover objects around pond perimeter. Pitfall traps and drift fences are also effective but can incur mortality (some other species are highly vulnerable); this approach may be more relevant to research studies.
109	Look under cover objects in forest or adjacent to ponds, or on warm, rainy nights. Pitfall traps and drift fences are also effective but can incur mortality (some other species are highly vulnerable); this approach may be more relevant to research studies.
110	Singly-oviposited eggs may be difficult to census
111	Use flashlight/spotlight to illuminate the bottom of breeding ponds in the summer for larvae.
112	Look under cover objects in forest or adjacent to ponds, or look on warm, rainy nights. Pitfall traps and drift fences are also effective but can incur mortality (some other species are highly vulnerable); this approach may be more relevant to research studies. Use flashlight/spotlight to illuminate the bottom of breeding ponds in the spring for adults.
113	Down wood associate, arboreal tendencies becoming known and animal is found in red tree vole nests high in forest canopy
114	Apparent riparian association in interior dry portion of range
115	Down wood associate
116	Adults can be found in upland forests or in/along streams. Night stream surveys likely also effective.
117	Most adults are paedomorphic. Night stream surveys. Terrestrial forms SP, AU, rainy nights
118	Adults can be found in upland forests or in/along streams. Night stream surveys also effective.
119	Down wood associate, road cruising has detected this species
120	Stream bank surveys during stream monitoring.
121	Also, stream bank surveys during stream monitoring, road cruising has detected this species.
122	Eggs are readily observed but identification to species may be problematic. Egg stage is of relatively longer duration (typically 3-4 weeks) and egg surveys are valid only during this period.

SALAMANDERS: Comment Codes	
Code	Comments
123	Larval identification to species can be a problem. The long larval period enables a long sampling window.
124	Visual searches should be at breeding ponds during breeding period. Visual surveys more productive at night and during or immediately following rains. Trapping surveys should include at least one rain event during the sampling period(s).
125	Eggs are readily observed but identification to species may be problematic. Egg stage is of unknown duration and egg surveys are valid only during the egg-laying and incubation period.
126	Larval identification to species can be a problem.
127	Visual searches and trapping should be at breeding ponds during breeding period. Visual surveys more productive at night and during or immediately following rains. Trapping surveys should include at least one rain event during the sampling period(s).
128	Visual searches should be at breeding ponds during breeding period. Visual surveys more productive at night and during or immediately following rains. Trapping surveys should include at least one rain event during the sampling period(s). Cover objects may be used post-breeding in terrestrial habitat as well but efficacy needs further testing. Night road cruises during rains sometimes productive, research needed into reliability.
129	Nests and eggs are readily discovered by observers familiar with microhabitat selection, but data on detection probabilities are needed.
130	Visual searches (for nests) should be at breeding ponds during breeding period. Trapping surveys should include at least one rain event during the sampling period(s).
131	Eggs are readily observed but identification to species may be problematic. Egg stage is of unknown duration and egg surveys are valid only during this period.
132	Eggs are readily observed but identification to species may be problematic. Egg stage is 2-8 weeks and egg surveys are valid only during this period.
133	Eggs are readily observed but identification to species may be problematic. Egg stage is of relatively longer duration (typically 2-4 weeks) and egg surveys are valid only during this period.
134	Survey and monitoring techniques poorly known.
135	Best results in April, May, and October. Visual searches return best results at night.
136	Survey and monitoring techniques poorly known. Generally reported as turning of flat rocks and electroshocking, with or without downstream netting. Various trapping techniques may merit investigation.
137	Survey and monitoring techniques poorly known. Generally reported as turning of flat rocks in stream. Various trapping techniques may merit investigation, such as aquatic and shoreline funnel and pitfall traps, cover objects, and leaf litter bags.
138	Survey and monitoring techniques poorly known. Various trapping techniques may merit investigation, such as aquatic funnel traps, cover objects, and leaf litter bags.
139	Survey and monitoring techniques poorly known. Generally reported as turning of flat rocks in stream and dip netting. Various trapping techniques may merit investigation, such as aquatic and terrestrial funnel and pitfall traps, cover objects, and leaf litter bags.
140	Egg masses found underneath & attached to undersides of cobbles or flagstones in shallow flowing water, or mini-channel pools, mid-May to mid-June.
141	Small, spring or seep-fed stream channels; VES, larvae found underneath, gently lifting cobbles and flagstones in shallow, flowing water, or mini-channel pools; Abundance quantified using Time-Constrained VES along 5-20m transects or Area-Constrained within a measured distance & width of channel section.

SALAMANDERS: Comment Codes	
Code	Comments
142	Small, spring or seep-fed stream channels; VES underneath cobbles and flagstones; Abundance quantified using Time-Constrained VES along 5-20m transects or Area-Constrained within a measured distance & width of channel section.
143	Survey and monitoring techniques poorly known. Various trapping techniques may merit investigation, such as aquatic and terrestrial funnel and pitfall traps, cover objects, and leaf litter bags.
144	Survey and monitoring techniques poorly known. Various trapping techniques may merit investigation, such as funnel and pitfall traps.
145	Survey and monitoring techniques poorly known. Various trapping techniques may merit investigation, such as aquatic funnel traps and leaf litter bags.
146	Nest searches very effective if surveyor is familiar with microhabitat requirements.
147	Surveys for larvae likely not very productive, but not well studied.
148	Nest searches are preferred method. Efficacy of trapping adults or detection with cover objects or visual surveys lower but not well known.
149	Can sample year-round. Detection probabilities needed for baited minnow traps, which may be the best detection method.
150	Visual searches most productive at breeding ponds during breeding period, at night, and during or immediately following rains. Trapping surveys effective over long sampling period in permanent ponds and lakes. Cover objects may be used post-breeding in terrestrial habitat as well but efficacy needs further testing. Night road cruises during rains sometimes productive, research needed into reliability.
151	Detection probabilities needed. Sampling most productive April, May, October. Nocturnal visual searches more effective.
152	Little is known about sampling techniques for this species.
153	Breeding adults are occasionally found under cover objects in dry pond basins in autumn
154	In sandstone and limestone rock outcrops, look on blocky bark of big hardwood trees
155	The best way to establish presence-absence of a population for *E. aquatica* is sampling larvae; seasonal effects and site idiosyncrasies limit ability to find adults. Specifically, dip-netting watercress; shaking mats of aquatic vegetation into dipnet. I have no data for summer, but presumably larvae are still abundant in springs at this time
156	Species existence is based on two museum specimens; Life history unknown. Techniques recommendations impossible to provide with confidence
157	Most often found in first-order streams, in areas of low flow or in leaf litter around stream margin. Adults best found in March; larvae year-round.
158	Road cruising is effective in spring and autumn at night.
159	Species is present all year, but is more efficient and effective to survey when water level is low.

SNAKES: Comment Codes	
Code	Comments
1	Steel minnow traps work better than plastic minnow traps because they have larger openings
2	Associated with pocket gopher colonies
3	Along crayfish streams
4	Very cryptic, rarely seen
5	Search wetland edges
6	Very difficult species to find via VES, survey efforts can very easily produce false-negatives for presence absence

SNAKES: Comment Codes	
Code	Comments
7	Unlike montane populations, the Coastal Plain population does not have communal dens or hibernacula. Only 1 or 2 snakes share an overwintering site
8	Most often encountered in rocky canyons and washes
9	In areas without talus they can be found on partially wooded slopes, often near shrubs and boulders. Active during the day
10	Specimens not confirmed for New Mexico but collected on AZ/NM border
11	Often found along talus slopes of partially wooded canyons and washes. Dusk is a good time to search
12	Peak IL-MI activity appears to be June and Sept (juveniles)
13	Easily Detected After Monsoon Rains in Canyons and Washes. In and Around water
14	Telemetry is Difficult
15	Nocturnal spotlighting beaver lodges & drying pools is also productive.
16	Surface activity often associated with rains.
17	Cover boards/corrugated tin must be placed over crayfish burrows for maximum effectiveness
18	Driftless areas/hill prairies, woodland glades; seldom DOR. Peak activity appears to be May in IL and MI
20	For best results set 90+ minnow traps with sufficiently large funnel openings.
21	Peak IL-MI activity appears to be June and Sept (juveniles). In autumn listen for dry leaves rustling on visual surveys.
22	Rain increases snake activity.
23	Nocturnal visual search of vegetation.
24	Nocturnal shoreline searches.
25	Shoreline searches, day and night.
26	Visual surveys best during early spring emergence.
27	Driftless areas/hill prairies, talus slopes in SW IL; most success after heavy rains
28	Visual surveys along shorelines and aquatic vegetation beds. Cover objects used sparingly.

TURTLES: Comment Codes	
Code	Comments
1	Mid-Apr to Mid-June & Mid-Sept to Mid-Oct optimal (SE population).
2	Note that there are river system specific for each species
3	For hatchlings, visual surveys of shallow backwater areas
4	No nesting occurrences
5	Recovery of turtles with radios may be challenging
6	This is one of the few species of turtle that can be surveyed during every season. To reduce stress on turtles when marking and processing hibernating wood turtles, they should be kept in a bucket of cold water from the stream. See Bowen and Gillingham (2004) Region 9 Species Conservation Assessment for Wood Turtles
7	Mid-Apr to Mid-June & Mid-Sept to Mid-Oct optimal
8	A habitat generalist, occurring in both lakes and rivers, relative to other map turtles
9	In SW Virginia rivers
10	SP, SU, AU survey after a heavy rain
11	Canned sardines in soybean oil, or a chum bag filled with ground fish. For hatchlings, visual surveys of shallow backwater areas

TURTLES: Comment Codes	
Code	Comments
12	Line distance sampling is employed to monitor range wide populations of the Mojave desert tortoise
13	Now nest on Texas beaches
14	Road-cruising following monsoonal rains early morning and late afternoon
15	Visual surveys most productive during nesting, basking surveys in early spring.
16	Less effective than trapping are visual surveys during nesting, and basking surveys in early spring.
17	Hoop nets very effective but basking surveys also effective and much cheaper.
18	Early spring basking surveys, and nesting surveys, are most productive.
19	Most active early April through mid-June; early to mid fall activity is known and VES & hoop-net trapping might be effective in autumn. Readily captured in baited hoop-net traps, but usually need small diameter traps that can be placed in shallow waters. Nocturnal searches productive.
20	Does not respond well to baited traps; unbaited fyke nets preferable
21	Baited hoop nets very effective, as is early spring visual searches of wetland edges. Basking surveys sometimes effective in early spring.
22	Visual (basking) surveys productive entire season.
23	Detection probabilities needed for various sampling methods.
24	Early spring visual searches most productive.

LIZARDS: Comment Codes	
Code	Comments
1	Survey techniques poorly known
2	Very difficult to find or predict
3	May be able to find mole skinks by raking loose soil of pocket gopher mounds
4	Glass lizards avoid pitfall traps
5	Spring is best for I & M efforts; limited success with pitfall trapping
6	Visual surveys of suitable habitat are most productive. Tracks and burrows can be found as well.
7	Detection probabilities needed and more research into effective sampling methods.
8	Visual surveys of suitable habitat are productive. Detection probabilities needed and more research into effective sampling methods.
9	Detection probabilities needed and more research into effective sampling methods. Avoids pitfall traps.
10	Include standing trees and fences in visual searches. Detection probabilities needed for sampling methods.

5. TAXONOMIC SYNONYMS: Provides information about changes in scientific and common names of a given species (Crother et al. 2008)

FROGS AND TOADS		
Old Name	**New Name (re: Crother et al. 2008)**	**Common Name**
Bufo alvarius	*Ollotis alvaria*	Colorado River Toad
Bufo americanus	*Anaxyrus americanus*	American Toad
Bufo baxteri	*Anaxyrus baxteri*	Wyoming Toad
Bufo boreas	*Anaxyrus boreas*	Western Toad
Bufo californicus	*Anaxyrus californicus*	Arroyo Toad
Bufo canorus	*Anaxyrus canorus*	Yosemite Toad
Bufo cognatus	*Anaxyrus cognatus*	Great Plains Toad
Bufo debilis	*Anaxyrus debilis*	Green Toad
Bufo exsul	*Anaxyrus exsul*	Black Toad
Bufo fowleri	*Anaxyrus fowleri*	Fowler's Toad
Bufo hemiophrys	*Anaxyrus hemiophrys*	Canadian Toad
Bufo houstonensis	*Anaxyrus houstonensis*	Houston Toad
Bufo microscaphus	*Anaxyrus microscaphus*	Arizona Toad
Bufo nelsoni	*Anaxyrus nelsoni*	Amargosa Toad
Bufo punctatus	*Anaxyrus punctatus*	Red-spotted Toad
Bufo quercicus	*Anaxyrus quercicus*	Oak Toad
Bufo retiformis	*Anaxyrus retiformis*	Sonoran Green Toad
Bufo speciosus	*Anaxyrus speciosus*	Texas Toad
Bufo terrestris	*Anaxyrus terrestris*	Southern Toad
Bufo valliceps	*Anaxyrus valliceps*	Gulf Coast Toad
Bufo valliceps	*Ollotis nebulifer*	Coastal Plain Toad
Bufo woodhousii	*Anaxyrus woodhousii*	Woodhouse's Toad
Eleutherodactylus augusti	*Craugastor augusti*	Barking Frog
Pternohyla fodiens	*Smilisca fodiens*	Lowland Burrowing Treefrog
Rana areolata	*Lithobates areolatus*	Crawfish Frog
Rana berlandieri	*Lithobates berlandieri*	Rio Grande Leopard Frog
Rana blairi	*Lithobates blairi*	Plains Leopard Frog
Rana capito	*Lithobates capito*	Gopher Frog
Rana catesbeiana	*Lithobates catesbeianus*	American Bullfrog
Rana chiricahuensis	*Lithobates chiricahuensis*	Chiricahua Leopard Frog
Rana clamitans	*Lithobates clamitans*	Green Frog
Rana gylio	*Lithobates grylio*	Pig Frog
Rana heckscheri	*Lithobates heckscheri*	River Frog
Rana okaloosae	*Lithobates okaloosae*	Florida Bog Frog
Rana onca	*Lithobates onca*	Relict Leopard frog
Rana palustris	*Lithobates palustris*	Pickerel Frog
Rana pipiens	*Lithobates pipiens*	Northern Leopard Frog
Rana septentrionalis	*Lithobates septentrionalis*	Mink Frog
Rana sevosa	*Lithobates sevosus*	Dusky Gopher Frog
Rana sphenocephala	*Lithobates sphenocephala*	Southern Leopard Frog
Rana sylvatica	*Lithobates sylvaticus*	Wood Frog
Rana tarahumarae	*Lithobates tarahumarae*	Tarahumara Frog
Rana virgatipes	*Lithobates virgatipes*	Carpenter Frog

FROGS AND TOADS		
Old Name	**New Name (re: Crother et al. 2008)**	**Common Name**
Rana yavapaiensis	*Lithobates yavapaiensis*	Lowland Leopard Frog
Syrrhophus cystignathoides	*Eleutherodactylus cystignathoides*	Rio Grande Chirping Frog
Syrrhophus gutillatus	*Eleutherodactylus gutillatus*	Spotted Chirping Frog
Syrrhophus marnockii	*Eleutherodactylus marnockii*	Cliff Chirping Frog
N/A	*Pseudacris hypochondriaca*	Baja California Treefrog
N/A	*Pseudacris illinoensis*	Illinois Chorus Frog
N/A	*Rana sierrae*	Sierra Nevada Yellow-legged Frog
SALAMANDERS		
Old Name	**New Name (re: Crother et al. 2008)**	**Common Name**
Ambystoma triginum mavortium	*Ambystoma mavortium*	Barred Tiger Salamander
Typhlotriton spelaeus	*Eurycea spelaea*	Grotto Salamander
N/A	*Ambystoma bishopi*	Reticulated Flatwoods Salamander
N/A	*Batrachoseps gabrieli*	San Gabriel Mountains Slender Salamander
N/A	*Batrachoseps gavilanensis*	Gabilan Mountains Slender Salamander
N/A	*Batrachoseps gregarius*	Gregarious Slender Salamander
N/A	*Batrachoseps incognitus*	San Simeon Slender Salamander
N/A	*Batrachoseps kawia*	Sequoia Slender Salamander
N/A	*Batrachoseps luciae*	Santa Lucia Mountains Slender Salamander
N/A	*Batrachoseps minor*	Lesser Slender Salamander
N/A	*Batrachoseps nigriventris*	Black-bellied Slender Salamander
N/A	*Batrachoseps pacificus*	Channel Islands Slender Salamander
N/A	*Batrachoseps regius*	Kings River Slender Salamander
N/A	*Batrachoseps relictus*	Relictual Slender Salamander
N/A	*Batrachoseps robustus*	Kern Plateau Salamander
N/A	*Batrachoseps simatus*	Kern Canyon Slender Salamander
N/A	*Batrachoseps stebbinsi*	Tehachapi Slender Salamander
N/A	*Desmognathus abditus*	Cumberland Dusky Salamander
N/A	*Desmognathus folkertsi*	Dwarf Black-bellied Salamander
N/A	*Eurycea aquatica*	Brown-backed Salamander
N/A	*Eurycea chamberlaini*	Chamberlain's Dwarf Salamander
N/A	*Eurycea pterophila*	Fern Bank Salamander
N/A	*Gyrinophilus gulolineatus*	Berry Cave Salamander
N/A	*Hydromantes brunus*	Limestone Salamander
N/A	*Plethodon ainsworthi*	Bay Springs Salamander
N/A	*Plethodon amplus*	Blue Ridge Gray-cheeked Salamander

SALAMANDERS		
Old Name	**New Name (re: Crother et al. 2008)**	**Common Name**
N/A	*Plethodon chattahoochee*	Chattahoochee Slimy Salamander
N/A	*Plethodon cheoah*	Cheoah Bald Salamander
N/A	*Plethodon grobmani*	Southeastern Slimy Salamander
N/A	*Plethodon kiamichi*	Kiamichi Slimy Salamander
N/A	*Plethodon kisatchie*	Louisiana Slimy salamander
N/A	*Plethodon meridianus*	South Mountain Gray-cheeked Salamander
N/A	*Plethodon metcalfi*	Southern Gray-cheeked Salamander
N/A	*Plethodon mississippi*	Mississippi Slimy Salamander
N/A	*Plethodon ocmulgee*	Ocmulgee Slimy Salamander
N/A	*Plethodon savannah*	Savannah Slimy Salamander
N/A	*Plethodon sequoyah*	Sequoyah Slimy Salamander
N/A	*Plethodon shermani*	Red-legged Salamander
N/A	*Plethodon variolatus*	South Carolina Slimy Salamander
SNAKES		
Old Name	**New Name (re: Crother et al. 2008)**	**Common Name**
Charina trivirgata	*Lichanura trivirgata*	Rosy Boa
Chilomeniscus cinctus	*Chilomeniscus stramineus*	Burrowing Sandsnake
Drymarchon corais	*Drymarchon melanurus*	Central American Indigo Snake
Elaphe bairdi	*Pantherophis bairdi*	Baird's Ratsnake
Elaphe emoryi	*Pantherophis emoryi*	Great Plains Ratsnake
Elaphe gloydi	*Pantherophis gloydi*	Eastern Foxsnake
Elaphe guttata	*Pantherophis guttatus*	Red Cornsnake
Elaphe obsoleta	*Pantherophis alleganiensis*	Eastern Ratsnake
Elaphe obsoleta	*Pantherophis obsoletus*	Texas Ratsnake
Elaphe obsoleta	*Pantherophis spiloides*	Gray Ratsnake
Elaphe rosaliae	*Bogertophis rosaliae*	Baja California Ratsnake
Elaphe triapsis	*Senticollis triapsis*	Green Ratsnake
Elaphe slowinskii	*Pantherophis slowinskii*	Slowinski's Cornsnake
Elaphe subocularis	*Bogertophis subocularis*	Trans-Pecos Ratsnake
Elaphe vulpina	*Pantherophis vulpinus*	Western Foxsnake
Hypsiglena torquata	*Hypsiglena jani*	Chihuahuan Nightsnake
Hypsiglena torquata	*Hypsiglena ochrorhyncha*	Coast Nightsnake
Hypsliglena torquata	*Hypsiglena chlorophaea*	Desert Nightsnake
Masticophis bilineatus	*Coluber bilineatus*	Sonoran Whipsnake
Masticophis flagellum	*Coluber flagellum*	Coachwhip
Masticophis fuliginosus	*Coluber fuliginosus*	Baja California Coachwhip
Masticophis lateralis	*Coluber lateralis*	Striped Racer
Masticophis schotti	*Coluber schotti*	Schott's Whipsnake
Masticophis taeniatus	*Coluber taeniatus*	Striped Whipsnake
Stilosoma extenuatum	*Lampropeltis extenuatum*	Short-tailed Snake
N/A	*Storeria victa*	Florida Brownsnake
N/A	*Tantilla planiceps*	Western Black-headed Snake
N/A	*Thamnophis hammondii*	Two-striped Gartersnake

TURTLES		
Old Name	**New Name (re: Crother et al. 2008)**	**Common Name**
Clemmys insculpta	*Glyptemys insculpta*	Wood Turtle
Clemmys marmorata	*Actinemys marmorata*	Western Pond Turtle
Clemmys muhlenbergii	*Glyptemys muhlenbergii*	Bog Turtle
Graptemys gibbonsi	*Graptemys pearlensis*	Pearl River Map Turtle
N/A	*Chrysemys dorsalis*	Southern Painted Turtle
N/A	*Pseudemys peninsularis*	Peninsula Cooter
LIZARDS		
Old Name	**New Name (re: Crother et al. 2008)**	**Common Name**
Cnemidophorus arizonae	*Aspidoscelis arizonae*	Arizona Striped Whiptail
Cnemidophorus burti	*Aspidoscelis burti*	Canyon Spotted Whiptail
Cnemidophorus dixoni	*Aspidoscelis dixoni*	Gray Checkered Whiptail
Cnemidophorus exsanguis	*Aspidoscelis exsanguis*	Chihuahuan Spotted Whiptail
Cnemidophorus flagellicauda	*Aspidoscelis flagellicauda*	Gila spotted Whiptail
Cnemidophorus gularis	*Aspidoscelis gularis*	Common Spotted Whiptail
Cnemidophorus gypsi	*Aspidoscelis gypsi*	Little White Whiptail
Cnemidophorus hyperythra	*Aspidoscelis hyperythra*	Orange-throated Whiptail
Cnemidophorus inornata	*Aspidoscelis inornata*	Little Striped Whiptail
Cnemidophorus laredoensis	*Aspidoscelis laredoensis*	Laredo Striped Whiptail
Cnemidophorus marmorata	*Aspidoscelis marmorata*	Marbled Whiptail
Cnemidophorus neomexicana	*Aspidoscelis neomexicana*	New Mexico Whiptail
Cnemidophorus neotesselata	*Aspidoscelis neotesselata*	Colorado Checkered Whiptail
Cnemidophorus pai	*Aspidoscelis pai*	Pai Striped Whiptail
Cnemidophorus scalaris	*Aspidoscelis septemvittata*	Plateau Spotted Whiptail
Cnemidophorus sexlineatus	*Aspidoscelis sexlineata*	Six-lined Racerunner
Cnemidophorus sonorae	*Aspidoscelis sonorae*	Sonoran Spotted Whiptail
Cnemidophorus tesselata	*Aspidoscelis tesselata*	Common Checkered Whiptail
Cnemidophorus tigris	*Aspidoscelis tigris*	Tiger Whiptail
Cnemidophorus uniparens	*Aspidoscelis uniparens*	Desert Grassland Whiptail
Cnemidophorus velox	*Aspidoscelis velox*	Plateau Striped Whiptail
Cnemidophorus xanthonota	*Aspidoscelis xanthonota*	Red-backed Whiptail
Eumeces anthracinus	*Plestiodon anthracinus*	Coal Skink
Eumeces callicephalus	*Plestiodon callicephalus*	Mountain Skink
Eumeces egregius	*Plestiodon egregius*	Mole Skink
Eumeces fasciatus	*Plestiodon fasciatus*	Common Five-lined Skink
Eumeces gilberti	*Plestiodon gilberti*	Gilbert's Skink
Eumeces inexpectatus	*Plestiodon inexpectatus*	Southeastern Five-lined Skink
Eumeces laticeps	*Plestiodon laticeps*	Broad-headed Skink
Eumeces multivirgatus	*Plestiodon multivirgatus*	Many-lined Skink
Eumeces obsoletus	*Plestiodon obsoletus*	Great Plains Skink
Eumeces septentrionalis	*Plestiodon septentrionalis*	Prairie Skink
Eumeces skiltonianus	*Plestiodon skiltonianus*	Western Skink
Eumeces tetragrammus	*Plestiodon tetragrammus*	Four-lined Skink

LIZARDS		
Old Name	**New Name (re: Crother et al. 2008)**	**Common Name**
Gambelia sila	*Gambelia silus*	Blunt-nosed Leopard Lizard
Neoseps reynoldsi	*Plestiodon reynoldsi*	Florida Sand Skink
Phrynosoma coronatum	*Anota blainvillii*	Blainville's Horned Lizard
Phrynosoma douglasii	*Tapaja douglasii*	Pygmy Short-horned Lizard
Phrynosoma goodei	*Doliosaurus goodei*	Goode's Horned Lizard
Phrynosoma hernandesi	*Tapaja hernandesi*	Greater Short-horned Lizard
Phrynosoma modestum	*Doliosaurus modestum*	Round-tailed Horned Lizard
Phrynosoma platyrhinos	*Doliosaurus platyrhinos*	Desert Horned Lizard
Phyllodactylus xanti	*Phyllodactylus nocticolus*	Peninsular Leaf-toed Gecko
Sauromalus obesus	*Sauromalus ater*	Common Chuckwalla
N/A	*Anniella pulchra*	California Legless Lizard
N/A	*Crotaphytus vestigium*	Baja California Collared Lizard
N/A	*Gambelia copeii*	Cope's Leopard Lizard
N/A	*Urosaurus nigricaudus*	Baja California Brush Lizard
N/A	*Xantusia riversiana*	Island Night Lizard
N/A	*Xantusia sierrae*	Sierra Night Lizard
N/A	*Xantusia wigginsi*	Wiggins' Night Lizard

6. REFERENCES FOR TECHNIQUES TABLE: a list of books and articles, organized by PARC region, that are referenced or suggested in the *Species x Techniques Table* (see Comments).

REGION	LITERATURE CITED
SE	Applegarth, J.S. 1994. Wildlife Surveying and Monitoring Methods: Amphibians and Reptiles of the Eugene District. U.S.D.I., Bureau of Land Management, Eugene, Oregon. 59p.
SE	Boughton, R.G. and J. Staiger. 2000. Use of PVC pipe refugia as a sampling technique for Hylid treefrogs. American Midland Naturalist 144: 168-177.
SE	Briggler, J.Y., J.E. Johnson, and D.D. Rambo. 2004. Demographics of a ringed salamander (*Ambystoma annulatum*) breeding migration. The Southwestern Naturalist 49(2): 209-217.
SE	Bury, R.B. and S.P. Corn. 1991. Sampling methods for amphibians in streams in the Pacific Northwest. General Technical Report, PNW-GTR-275. U.S.D.A. Forest Service, Pacific Northwest Research Station, Portland, OR. 29 p.
SE	Campbell, H.W. and S.P. Christman. 1982. Field techniques for herpetofaunal community analysis. Pp. 193-200 In N.J. Scott (ed.). Herpetological Communities. U.S.D.I. Fish and Wildlife Service Research Report 13. 239 p.
SE	Fellers, G.M. and K.L. Freel. 1995. A standardized protocol for surveying aquatic amphibians. Technical Report NPS/WRUC/NRTR-95-01, U.S.D.I. National Park Service/National Biological Survey, University of California, Davis, CA. 117 p.
SE	Grant, B.W., A.D. Tucker, J.E. Lovich, A.M. Mills,P.M. Dixon, and J.W. Gibbons. 1992. The use of coverboards in estimating patterns of reptile and amphibian biodiversity. Pp. 379-403 in D.R. McCullough and R.H. Barrett (eds). Wildlife 2001. Elsevier Science

REGION	LITERATURE CITED continued
SE	Grant, E.H.C.and L.L. Bailey. 2009. Vernal Pool Amphibian Monitoring Protocol- Longterm monitoring, including dipnet surveys. USGS.
SE	Heyer, R.W., M.A. Donnelly, R.W. McDiarmid, L.C. Hayek, and M.S. Foster (eds). 1994. Measuring and Monitoring Biological Diversity: Standard Methods for Amphibians. Smithsonian Institution Press, Washington DC.
SE	Humphries, W.J. and T.K. Pauley. 2005. Life history of the Hellbender, *Cryptobranchus alleganiensis*, in a West Virginia stream. Am. Midl. Nat. 154: 135-142.
SE	Iverson, J.B. 1977. Geographic variation in the musk turtle, *Sternotherus minor.* Copeia 502-517.
SE	Kolbe, J. J., B. E. Smith, and D. M. Browning. 2002. A large aggregation of tiger salamanders (*Ambystoma tigrinum melanostictum*) at a black-tailed prairie dog (*Cynomys ludovicianus*) town in southwestern South Dakota. Herpetological Review 33:95-99.
SE	Micacchion, Mick. 2004. Integrated Wetland Assessment Program. Part 7: Amphibian Index of Biotic Integrity (AmphIBI) for Ohio Wetlands. Ohio EPA Technical Report, WET/2004-7. Ohio Environmental Protection Agency, Wetland Ecology Group, Division of Surface
SE	Mitchell, J.C. 2000. Amphibian Monitoring Methods and Field Guide. The National Zoological Park's Conservation and Research Center, Front Royal, Virginia. 56 p.
SE	Olson, D.H., W.P. Leonard, and R.B. Bury (eds). 1997. Sampling Amphibians in Lentic Habitats: Methods and approaches for the Pacific Northwest. Society for Northwestern Vertebrate Biology, Northwest Fauna 4. Olympia, WA.
SE	Pauley, T.K. and M. Little. 1998. A new technique to monitor larval and juvenile salamanders in stream habitats. Banisteria 12: 32-36.
SE	Peterson, C.L., R.F. Wilkinson, D. Moll, and T. Holder. 1992. Estimating the number of female *Ambystoma annulatum* (Caudata: Ambystomatidae) based on oviposition. The Southwestern Naturalist 37(4): 425-426.
SE	Williams, R.D., J.E. Gates, and C.H. Hocutt. 1981. An evaluation of known and potential sampling techniques for Hellbender, *Cryptobranchus alleganiensis*. Journal of Herpetology 15(1): 23-27.
SE	Wilson, J.J. and T.J. Maret. 2002. A comparison of two methods for estimating the abundance of amphibians in aquatic habitats. Herpetological Review 33(2): 108-110.
NE	Chazal, Anne C. 2005. Results of Surveys for the Peaks of Otter Salamander (*Plethodon hubrichti*) in 2005. Natural Heritage Technical Report 06-05. Virginia Department of Conservation and Recreation, Division of Natural Heritage. Richmond, VA 13 pp.
NE	Glorioso, B.M. and M. L. Niemiller. 2006. Herpetol. Rev. 37(2):185-187.
NE	Graham, T.E. 1969. Pursuit of the Plymouth turtle.J. Int. Turtle and Tortoise Soc. 3(1): 10-13.
NE	Graham, T.E. 1981. New approaches to endangered turtle reseach. Bios 52: 121-126. Graham 1971. Growth rates, basking Copeia 353-356.
NE	http://www.dgif.virginia.gov/wildlife/information/?t=1
NE	http://www.mass.gov/dfwele/dfw/nhesp/species_info/nhfacts/ambystoma_laterale.pdf
NE	Jung, R. E., S. Droege, J. R. Sauer, and R. B. Landy. 2000. Evaluation of terrestrial and streamside salamander monitoring techniques at Shenandoah National Park. Environmental Monitoring and Assessment 63:65-79.
NE	Pauley, T. K. 1995. Aquatic salamanders. Pp. 15-22 in: Reardon, R. C. (Ed.). Effects of diflubenzuron on non-target organisms in broadleaf forested watersheds in the northeast. USDA Forest Service. National Center of Forest Health Management. FHM-NC-05-95.

REGION	LITERATURE CITED continued
NE	Pauley, T. K. 2007. Revised notes on the range of the Cheat Mountain Salamander, *Plethodon nettingi*, (Amphibia: Caudata). Proceedings of the West Virginia Academy of Science 79(2):16-21.
NE	Pauley, T. K. 2008b. The Appalachian Inferno: A possible reason for the disjunct distribution of a federally protected salamander species, *Plethodon nettingi* (Cheat Mountain Salamander). Northeastern Naturalist 15(4):595-606.
NE	Pauley, T. K., and M. Little. 1998. A new technique to monitor larval and juvenile salamanders in stream habitats. Banisteria 12:32-36.
NE	Pauley, T.K. 2005. Reflections Upon Amphibian Conservation. *In* Amphibian Declines: The Conservation Status of the United States Species. M.J. Lannoo, Editor. University of California Press, Berkeley, California. Chapter 40, Pp. 277-281.
NE	Pauley, T.K. 2008a. Status of the Federally Threatened Cheat Mountain Salamander, *Plethodon nettingi*, (Amphibia: Caudata): Sixty years later. In Press.
NE	Petranka, J. W. 1998. Salamanders of the United States and Canada. Smithsonian Institute Press Washington and London.
NE	Riechenbach et al. 2000. Population dynamics of the cave salamander (*Eurycea lucifuga*), long-tailed salamander (*E. longicauda longicauda*), and three-lined salamander (*E. guttolineata*) in the county record location for *E. lucifuga* and *E. l. longicauda* in Amherst County: Final Report
NE	Sorensen. K. 2002. Developing a monitoring protocol for Siren and Amphiuma in the Southeastern United States. USGS Technical Publication
SW	Corn, P.S., E. Muths, and W. M. Iko. 2000. A comparison in Colorado of three methods to monitor breeding amphibians. Northwestern Naturalist 81: 22-30.
SW	Hammerson, G. A. 1989. A field survey of amphibians in the Rocky Mountains of Colorado, August 1989. 53 In Colorado Division of Wildlife.
SW	Lambert, B. 2004. Canyon of the Ancients National Monument amphibian and reptile inventory. 28 In Colorado Natural Heritage Program, Colorado State University.
SW	Livo, L. J. 1995. Amphibian surveys in Boulder, Clear Creek, and Gilpin Counties, Colorado, 1994. Colorado Division of Wildlife, Denver, CO.
SW	Livo, L. J. 1997. City of Boulder 1996 amphibian and reptile survey. Unpublished report, City of Boulder Open Space.
SW	Mackessy, S. P. 1998. A survey of the herpetofauna of southeastern Colorado with a focus on the current status of two candidates for protected species status: the massasauga rattlesnake and the Texas horned lizard. Unpublished final report, Colorado Division of Wildlife.
SW	Muths, E. 2003. A radio transmitter belt for small ranid frogs. *Herpetological Review* 34(4): 345-348.
SW	Muths, E. 2003. Home range and movements of boreal toads in undisturbed habitats. *Copeia* 2003(1): 161-165.
SW	Muths, E., R. D. Scherer, P. S. Corn, and B. A. Lambert. 2006. Estimation of temporary emigration in male toads. Ecology 87: 1048-1056.
SW	Scherer, R. D. 2008. Detection of wood frog egg masses and implications for monitoring amphibian populations. Copeia 3:669-672
SW	Scherer, R. D., E. Muths, B. R. Noon, AND P. S. Corn. 2005. An evaluation of weather and disease as causes of decline in two populations of boreal toads. Ecological Applications 15: 2150-2160.

REGION	LITERATURE CITED continued
SW	Schorr, R. A. and B. A. Lambert. 2006. Longnose leopard lizard (*Gambelia wislizenii*) home range and habitat use on Cannonball Mesa, Colorado. Unpublished report, Bureau of Land Management, Dolores, CO.
SW	Shipley, B. K., and R. P. Reading. 2002. A comparison of herpetofauna abundance between black-tailed prairie dog colonies and non-colonized grassland. 1-13 In Denver Zoological Gardens.
SW	Turner, F.B. and P.A. Medica. 1982. The distribution and abundance of the flat-tailed horned lizard (*Phrynosoma mcallii*). Copeia 1982(4): 815-823.
NW	Clayton, D.R. and D.H. Olson. 2009. Conservation assessment for the Oregon Slender Salamander *(Batrachoseps wrighti*). U.S.D.A. Forest Service Region 6 and U.S.D.I. Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml
NW	Corkran, C. C., and C. Thoms. 2006. *Amphibians of Oregon, Washington, and British Columbia: A Field Identification Guide. Revised edition. Lone Pine Publishing. Edmonton, Alberta. 176 pp.*
NW	COSEWIC 2002. COSEWIC assessment and status report on the western toad *Bufo boreas* in Canada. Committee on the Status of Endangered Wildlife in Canada. Ottawa. vi + 31 pp.
NW	Crisafulli, C.M., D.R. Clayton, and D.H. Olson. 2008. Conservation assessment for the Larch Mountain Salamander *(Plethodon larselli*). U.S.D.A. Forest Service Region 6 and U.S.D.I. Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml
NW	Cushman, K.A. and C.A. Pearl. 2007. A conservation assessment for the Oregon Spotted Frog (*Rana pretiosa*).U.S.D.A. Forest Service Region 6 and U.S.D.I. Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml
NW	Davis, R. and D. Olson. 2007. Field validation of a landscape habitat model for the foothill yellow-legged frog (*Rana boylii*) in Oregon. U.S.D.A. Forest Service Region 6 and U.S.D.I. Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/documents/inventories/inv-rpt-ha-rabo-hab-model-field-valid-2008.pdf
NW	Heyer, W.R., Donnelly, M.A., McDiarmid, R.W., Hayek, L.-A.C., and Foster, M.S., eds., 1994, Measuring and monitoring biological diversity–Standard methods for amphibians: Washington, D.C., Smithsonian Institution Press, 364 p
NW	Howell, B.L. and C.R. Roberts. 2008 A conservation assessment for the Olympic torrent salamander (*Rhyacotriton olympicus*). U.S.D.A. Forest Service Region 6 and U.S.D.I. Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml
NW	Jones, L.L.C., W.P Leonard, and D.H. Olson (eds.). 2005. *Amphibians of the Pacific Northwest.* Seattle Audubon Society. 227 p.
NW	Kendell, K. 2002. Survey protocol for the northern leopard frog. Alberta Sustainable Resource Development, Fish and Wildlife Division, Alberta Species at Risk Report No. 43, Edmonton, AB. 30 pp.
NW	Myers, C. H., L. Eigner, J. A. Harris, R. Hilman, M. D. Johnson, R. Kalinowski, J. J. Muir, M. Reyes, and L. E. Tucci. 2007. A comparison of ground-based and tree-based polyvinyl chloride pipe refugia for capturing *Pseudacris regilla* in northwestern California. Northwestern Naturalist 88:147-154.

REGION	LITERATURE CITED continued
NW	Nauman, R.S. and D.H. Olson. 2004. Surveys for terrestrial amphibians in Shasta County, California with notes on the distribution of Shasta salamanders (*Hydromantes shastae*). Northwestern Naturalist 85:29-32.
NW	Olson, D. H. and G. Weaver. 2007. Vertebrate assemblages associated with headwater hydrology in western Oregon managed forests. Forest Science 53(2):343-355.
NW	Olson, D.H. (ed.) 1999. Survey protocols for amphibians under the Survey and Manage provision of the Northwest Forest Plan. Version 3.0. Interagency publication of the Regional Ecosystem Office, Portland, OR. Bureau of Land Management Publication BLM/OR/WA/PT-00/033+1792; U.S. Government Printing Office: 2000--589-124/04022 Region No. 10. 310 p. This is being reprinted as Chapter 6 in Olson, D.H., Clayton, D., Nauman, R., Welsh, H.H. Jr. 2009. Conservation of the Siskiyou Mountains Salamander (*Plethodon stormi*). Northwest Fauna. In press.
NW	Olson, D.H. 2008. Conservation assessment for the Black Salamander (*Aneides flavipunctatus*) in Oregon. U.S.D.A. Forest Service Region 6 and U.S.D.I. Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml
NW	Olson, D.H. 2008. Conservation assessment for the California Slender Salamander *(Batrachoseps attenuatus*) in Oregon. U.S.D.A. Forest Service Region 6 and U.S.D.I. Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml
NW	Olson, D.H. and R. Davis. 2007. Conservation assessment for the Foothill Yellow-Legged Frog *(Rana boylii*) in Oregon. U.S.D.A. Forest Service Region 6 and U.S.D.I. Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml
NW	Olson, D.H., David Clayton, Edward C. Reilly, Richard S. Nauman, Brenda Devlin, and Hartwell H. Welsh, Jr.. 200s. Conservation strategy for the Siskiyou Mountains Salamander *(Plethodon stormi*), northern portion of the range. U.S.D.A. Forest Service Region 6 and U.S.D.I. Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml
NW	Olson, D.H., W.H. Leonard, and R.B. Bury (eds). 1997. Sampling amphibians in lentic habitats. Northwest Fauna 4:1-134.
NW	Pretzlaw, T., M. Huynh, L. Takats, and L. Wilkinson. 2002. Protocol for monitoring long-toed salamander (*Ambystoma macrodactylum*) populations in Alberta. Alberta Sustainable Resource Development, Fish and Wildlife Division, Alberta Species at Risk Report No. 35. Edmonton, Alberta. 17 pages
NW	Richter, K.0 . 1995. A simple aquatic funnel trap and its application to wetland amphibian monitoring. Herpetol. Rev. 26:90-9 1.
NW	Tait, C. 2007. Conservation assessment of the Great Basin Population of the Columbia Spotted Frog (*Rana luteiventris*). U.S.D.A. Forest Service Region 6 and U.S.D.I. Bureau of Land Management Interagency Special Status and Sensitive Species Program. Available at: http://www.fs.fed.us/r6/sfpnw/issssp/species-index/fauna-amphibians.shtml
NW	Thoms, C., C. C. Corkran, and D. H. Olson. 1997. Basic amphibian survey for inventory and monitoring in lentic habitats. In D. H. Olson, W. P. Leonard, and R. B. Bury, editors. Sampling Amphibians in Lentic Habitats: Methods and Approaches for the Pacific Northwest. Society for NW Vertebrate Biology. Olympia, WA. Northwest Fauna 4:35-46.

REGION	LITERATURE CITED continued
MW	Bishop, C. A. and K. E. Pettit (editors). 1992. Declines in Canadian amphibian populations: designing a national monitoring strategy. Occ. Paper No. 76, Canadian Wildlife Service, 120 pp.
MW	Busby, W. H. and W. R. Brecheisen. 1997. Chorusing Phenology and Habitat Associations of the Crawfish Frog, *Rana areolata* (Anura: Ranidae), in Kansas. The Southwestern Naturalist, 42(2):210-217.
MW	Carroll, D. M. 2001. The Year of the Turtle. Camden House Publishing, Inc. Charlotte, VT, 172 pp.
MW	Ernst, C. H. and E. M. Ernst. 2003. Snakes of the United States and Canada. Smithsonian Institution Press, Washington, D.C.
MW	Ernst, C. H. and J. E. Lovich. 2009. Turtles of the United States and Canada. 2nd edition. The Johns Hopkins University Press, Baltimore, MD.
MW	Heyer, W. R., M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (editors). 1994. Measuring and monitoring biological diversity. Standard methods for amphibians. Smithsonian Institution Press, Washington. 364 pp.
MW	Karns, D. R. 1986. Field herpetology: Methods for the study of amphibians and reptiles in Minnesota. James Ford Bell Museum of Natural History Occasional Paper No. 18. Minneapolis, MN.
MW	Lannoo, M. J. (Editor). 2005. Amphibian Declines: the Conservation Status of United States Species. Univ. California Press, Berkeley and Los Angeles, California. 1094 pp.
MW	Mauger, D., T. Bell & E. Peters. 2000. Distribution and Habitat of the Southern Two-Lined Salamander, *Eurycea cirrigera*, in Will County, Illinois: Implications For Population Management and Monitoring. Jour. Iowa Acad. Sci. 107(3): 168-174.
MW	Smith, H. M. and D. Frost. 1995 (1946). Handbook of Lizards: Lizards of the United States and of Canada. Comstock Publishing, Ithaca, N.Y.

American alligator (*Alligator mississipiensis*)

John White

CHAPTER 6. ANALYSIS OF INVENTORY AND MONITORING DATA

Christopher T. Winne

INTRODUCTION

This chapter outlines the basic approaches for analysis of data resulting from amphibian and reptile inventory and monitoring programs. Analytical techniques are highly varied and specialized according to project goals, techniques applied, and resources available. Additionally, much of the mathematics used in such analyses is beyond the scope of this book. Thus, the goal of this chapter is to familiarize the reader with some of the primary analytical approaches and provide references for further exploration of the topic.

The primary result of most inventories is a list of species, thus we begin with a discussion of some of the techniques used to estimate species richness and evaluate the completeness of a species list. This is followed by a discussion of coefficients of association which use presence/absence (i.e., detection/non-detection) data to make inferences about species and habitat associations. Comprehensive surveys and monitoring programs may require population estimates; the section on estimating population size describes several techniques used to make mathematical predictions of actual population sizes. The section on diversity indices describes a method for using species counts and relative abundances to create a single index describing the community composition. Finally, the section on occupancy models describes a technique useful for monitoring species presence over larger spatial scales.

Although the goals of each study are different and the data generated will depend on the study design employed, the following is a guideline of the applicability of each section (described above) to the four basic study types defined by this manual:

Rapid assessment inventory

- *Estimating species richness*: Quadrat sampling and multiple sampling can be used if the study design allows.
- *Coefficients of association*: May be applicable if multiple sites are surveyed.

Comprehensive survey inventory

- *Estimating species richness*: Species accumulation rates, quadrat sampling, and multiple sampling may be used.
- *Coefficients of association*: May be applicable if multiple sites are surveyed.

- *Diversity indices*: May be calculated if population/ relative abundance data are collected at the community level.

- *Estimating population size*: Population sizes can be estimated for species of interest.

Presence/absence monitoring

- *Estimating species richness*: Richness estimates over time may reveal community level trends.

- *Coefficients of association*: May be applicable if multiple sites are surveyed.

- *Occupancy models*: May be applicable if the scale of the study area is large and multiple sites are monitored.

Population status monitoring

- *Estimating population size*: Population estimates made over time may reveal long-term trends.

- *Diversity indices*: May be calculated if population/relative abundance data are collected at the community level.

ESTIMATING SPECIES RICHNESS

Species richness is simply the number of unique species occurring within a defined area. Even though the methods detailed in this manual are likely to discover a large proportion of the animals that exist in the study area, it is inevitable that some species will go undetected. Therefore a simple species count from capture data is likely to be an underestimate of true richness and a mathematical estimate will give a better idea of the number of species that are actually present. The following techniques allow assessment of the completeness of a species list and estimation of true species richness.

Species accumulation rates

A simple method for determining the completeness of a species list and estimating true species richness is visual inspection of a graph of the cumulative number of species detected versus the cumulative search time. The general shape of these graphs is shown below (Fig. 6-1). The number of species detected rises rapidly early in the search period as common and easily detectable species are quickly added to the species list. The rate begins to slow as only rarer and less detectable species are added to the list. The curve eventually approaches an asymptote, or leveling off point, that approximates the true species richness. If data plotted in this way do not appear to approach an asymptote, it is likely that the species list is somewhat incomplete and that additional sampling should be performed to make an accurate richness estimate.

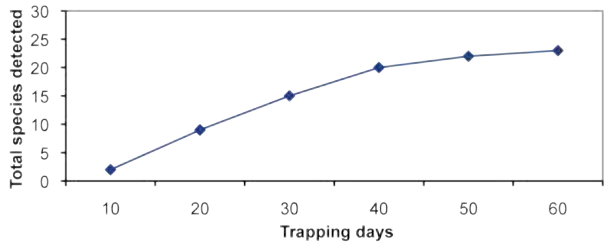


Figure 6-1. A plot of total species detected versus number of trapping days demonstrating the shape of many species accumulation curves. Here the shape of the curve suggests that most species available for capture (given techniques used and timing of study) have been captured as the curve approaches an asymptote of approximately 24 species.

Quadrat sampling

Quadrat sampling to estimate richness relies on the same principles as mark-recapture models for estimation of abundance (see Chapter 5 techniques section on Quadrat sampling for more information). The study area is divided into quadrats of equal size and shape of which a randomly chosen subset is sampled. Identical capture techniques should be applied in each quadrat to develop a complete species list. Species richness is estimated based on the assumption that each species has equal capture probability in all quadrats. With a two quadrat design, the total number of species (N) can most simply be estimated by the following:

$$\hat{N} = \frac{n_1 n_2}{m}$$

n_1 = number of species identified in quadrat 1
n_2 = number of species identified in quadrat 2
m = number of species identified in both quadrats

As mentioned above, this estimate is limited by the assumption that detection probabilities are equal for all species. This will rarely be the case in real situations given differences in relative abundance, behavior, habitat, and crypsis. Therefore, a more useful estimator may be the following jack-knife method developed by Burnham and Overton (1978) that relaxes that assumption:

$$\hat{N} = S + \frac{(t-1) f_2}{t}$$

S = total number of species detected
t = number of quadrats
f_1 = number of species found on only one quadrat

Computer software, such as CAPTURE (Rexstad and Burnham 1991), can also be used to estimate richness from quadrat data.

Multiple sampling

Multiple sampling relies on the principle of decreasing species accumulation rates, but formalizes the sampling so that mathematical estimates of richness can be made. The entire survey area is sampled on multiple occasions using identical sampling methods and effort among the sampling periods. The premise of this estimation is that the manner in which the number of new species detected decreases with each subsequent sample reveals information about the number of species remaining. The essential data for this method are an enumeration of the number of new species detected during each sampling period. Estimators using this method have been developed by Otis et al. (1978) and Pollock and Otto (1983) and can be computed using CAPTURE (Rexstad and Burnham 1991).

COEFFICIENTS OF ASSOCIATION

Coefficients of association are important tools for analysis of presence/absence (i.e., detection/non-detection) lists and other binary (two state) data. Generally, coefficients of association reveal the degree of relatedness between two samples. A primary use of these coefficients in herpetofaunal inventories is to assess the similarity of the species lists generated at two or more sample sites. They can also be used to assess habitat preferences and the degree of coexistence between species in different habitats.

Species B or habitat or state	Species A		
	Present	Absent	Total
Present	a	b	a + b
Absent	c	d	c + d
Total	a + c	b + d	n

To use coefficients of association, the data are arranged in a table where the cells represent the frequency that each case occurred. For example "a" might be the number of plots in which both Species A and Species B were found.

Once the data are properly arranged, a number of coefficients can be applied. Which coefficient to choose is determined by the question being asked and the properties of the underlying data. A full discussion of the properties of coefficients of association is beyond the scope of this book, but Hayek (1994) presents a useful list of popular coefficients along with discussion of their use.

Perhaps the most commonly used coefficient of association in biological studies is the χ^2 (chi square) statistic which takes the following form for a 2x2 table:

$$\chi^2 = \frac{n\,(ad - bc)^2}{(a + b)\,(c + d)\,(a + c)\,(b + d)}$$

Once calculated, the χ^2 statistic is compared to a published table of critical values. If the calculated value exceeds the critical value, then there is evidence for support of a (statistically significant) relationship between the values in the table.

Example

Fifty (50) isolated wetlands were surveyed using time-constrained dipnetting. At each wetland the presence or absence of fish was recorded as well as the presence or absence of mole salamander (*A. talpoideum*) larvae. The results are summarized in a 2x2 table as follows:

Fish	*A. talpoideum*		
	Present	Absent	Total
Present	3	15	10
Absent	26	6	32
Total	29	21	50

The χ^2 statistic is then calculated as follows:

$$\chi^2 = \frac{50\ (18 - 390)^2}{(18)\ (32)\ (29)\ (21)} = 19.72$$

The critical value for a 2x2 table with a=0.05 is 3.84. Since 19.72 is greater than 3.84 we conclude that the two variables are not independent. In this case, wetlands where fish are present are significantly less likely to have *A. talpoideum* than wetlands where fish are absent.

ESTIMATING POPULATION SIZE

Whereas the goal of many monitoring programs may be to simply detect population trends, population status monitoring will require determination of absolute population size. Since it will be impossible, in most cases, to census the entire population of interest, population size must be mathematically estimated. The

two primary methods used to make such estimates are mark-recapture and removal sampling. In general, mark-recapture studies are more labor intensive, yet may provide better estimates per unit time. These methods have developed rapidly in recent years, and a full accounting of the available techniques and their mathematical calculations is beyond the scope of this book (see Schwartz and Seber 1999 for a review of recent progress). We do, however, present some of the most commonly used methods with basic calculations and provide references in which more detailed information may be found.

Mark-recapture

As the name implies, mark-recapture studies require that target animals are caught during an initial sampling period, marked for later identification, released, and recaptured during at least one subsequent sampling period. Appendices I, II, and III provide detailed information on capturing, handling, and marking amphibians and reptiles.

Software

- Several computer programs have been developed to make increasingly complex and computation intensive population estimators available to researchers.
- Many new developments in the field are now made through new software releases.
- State of the art software estimators can give good results despite violation of assumptions, can incorporate telemetry data, calculate a variety of demographic parameters, and are adaptable to numerous study designs.
- However, a coherent and inclusive package has yet to be developed, so care must be taken in choosing the appropriate software and understanding its capabilities and limitations.
- Colorado State Univesity wildlife researchers maintain a website for one of the more popular, free population modeling software programs, program MARK:

http://www.warnercnr.colostate.edu/~gwhite/mark/mark.htm

- The USGS Patuxent Wildlife Research Center also maintains a collection of freely distributed software population estimators, including PRESENCE, at:

http://www.mbr-pwrc.usgs.gov/software.html

Before conducting a mark-recapture study, the goals of the project should be clearly defined (see Chapters 3 and 4). Many population estimators have been developed for analysis of mark-recapture data. The estimators differ in the assumptions that must be met, sampling required, and parameters that can be estimated. Therefore, the goals of the study, the biology of the study organism, and the available resources will determine the appropriate population estimator and associated sampling protocol.

Population estimators

Population estimators can generally be broken down into three groups based on whether they assume the population is closed, open, or mixed. A closed population is one in which no immigration, emigration, birth, or death occurs over the study period. The assumption of a closed population is best satisfied by a short term study with few sampling periods. Estimators that assume closed population can only estimate population size, but can require as few as two sampling periods.

An open population is one in which one or more of those processes occur during the study period. Open-population estimators are most useful for long term studies in which the assumption of a closed population cannot be met or when estimates of mortality and recruitment are needed in addition to an estimate of population size. These estimators can provide more information, however they require three or more sampling periods and thus demand greater resource investment.

A mixed population is one that undergoes periods during which the population is open and periods during which the population is closed. Robust design population models require the researcher to perform a series of closed-population model sampling events, with each sampling event separated by longer (open) periods of time. The primary (open) periods are used to calculate survival rates using an open model approach. The secondary (closed) periods are used to calculate population size and capture and recapture probabilities using a closed model approach. Information from both primary and secondary periods is pooled to estimate temporary emigration rates. Robust design models are most useful when the goal is to closely monitor both the population size and vital rates of one or a few focal populations, particularly for reptile and amphibian species that exhibit temporal variation in detectability or that rely on temporary migration as a normal part of their life cycle (Bailey et al. 2004b; Bailey et al. 2004c). Robust design models also have the advantage of increased precision in parameter estimates such as survival and population size compared to estimates generated by open or closed models alone.

Closed population models – The Peterson estimate, also known as the Lincoln-Peterson or the Lincoln Index, is the most fundamental closed-population estimator for mark-recapture data (Peterson 1896;

Lincoln 1930). This estimate requires two sampling periods and makes the following assumptions:

1. The sampled population is closed (i.e., no birth, death, immigration, or emigration). This may be relaxed if only death and emigration are occurring (in which case the population size at the first sampling is estimated) or if only recruitment is occurring (in which case the population size at the second sampling is estimated).

2. All animals (including marked individuals) have equal probability of capture during each sampling period.

3. All marks are permanent for the duration of the study.

4. Marking does not influence survival.

5. Sampling is conducted randomly over the study area. Alternatively, sampling may be conducted systematically if marked animals redistribute randomly after release.

The Peterson estimate of population size N is:

$$\hat{N} = \frac{n_1 n_2}{m}$$

n_1 = animals caught, marked, and released during first sampling period
n_2 = total number of animals caught during second sampling period
m = number of marked animals caught during second sampling period

Bailey (1951) and Chapman (1951) each devised modifications of the Peterson estimate that are useful when the number of animals recaptured is low. These modifications also allow calculation of the standard error for the population estimate. A flexible computer program for closed populations is CAPTURE (White et al. 1978; Rexstad and Burnham 1991). The capture program includes several population estimators that allow relaxation of some of the normal assumptions. For example, use of the Jack-knife estimator allows capture probabilities to vary by individual animal. Additionally, program MARK allows the use of simple closed population models, as well as other more complex closed population models (White and Burnham 1999).

Open population models – The Jolly-Seber model (Jolly 1965; Seber 1965) is most useful when sampling is to be conducted over a long time period and the assumption of a closed population cannot be met. This method also allows for an unlimited number of sampling periods and can estimate survival rates and population gains (birth and immigration). Assumptions 2-5 from the Peterson estimate apply to the Jolly-Seber method in addition to the assumption that all animals have an equal probability of survival between any two sampling periods.

The estimators of population size, gains, and losses are based on the following statistics that can be directly obtained from the capture-history:

m_i = number of marked animals caught during sampling period i.
n_i = total number of animals caught during sampling period i.
r_i = number of marked animals released during period i.
y_i = number of animals marked and released during period i that are captured during a subsequent sampling period.
z_i = number of animals caught during a sampling period before and after period i, but not during period i.

M_i is the estimate of the total number of marked animals remaining in the population evaluated as follows:

$$M_i = m_i + \frac{z_i r_i}{y_i}$$

The estimate for population size during sampling period i is given by:

$$N_i = \frac{M_i (n_i + 1)}{(m_i + 1)}$$

The survival rate at time i can be estimated with the following expression:

$$\phi_i = \frac{M_{i+1}}{(M_i - m_i + r_i)}$$

The population gain (births and immigration) between sampling period i and period i + 1 is estimated as follows:

$$g_i = N_{i+1} - \phi_i N_i$$

There are many other population estimators for open populations. Triple Catch is a useful estimator that requires three sampling periods and estimates survival and gains in addition to population size (Begon 1979). The Fisher-Ford method estimates gains, losses, and population size, but cannot estimate survival rates and standard errors for estimated parameters. A number of other useful open-population estimators have been incorporated into computer programs. Some of the most valuable and widely used programs are JOLLY (Pollock et al. 1990), POPAN (Arnason and Schwartz 1987), and MARK (White and Burnham 1999).

Robust design population models – Advances in computer processing speed and modeling approaches now allow researchers to use a single model to simultaneously incorporate features of both closed and open population models to estimate demographic parameters from mark-recapture data. In addition, robust design models can pool information from closed and open capture periods to estimate temporal varia-

A Case Study

A research group from the University of Georgia was interested in a population of diamondback terrapins (*Malaclemys terrapin*) in a tidal creek on the South Carolina coast. Increased crab trapping in this creek may be threatening the turtles through mortality in crab traps. The researchers' goal was to estimate population size and adult survivorship so these parameters could be compared to historical data and serve as a baseline for future studies.

Since detectability for this species may be fairly low, mark-recapture was clearly preferable to removal sampling. They determined that they had the resources for annual one-week sampling intervals over a period of four years. Since the time interval between samplings was long and diamondback terrapins are known to make extrapopulational movements, the researchers decided that an open population model was necessary for an accurate population estimate. Because they wanted estimates of survival, the researchers chose to use the Jolly-Seber model to estimate population size.

Their collection data after four years were as follows ("x" indicates turtle was captured that year):

Turtle ID	2000	2001	2002	2003
ABC	x	x	x	
ACD		x		x
AHV	x		x	
AKV		x	x	
ANX	x			
BDX	x	x		
BHV	x			
BKL		x	x	
BNX	x			
HJK			x	x
HJLO			x	x
IKV			x	x
INX		x	x	
JLN	x			
JKLV	x			
JKV		x		
KLU		x		
KNX	x		x	x
LNO				x
LMU				x

From this data the statistics associated with the Jolly-Seber method were computed.

2000	2001	2002	2003
x	x	x	
	x		x
x		x	
	x	x	
x			

To estimate the population during 2001, they first calculated M_2 as the following:

$$M_2 = m_2 + \frac{z_2 r_2}{y_2} = 2 + \frac{2 \times 8}{4} = 6$$

The population size N_2 was then estimated as:

$$N_2 = \frac{M_2 (n_2 + 1)}{(m_2 + 1)} = \frac{6 (9)}{3} = 18$$

To estimate survival they first calculated M_3:

$$M_3 = m_3 + \frac{z_3 r_3}{y_3} = 6 + \frac{1 \times 9}{3} = 9$$

Then they used the formula for survival:

$$\phi_2 = \frac{M_3}{(M_2 - m_2 + r_2)} = \frac{9}{6 - 2 + 8} = 0.75$$

Thus in 2001, the population size was approximately 18 terrapins, and each individual had a 75% chance of surviving from one year to the next. Since this annual survival rate was much lower than that detected in other healthy populations, it supported the researchers' hypothesis that crab pot mortality was a factor in the demographics of this population.

tion in detectability, as well as other information. We have outlined the appropriate steps for collecting data suitable for analyses using mixed population models (see Chapter 3), but the details of the mathematics of mixed population models are beyond the scope of this chapter. Nevertheless, program MARK is capable of performing robust design population analysis and is freely available (see Software box in this chapter). For further information and examples of the use of robust design population models to analyze mark-recapture data, see Pollock 1982; Kendall and Nichols 1995; Kendall et al. 1997; Bailey et al. 2004a,b,c.

Removal Sampling

Removal sampling is a means of population estimation in which captured animals are physically removed from a chosen area on two or more sampling occasions. Sample plots should be chosen so that they are large enough to represent the population of interest, small enough to facilitate removal of a large proportion of the animals, and isolated to minimize immigration and emigration during the study. The conditions for effective estimation from removal sampling include the following:

1. The population is closed or stationary (no net gains or losses) during the study period.

2. High detectability of target organisms, allowing high removal rates.

3. Equal catchability of all animals within a sampling period (this condition can be relaxed with some of the mathematical estimators).

4. Catchability of individuals is equal over sampling periods (this may be influenced by investigator proficiency as well as animal behavior and environmental conditions).

5. Sampling effort remains constant over sampling periods.

The two basic removal methods are the change-in-ratio (CIR) method and the catch-per-unit effort (C/E) method. Change-in-ratio estimators are useful when the species of interest can be divided into two classes (e.g., males vs. females or juveniles vs. adults). One of the two classes is selectively removed during sampling periods and the ratio of the two classes is compared before and after this removal. Detailed descriptions of this method can be found in Seber (1973) and Udevitz and Pollock (1991).

Catch per unit effort methods use the idea that fewer animals will be caught per unit effort as increasing numbers of animals are removed from the study area. Examination of the decline in catch per unit effort in subsequent sampling leads to a predicted total catch where catch per unit effort will equal zero. This catch, estimated through linear regression, is an estimate of the population size before removals began. Detailed descriptions of this method are given by Overton and Davis (1969) and Seber (1973). Program CAPTURE (Rexstad and Burnham 1991) provides for simple analysis of removal sampling data.

DIVERSITY INDICES

Species diversity is a community property that takes into account both the number of species (richness) and their relative proportions (evenness). Since relative proportions are necessary to calculate diversity, a simple species list will not suffice and must be supplemented with population estimates or other measures of relative abundance. A single operational definition for diversity does not exist, yet a number of indices have been devised that weigh richness and evenness to produce a diversity metric that can be compared across sites or across time intervals.

One of the most widely used indices is the Shannon index which calculates diversity (H) as follows:

$$H = \sum_{i=1}^{s} p_i \ln p_i$$

p_i = number of individuals captured from species i / total number of individuals captured (n_i / N).

Like all diversity indices, the Shannon index increases as the number of species increases and the relative abundances of species become more similar. Another basic diversity index is Simpson's index which calculates diversity (c) based on the probability of two captured individuals being of the same species. It is computed as follows:

$$C = \sum_{i=1}^{s} p_i^2$$

Diversity may also be estimated by a number of other indices (see Hayek 1994) and each will rate a given community differently depending on how richness and evenness are weighted. This betrays the fact that no single index can effectively rank communities based on their composition. Since the relative importance of richness and abundance distributions depends on complex ecological interactions that, for the most part, remain unknown, any single index is some what arbitrary and should thus be applied with caution.

OCCUPANCY MODELS

Occupancy is the proportion of sites occupied by a species. Monitoring occupancy is an ideal way to determine the status of species over a large geographic area and may be well suited to species that have large population fluctuations that are not indicative of general population trends (e.g., many amphibian species). As discussed above, detection of species is never perfect; a recorded absence may be a true absence or simply a failed detection. Thus detection probabilities need to be taken into account when estimating occupancy.

Occupancy models estimate detectability by using information from repeated observations at each site. The necessary data for analysis are simply a record of whether a species was detected or not during each visit to each site. Maximum likelihood techniques are then used to estimate model parameters, including occupancy and detectability, from these capture histories.

The primary assumptions of occupancy models are:

1. Closed occupancy state. No changes in presence or absence occur during a sampling period.

2. Independence of sites. Detection at one site does not influence detection at another site.

3. No unexplained heterogeneity in occupancy. Differences in occupancy probability must be explained through the use of covariates.

4. No unexplained heterogeneity in detectability. Detectability should be the same across sites or be related to a measured covariate (e.g., total vegetative cover).

To meet these assumptions, the study must be carefully designed (see Chapter 3). Timing and location of surveys must be well chosen to maximize closure and independence of sites. Standardized sampling needs to be implemented to reduce differences in detectability. A solid knowledge of the target species' natural history is necessary for collecting covariate data for modeling differences in occupancy and detectability.

The details of the mathematics of occupancy models are beyond the scope of this chapter. However, programs PRESENCE and MARK are both capable of performing occupancy analysis and are freely available (see Software box in this chapter). For further information see MacKenzie et al. (2002), MacKenzie et al. (2003), MacKenzie et al. 2004, and a USGS fact sheet on using occupancy models (http://fresc.usgs.gov/products/papers/1443_adams.pdf).

Detection Probability

Knowing the detection probability (DP) is critical to designing and interpreting biotic surveys (Mazerolle et al 2007). Survey objectives generally fall into two categories. The survey manager may be interested in a) determining whether or not a species is actually present at a specific site (often a specific property), or b) tracking trends in species occupancy among many sites (without knowing exactly which sites are occupied). Estimating population size and trends is not addressed here, as these require tracking and counting individuals and different methods, study designs, and analyses. This chart (see pg. 211) assists in determining needed sampling effort for the above two objectives, given a known average detection probability (DP) for a particular species survey method.

Note that DPs are variable, and selection of a survey method with low DP variance for the species of interest is very important (Mackenzie and Royle 2005). For determining needed sampling effort use an average DP derived from many sites and preferably across several years. The number of published DPs grows annually, and these may be substituted for original local data if local conditions are similar to published studies. Survey protocols should provide detailed instructions designed to minimize variance from variables such as time of day of sampling, weather conditions, etc. Periodic recalibration of DPs is recommended through over-sampling, especially if variables change which are suspected to affect DPs (i.e. habitat or climate changes). Automated audio recording units are a relatively cheap way to obtain large sample sizes for calculating DPs for species which have audible calls.

Example 1: Is species X present or not? The land manager wants to determine how many samples are needed to know if a species is present or absent on a specific site. For a given DP, the minimum number of samples needed is shown for 75%, 85%, and 95% confidence in the survey results. For example, to be 95% sure that a species with an average DP of 0.5 is not overlooked, conduct 5 samples.

Example 2: I can only afford to run 3 surveys, what species will this detect? Note that survey effort is more often dictated by real world capacity (money and manpower) than by theoretical study design. With 3 samples, from the chart we see that species with DPs >0.60 will be detected with certainty if present (95% confidence), and not require corrections for false negatives. So if these species are not recorded they can be considered absent. Species with DPs <0.15 are so poorly detected at this level of effort that negative results are essentially meaningless, i.e. negative results do not mean the species are absent (note that for 1 sample this threshold is 0.3 and would apply to most existing frog calling survey programs). Species with DPs between these extremes can be used for occupancy modeling, and negative results provide various probabilities of having missed species that are actually present. For example, with 3 samples and a DP of 0.40, confidence is 78%, so this species will be missed even when present in 2.2 out of every 10 surveys, on average. Of course, when species are detected they add to cumulative species richness lists for sites (i.e. checklists), so positive detections always have some value.

N Samples and confidence achieved for a given DP

Average DP	Min. N Samples for 75% Conf.	Min. N Samples for 85% Conf.	Min. N Samples for 95% Conf.	1	2	3	4	5	6	7	8	9	10
0.10	14	19	29	10%	19%	27%	34%	41%	47%	52%	57%	61%	65%
0.15	9	12	19	15%	28%	39%	48%	56%	62%	68%	73%	77%	80%
0.20	7	9	14	20%	36%	49%	59%	67%	74%	79%	83%	87%	89%
0.25	5	7	11	25%	44%	58%	68%	76%	82%	87%	90%	92%	94%
0.30	4	6	9	30%	51%	66%	76%	83%	88%	92%	94%	96%	97%
0.35	4	5	7	35%	58%	73%	82%	88%	92%	95%	97%	98%	99%
0.40	3	4	6	40%	64%	78%	87%	92%	95%	97%	98%	99%	99%
0.45	3	4	6	45%	70%	83%	91%	95%	97%	98%	99%	100%	100%
0.50	2	3	5	50%	75%	88%	94%	97%	98%	99%	100%	100%	100%
0.55	2	3	4	55%	80%	91%	96%	98%	99%	100%	100%	100%	100%
0.60	2	3	4	60%	84%	94%	97%	99%	100%	100%	100%	100%	100%
0.65	2	2	3	65%	88%	96%	98%	99%	100%	100%	100%	100%	100%
0.70	2	2	3	70%	91%	97%	99%	100%	100%	100%	100%	100%	100%
0.75	1	2	3	75%	94%	98%	100%	100%	100%	100%	100%	100%	100%
0.80	1	2	2	80%	96%	99%	100%	100%	100%	100%	100%	100%	100%
0.85	1	1	2	85%	98%	100%	100%	100%	100%	100%	100%	100%	100%
0.90	1	1	2	90%	99%	100%	100%	100%	100%	100%	100%	100%	100%
0.95	1	1	1	95%	100%	100%	100%	100%	100%	100%	100%	100%	100%

survey effort above double line (confidence <0.3) is not recommended, and is insufficient even for occupancy modeling

below thick line 95% confidence is achieved with no need to correct for false negatives

Minimum number of samples needed = LOG(significance level)/LOG(1-DP)
Percent confidence in detecting species = 1-(1-DP)^(N samples)

Green treefrog (*Hyla cinerea)*

Tom Luhring

CHAPTER 7. CONCLUSIONS AND RECOMMENDATIONS

Gabrielle J. Graeter

The intention in writing this guide was to offer a resource that a wide array of people would find useful, including land managers, private land owners, biologists, and conservationists. Our main objectives included providing: (1) the information necessary to initiate an inventory or monitoring program, including descriptions of sampling design and guidance on study planning, (2) comprehensive summaries of available techniques, including practical advice on the use of each technique for inventory and monitoring programs, (3) an outline of the basic statistical options available and sources for more thorough descriptions of statistical methods, and (4) a large amount of supplementary information in the appendix sections. For many users, the most useful aspect of the guide may be the extensive Species x Techniques Table 5.1, which details the techniques recommended by experts for each species and life stage in the United States. Throughout the guide, we have also made a concerted effort to include helpful resources on each topic.

THE IMPORTANCE OF INVENTORY DATA

An inventory is the first step that must be taken in a particular area or for a particular species, population, or community to establish what habitats are present and what species of amphibians and reptiles occupy them. Data from an inventory program provide the baseline information necessary for future herpetofaunal monitoring or research projects. For example, a researcher must know if a particular species is present in great enough numbers to conduct a study. Similarly, having information on habitat types within a potential research area is critical. Once an inventory is completed, monitoring of the populations present in a given area can be initiated.

Inventory data are a highly valued resource for many biologists and land managers. Knowing species' geographic ranges and the condition and status of a species or population is critical for making even basic management decisions. Some conservation decisions rely heavily on data from inventories and a thorough and recent inventory allows for better

informed conservation plans. Knowing whether a particular species occurs in a certain area is considered essential information to land managers and biologists working on management and conservation plans.

THE VALUE OF MONITORING

Monitoring is valuable in providing a fuller picture of the condition and viability of a species, population, or community in a study area than is accomplished with basic inventory. Monitoring can reveal patterns that are not apparent from a single inventory event or an assessment done over only one season or even one year. With monitoring, the greatest threats and issues for a particular species or population can be identified and information can be gathered to make prudent management and conservation decisions. With monitoring, information can also be obtained on community interactions and measures of diversity.

Long-term monitoring can be particularly valuable. When funding, time, and personnel are plentiful enough to continue a monitoring program over many years, patterns often begin to appear from within the noise of a variety of different factors. This may be especially true with amphibians and reptiles due to highly explosive breeding in some species, extremely variable breeding and behavior from one year to the next, and behavior strongly tied to climatic patterns and variability. Understanding the responses of amphibians and reptiles to weather and climatic factors over long periods of time will allow us to better understand and predict herpetofaunal responses to shifts that are likely to happen as global climate change occurs. Long-term monitoring can also help discern between natural changes in a population and anthropogenic changes, which should help in minimizing and mitigating human based effects. For example, through long term monitoring we can begin to distinguish the natural cycles and stochasticity of breeding amphibians from human related threats, such as the effects of habitat loss and fragmentation. Observing the effects of land-use changes, environmental pollution, or any other human-caused changes to a species' habitat can also help us understand their responses to these changes. The more that land managers and biologists understand about the responses these animals have to habitat changes, the better equipped they are to prescribe and implement effective management plans and conservation strategies.

MODIFICATION OF TECHNIQUES

With standardization of methods, similar datasets from programs in different regions can be compared. Rising concerns over declining amphibian and reptile populations throughout the United States and the world make the ability to compare datasets a great asset. Thus, we recommend that, whenever possible, these techniques be implemented in the manner previously established in prior publications and summarized herein. However, we recognize that some situations, species, and locations will require that a technique be modified.

Techniques may need to be changed slightly based on a variety of factors, including the particular objectives of a program, the landscape, environmental conditions, seasonal differences, target species, funding, personnel constraints, and equipment available. The first step to determining whether a particular technique will require modification is to go to the field site and walk through the logistical steps of setting up a particular study, while carefully considering any potential hurdles that could be encountered. Next, consulting with other biologists or experts on the techniques to be used can often make sampling more efficient and effective. The third step is to do a test run of techniques at the field site with the study species. A test run is highly recommended because it will allow alteration of the set-up and protocol before investing the full funds and personnel necessary for the project. In many cases, slight adjustments to a technique may be necessary after a sampling program is underway, due to factors such as a particularly dry or wet sampling season that were unable to be anticipated in advance.

DISSEMINATION OF RESULTS

In addition to a land manager's own reasons for doing inventory and monitoring work, the information collected can benefit others, from land managers and conservation planners to research herpetologists. To maximize the usefulness of the data collected, in almost all cases it is advantageous to publish the findings in a form accessible to others. The format and type of publication will depend on the type and amount of information available. Some collected information (e.g., auditory call surveys) is best suited for an online database. Publications in scientific journals or as government agency reports or in-house manu-

als or guides may also be appropriate. Having draft manuscripts of any planned publication assessed and critiqued by outside parties usually results in a stonger, more reliable document.

EPILOGUE

This book was written with the aim of providing a user-friendly guide to the techniques used for inventorying and monitoring amphibians and reptiles of the United States. The techniques previously established by others have been summarized in a practical manner, with an explanation of limitations, recommendations for use in inventory versus monitoring programs, and a list of necessary equipment. This guide is unique in providing a detailed table of recommended sampling techniques for specific life stages of every U.S. species. Another key feature is that the guide was written and formatted to be complementary with the Partners in Amphibian and Reptile Conservation (PARC) Habitat Management Guidelines (HMG) publications. Our target audience is broad, including land managers, private land owners, government agencies, and ecologists who are involved in herpetological studies. We have created this PARC guide with the aim of increasing communication among diverse public and private groups and individuals interested in amphibian and reptile conservation.

APPENDIX I. HANDLING LIVE AMPHIBIANS AND REPTILES

Brian D. Todd and Xavier A. Glaudas

INTRODUCTION

Handling live animals is one of many aspects of scientific studies. Data need to be collected and gathered to provide the raw material for research. Handling live animals should only be performed with an objective in mind (e.g., capturing the animal, taking measurements). The less they are exposed to handling, the better. Two safety issues should be considered when handling amphibians and reptiles. First, the handler's safety: like many animals, reptiles and amphibians can potentially inflict harm to a person, and injuries can prove life threatening or cause permanent damages in some cases (e.g., venomous snakes, crocodilians). Second, the animal's welfare is also important and there are techniques that eliminate or minimize the risk of injuring or stressing the animal while handling it. Following the guidelines provided below will help you become successful at handling amphibians and reptiles.

HYGIENE AND HERPETOFAUNA

It is advisable to wash your hands thoroughly before and after handling reptiles or amphibians. Many wild-caught animals may carry diseases that, although harmless to humans, are highly transmissible and potentially fatal to conspecifics (e.g., upper respiratory tract disease in gopher tortoises, Seigel et al. 2003; viruses and chytrid fungus in amphibians, Green et al. 2002). In some cases, the extent of such diseases in wild animal populations may be much greater than reported or previously assumed. Careful hygiene is a must if diseases such as these are to be prevented from spreading from animal to animal or population to population. It is also important to understand that many of these diseases may be carried by an animal that does not show any symptoms. One recommended precaution is to carefully disinfect any materials, equipment, and outerwear (boots, etc.) that you will use at multiple locations between visits to different sites. Bleach is one such commonly used disinfectant. Additionally, many amphibians have highly porous skin that can absorb toxins and chemicals from a person's hands. Clean hands prevent injury to susceptible amphibians.

From a human health perspective, hygiene is equally important. The US Centers for Disease Control and Prevention report several instances of amphibians and reptiles transmitting *Salmonella* to people (reports available at http://www.cdc.gov). Although this risk is relatively minor, precautions such as washing with an antibacterial soap following handling can further minimize the risk of transmission to humans. Additionally, although skin toxins from most North American amphibians are rarely dangerous, they may cause irritation or burning to a handler's mucous membranes or eyes. Careful hygiene before and after handling herpetofauna can prevent problems for both the animal and the handler.

HANDLING

Amphibians

Large frogs should be held with your fingers and thumb encircling the waist of the animal (Fig. 8-1). The strong legs of many frogs can facilitate a quick escape if held in another manner. However, small frogs can be held by their two hind limbs. Large salamanders should be held with the entire hand, gently restraining the animal between the thumb and the fingers just behind the head in a fashion similar to medium-sized lizards (Fig. 8-2). Small salamanders can be held between gently clasped hands or in a gently clenched fist. Large aquatic salamanders can be extremely slippery and it can be easier to handle them with a rag and both hands. Amphiumas rarely bite but are alleged to "bite savagely" and therefore should be given respect when handled (Conant and Collins 1998).

Kurt A. Buhlmann

Figure 8-1. The proper way to hold a frog.

Lizards

Lizards should be secured between the thumb and the fingers just behind the head, firmly but gently (Fig. 8-3). Some lizards will vigorously bite in defense (e.g., *Eumeces*) but bites are rarely painful. Lizards should not be captured by the tail given that it might break off and potentially affect their fitness (Downes and Shine 2001; Chapple et al. 2002). Even more caution should be used when handling legless lizards (*Ophisaurus*) considering that their tails represent most of the total body length (Fig. 8-4).

Snakes

Prior to capturing or handling a snake, identification of the species is required. Handling venomous snakes requires specific techniques and equipment not necessary when handling non-venomous species. Never handle a snake that cannot be identified.

Whenever possible, avoid restraining non-venomous snakes behind the head so as to avoid stressing the animal unnecessarily. Some species of snakes will let you handle them without biting. However, other snakes are very active at defending themselves and would not hesitate to bite (e.g., *Coluber constrictor, Nerodia, Masticophis*). As with lizards, securing the head between the thumb and the fingers is the best option (Fig. 8-5). Never let the body flail about freely because it can injure the spine of the animal.

Venomous snakes should only be handled with a snake hook or snake tongs. Handling or capturing venomous snakes by hand can have disastrous consequences. Pit vipers strike quickly, and can reach

Gabrielle Graeter

Figure 8-2. Proper handling techniques for large salamanders is shown here with a hellbender (*Cryptobranchus alleganiensis*).

Sarah Foster

Figure 8-3. The proper way to hold a lizard.

Kurt A. Buhlmann

Figure 8-4. When handling lizards, you must take care not to grasp or hold them by their tail because it can break off. Be especially careful to handle legless lizards properly, as shown in this photograph.

Jamie Bettaso

Figure 8-5. The proper way to hold a snake.

Gabrielle Graeter

Figure 8-6. Snake hook, bag, and bucket

about half of their body size. Consequently, a safe distance between the snake and the handler should be observed. The best equipment to handle or capture a venomous snake consists of a bag, a snake hook or tongs, and a bucket in which to place the bagged animal (Fig. 8-6).

Turtles

All turtles have powerful jaws, although bites can easily be avoided if fingers, or other parts, are kept away from the head. Most turtles can safely be held at mid-body (e.g., *Kinosternon*, *Sternotherus*, *Trachemys*, *Glyptemys*; Fig. 8-7). Additional attention should be given to their claws, which can be elongated and inflict deep wounds, especially snapping turtles (*Chelydra serpentina*). As suggested by their specific scientific name, snapping turtles have long necks and should not be held by the front of the carapace. The best way to handle them is to wear thick gloves and grab the carapace near the tail (Fig. 8-8).

Sarah Snyder/ Doris Jwo

Figure 8-7. The proper way to hold a turtle

Gabrielle Graeter

Figure 8-8. A large snapping turtle should be held by the shell or rear legs, not the tail, so as not to injure it.

Kurt Buhlmann

Figure 8-9. For safety while handling, be sure to secure the jaws of crocodilians with rope or duct tape.

Crocodilians (including alligators)

Crocodilians of all sizes should be handled very cautiously. Not only do they possess powerful jaws, but they also have a very muscular tail that can be used defensively. Crocodilians can be caught by hand (small specimens), by noosing the specimen around the neck with a noose, or with a snare trap (Murphy and Fendley 1975). Small crocodilians can be grabbed behind the head with one hand, while the other hand supports the weight of the animal (usually at the base of the tail). To ensure safer handling while making measurements and recording data, jaws should be closed securely (Fig. 8-9). In case handling is relatively long, eyes should be covered to reduce stress on the animal. For larger specimens, several people are required to immobilize the animal and to handle it.

MEASUREMENT

Amphibians, lizards, snakes, and crocodilians

The standardized measurement used for amphibians, squamate reptiles, and crocodilians is snout-vent length. Snout-vent length (SVL) is defined as the distance from the tip of the head to the end of the cloaca (Fig. 8-10A). For taking measurements on venomous snakes, two practical and safe methods exist: the use of an acrylic tube (Walczak 1991) or of a squeeze box (Quinn and Jones 1974). In addition, tail length (from the cloaca to the tip of the tail [TL]) is also recorded. Total length is then equal to SVL + TL (Fig. 8-10B). Tail length is rarely recorded for amphibians (obviously in the case of frogs), but may be recorded for salamanders if desired. Tail length is typically recorded for snakes, lizards, and crocodilians. Snout-vent length is used as the standard measurement because salamanders and squamate reptiles can lose parts of their tails as a result of defensive autotomy, predation, or intraspecific aggression.

All animals can be weighed using either a spring scale or an electronic scale. In the field, specimens can be bagged and weighed using a spring scale. For large alligators, a truck scale must be used.

Turtles

Techniques for measuring chelonians are varied and differ according to species. To measure snapping turtles (*Chelydra serpentina*) or musk turtles (*Sternotherus*) in which the plastron is highly reduced, carapace length is preferred. However, for most other species, plastron length is favored. For both carapace and plastron length, measurements can be taken as

straight-line or curved depending on what researchers are interested in (Fig. 8-11). For straight-line measurements, a caliper is the best tool. For curved measurements, a flexible ruler or a cord is used.

ANESTHESIA

General information

The effects of anesthesia are often species-specific and can also depend on the metabolism of the animal, which in ectotherms is greatly affected by temperature. Therefore, you should not extrapolate usage guidelines or doses from endothermic animals such as mammals or birds. Anesthetizing amphibians and reptiles should only be done for surgery (e.g., implantation of a radio-transmitter, marking, or blood collection) and you should check the published literature for possible recommendations regarding your species. Wright (2001) and Mader (1996) provide more information on amphibian and reptile anesthesia, including various anesthetic protocols that have previously been used with various species.

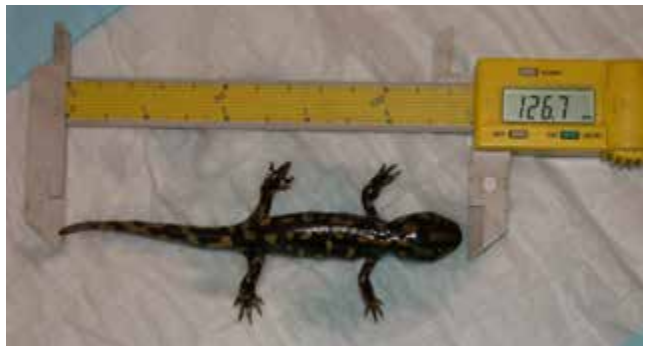
Anesthesia may be performed through inhalant or injectable anesthetics, or in the case of amphibians, through contact with buffered solutions of dissolved anesthetics. Anesthesia with inhalants is usually preferred for reptiles because recovery is often more rapid with this technique than with injectables (Bennett 1991; 1996). Moreover, effects of injectable anesthetics can be much less predictable.

Amphibians

The skin of nearly all amphibians is highly permeable, making dissolved anesthetics highly effective for both immersed adults and completely aquatic larvae. Tricaine methanesulfonate (TMS), also known as MS-222®, is the most commonly used anesthetic for amphibians. The dose should be calibrated for the species and size of the animal (possibly in previous test cases before applying in a research framework) but typical solutions range from 0.03 – 0.05% dissolved TMS in water. Tricaine methanesulfonate is acidic when dissolved in water and exposure of the unbuffered solution to amphibians may harm the skin of the animal or cause trauma and pain. Thus, it is important to buffer the solution, usually with sodium bicarbonate, and check to make sure the solution is pH neutral at or around 7.0. Wright (2001) provides information on recommended dosages and suggested buffering procedures. Pre-mixing solutions before they are required can prevent last minute guesswork in solution preparation.

Gabrielle Graeter

Figure 8-10A. Snout-vent length (SVL) of a hellbender (*Cryptobranchus alleganiensis*) being measured.



Valorie Titus

Figure 8-10B. Total Length of a tiger salamander (*Ambystoma tigrinum*) being measured.

Kitty Spivey

Figure 8-11. Biologist measuring carapace length of an alligator snapping turtle (*Macroclemys temminckii*) using large calipers.

Joe Mitchell

Supplies and tools needed for taking basic data at the time of capture. Includes a field notebook, caliper, measuring tape and ruler, Pesola mass scales, and a pencil, datasheet, and clipboard.

The animal should be immersed in the solution (Fig. 8-12) but prevented from drowning (not a problem for aquatic larvae except possibly late-stage larvae) until it becomes unresponsive and limp. When the animal can no longer right itself when placed upside down, or if it does not respond to toe pinches, it is properly anesthetized. The animal should be carefully monitored during the entire time it is anesthetized. During procedures, amphibians must be kept moist and prevented from drying out, particularly for animals with significant skin respiration (e.g., plethodontid salamanders). Following the procedure, amphibians should be gently rinsed in clean water. Animals may take up to a day to fully recover and should be kept cool with clean water. All animals should be fully responsive before they are released.

Reptiles

Inhalant anesthetics include isoflurane, halothane, and methoxyflurane. The two primary techniques that are used are chamber induction or inhalant intubation. Chamber induction has the disadvantage of requiring a long period of time before the specimen is actually anesthetized because of the low respiration rates of some reptiles. Additionally, some agents can be harmful to specimens if contact is made. Inhalant intubation is quicker, but it is more stressful to the animals. Using a speculum to prevent bites is usually recommended when animals are intubated (particularly important with venomous reptiles). Reptiles may be sedated before the intubation (e.g., Acepromazine). Intubation is highly recommended for longer surgeries as opposed to relatively quick, minor procedures.

The most commonly used injectable anesthetics are Ketamine and Telazol; barbiturates should be avoided. Injections should be made into the muscle of the forebody of the animal. The appropriate dosage varies by species, the surgery required, and the injection site (but see Mader 1996 for recommendations).

Reptiles generally recover more slowly than endotherms because of their low metabolic rates. Anesthesia and surgery should be performed at optimal temperature, which varies according to species. After surgery, the animal should be kept in a clean, warm, and dark environment for at least 24 hours (even longer if injectable anesthetics were used) and should be frequently monitored.

Valorie Titus

Figure 8-12. Amphibians should be immersed in anesthetic solution until unresponsive and limp.

APPENDIX II. DETERMINATION OF AGE, SEX, AND REPRODUCTIVE CONDITION

Robert N. Reed, Thomas M. Luhring, and Anton D. Tucker

INTRODUCTION

Accurately assigning age, sex, and reproductive status to a majority of reptiles and amphibians is difficult, and often requires taxon-specific techniques. Herein, we discuss basic methods for determining these features. We recommend that investigators take the time to examine live and preserved specimens of pertinent taxa before undertaking field research. If possible, investigators should complete a short apprenticeship with an expert, so as to better learn the nuances and common pitfalls associated with each technique. Lastly, we note that this brief account necessarily omits some topics; we recommend consulting more detailed accounts (e.g., Duellman and Trueb 1986, DeNardo 1996) for additional taxon-specific information.

DETERMINATION OF AGE

Frogs and toads

Marking and recapturing individuals of known age is the most reliable method of aging frogs and toads. This is achieved by marking a cohort of animals of known age (e.g., recent metamorphs) and recapturing them later at regular time intervals (see Durham and Bennett 1963). Skeletochronology (determining age of individuals from examination of bones) has been

successfully employed as a reliable aging technique on several anuran species (see Sagor et al. 1998). Under microscopic examination, differing rates of bone growth are visualized as wide zones reflecting periods of active bone growth, annuli reflecting periods of slow bone growth, and lines of arrested growth appearing within or bordering the annuli (Castanet et al. 1993; note that terminology varies by author, e.g., Zug and Rand 1987 use slightly different terms). Skeletochronology can be conducted on anuran phalanges and is thus readily coupled with ongoing mark-recapture studies that involve toe-clipping (see Appendix III - Marking Amphibians and Reptiles). Body size is not necessarily a reliable indicator of the age of an individual and should be used with caution unless previous studies warrant confidence in such a correlation (Halliday and Verrell 1988).

Salamanders

Techniques for aging salamanders are almost identical to those used for frogs and toads (see above). Although Halliday and Verrell (1988) found that body size is not always a trustworthy indicator of age, some species of salamanders do have a strong correlation between body size and age (see Parham et al. 1996).

Snakes and lizards

The most reliable method of determining age of free-ranging individuals is by marking and releasing neonates, followed by subsequent recaptures of the same individuals (see appendix section on marking methodologies). In studies of shorter duration, skeletochronology can be used. This approach has been used infrequently in squamate taxa as compared to its use in turtles and crocodilians. In long-lived reptile species, the technique is complicated by two main factors: 1) Growth rates typically decline in older individuals, such that annuli are closely spaced and difficult to distinguish; and 2) Internal bone layers may be resorbed over time.

Turtles

A basic approach for estimating the age of a turtle is to assign age from counts of growth marks or rings formed by the deposition of keratin to epidermal scute layers (Fig. 9-1). Exceptions can appear when turtle populations occupy relatively stable year-round thermal conditions (indistinct annuli; Tinkle 1958; Moll and Legler 1971) or when they are kept in captivity (multiple rings from feeding year round; Tracy and Tracy 1995). Other difficulties in interpreting the records of growth marks may arise when turtles are in unpredictable or unproductive habitats [e.g. difficulty in detecting overlapping rings under static growth conditions such as in droughts (Burbidge 1967) or from oligotrophic lakes (Georges 1985; Kennett and Georges 1990)]. Growth rings are more visible in juveniles but may be closely spaced or even overlapping in adults with slowed growth. Graham (1979) outlines the growth process to interpret growth rings. Periodic growth marks may also appear in laminar keratinized structures such as claws, which can assist age estimates with some emydids (Thomas et al. 1997). Wilson et al. (2003) offer a critical review of these techniques.

Gabrielle Graeter

Figure 9-1. The annuli on one scute of an adult eastern Box Turtle

Skeletochronology can be applied to live turtles by surgery and bone biopsy (Klinger et al. 1997), but is more commonly used post-mortem to investigate the skeletal elements. Skeletochronology is used most frequently in studies of long bones in sea turtles (Klinger and Musick 1995; Zug and Parham 1996; Parham and Zug 1997; Zug and Glor 1998) or the schlerotic ossicles of leatherbacks.

Crocodilians

Aging studies for crocodilians are established by clipping the tail scutes of hatchlings or adults with an individual or cohort numbering system. Mark-recapture studies then establish age and growth of individuals at

subsequent captures. Skeletochronology can be used with wild crocodilians or museum specimens using the long-bones of sacrificed or harvested animals or the non-destructive removal of osteoderms from free-living individuals (de Buffrénil 1982; Hutton 1986; Games 1990; Cooper-Preston 1992; Woodward and Moore 1992; Tucker 1997).

DETERMINATION OF SEX

Frogs and toads

The sex of frogs and toads is determined mostly by the presence or absence of secondary sexual characteristics. In many cases, external observation of an anuran in the breeding season will only tell the observer if the animal they are looking at is definitely a male. While dissecting individuals and looking for the presence of ovaries or testes is generally a fail-safe method, it is generally not a good technique for studies that require quick identification or the release of a live animal. One alternative to dissection is the use of a sonogram. Animals of breeding size that do not demonstrate "male-like" characters are generally assigned to the female gender. The one most obvious trait that makes a frog or toad a definite female is the presence of eggs. In the breeding season, eggs can often be seen through the skin of a gravid female. In smaller species or species with more opaque skin, eggs can be seen by extending the hind legs and gently moving the skin along the side of the animal. If eggs are present, they can sometimes be seen along the sides as small black dots that move independently from the overlying skin. The region of skin between the side of an anuran and the hind leg is often the best location to see eggs (Fig. 9-2). Care should be taken to be gentle with gravid females as rough handling could compromise her health or that of her clutch.

Tom Luhring

Figure 9-2. *Acris* with eggs showing through its skin

Male anurans have a diversity of secondary sexual characteristics that accompany or assist in calling and amplexing. However, males of some species demonstrate these characteristics only during that species' breeding season. The most reliable external characteristic in male *Rana* is the presence of an enlarged thumb (Wright 2002). A number of *Rana* males have larger tympanum than those of their female counterparts (see Conant and Collins 1998). In these species, the tympanum in males is larger than the eye while females have tympanum that are the same size as or smaller than the eye (Fig. 9-3). Wright (2002) states that there may be more of a yellowish tint to the throats of some male *Rana*.

Tom Luhring

Figure 9-3. The tympanum (i.e., eardrum) of many female ranids, including this bullfrog, *Rana catesbeiana*, is the same size or smaller than the eye. In males, the tympanum is larger than the eye.

Hylids are not easily sexed with any degree of accuracy outside the breeding season by external characteristics alone. During the breeding season, male hylids have loose folds of skin under their chin (Fig. 9-4). While *Pseudacris* exhibit darker throat patches than females during the breeding season (Fig. 9-5), the rest of the hylids do not always have sexually dimorphic throat coloration. However, a number of *Hyla* males may exhibit a different throat coloration than females in the same location and we recommend contacting an expert in the region of interest for site-specific characteristics.

Male bufonids exhibit dark throat patches (Fig. 9-6) and enlarged dark nuptial pads (Fig. 9-7) on their thumbs. Spadefoot toads (*Scaphiopus* sp) also exhibit darkened nuptial pads on their thumbs but can often fail to have darkened throat patches. *Gastrophryne* males in breeding condition generally have dark throat patches (Fig. 9-8) and are usually smaller than females. However, many individuals can demonstrate intermediate characteristics and be difficult to define as a male or female. If throat coloration is in question, then size is usually the next best defining characteristic as females are generally larger than males.

Tom Luhring

Figure 9-4. Loose folds of skin under the chin of a male hylid during the breeding season.

Tom Luhring

Figure 9-5. A male spring peeper, *Pseudacris crucifer*, with a dark throat patch.

Tom Luhring

Fig.ure 9-6. Many male bufonids, such as this southern toad, *Bufo terrestris*, have dark throat patches.

Tom Luhring

Figure 9-7. Dark nuptial pads on a spadefoot toad, *Scaphiopus holbrookii*.

Tom Luhring

Figure 9-8. During the breeding season, male *Gastrophryne* (here Eastern narrowmouth toad, *Gastrophryne carolinensis*) have dark throat patches.

Salamanders

Determining the sex of salamanders, as with frogs and toads, is done most effectively during the breeding season. Males of many species, when found outside of the breeding season, are nearly impossible to distinguish from females. Dissection may be the most reliable method of determining sex if external inspection is not conclusive. Eggs in females of some species can be seen through the skin and can be detected in the same manner as in female anurans (see above). Petranka (1998) provides detailed species accounts for salamanders. These accounts include species-specific characteristics (coloration, average size, etc.) that can be used to distinguish one sex from another.

During the breeding season, a male *Ambystoma* will usually exhibit a swollen cloaca (Fig. 9-9). Some species of *Ambystoma* are sexually dimorphic year-round with respect to size, coloration (Fig. 9-10), tail shape, and/or the presence/absence of specific internal cloaca characteristics. In addition to swollen cloacal openings like those of Ambystoma, proteids have two enlarged cloacal papillae that project posteriorly (Petranka 1998).

One of the more common features of species of plethodontid males is the presence of a mental gland that is usually found underneath the chin (Fig. 9-11). Males of a number of *Eurycea* species feature hedonic glands and enlarged cirri (Fig. 9-12). Many *Plethodon* males also exhibit enlarged cirri. *Aneides*, *Desmognathus, Eurycea, Hemidactylium* and *Plethodon* males may have a different jaw, tooth and/or cloacal morphology from their female counterparts and some can be distinguished by similar year-round characteristics as *Ambystoma* (see above).

Salamandrids demonstrate a suite of sexual dimorphisms that are species-specific (see Petranka 1998). Some of the more common male characteristics in this family include blackened ridges and pads on the inner thighs, colored gland clusters, hedonic pits, cornified toe tips, and swollen vents.

Aside from length differences in some species, male and female *Amphiuma, Gyrinophilus, Phaeognathus, Pseudotriton, Siren,* and *Stereochilus* are indistinguishable based on visual observation of external anatomy with the exception of the Three-toed Amphiuma, *Amphiuma tridactylum* (see Petranka 1998). While some authors suggest that *Siren* have enlarged masseter muscles (Hanlin and Mount 1978), Sorensen (2003) detected no such difference for greater sirens (*Siren lacertina*).

Bill Stagnaro

Figure 9-9. Swollen cloaca of male California tiger salamander (*Ambystoma californiense*).

John White

Figure 10. Female (on left) and male (on right). Marbled salamanders can often be distinguised by the brightness of the white crossbands on the back.

John Jensen

Figure 9-11. The mental gland of this male Pigeon Mountain salamander (*Plethodon petraeus*) is visible below the chin.

Tom Luhring

Figure 9-12. In many *Eurycea* species, males have enlarged cirri.

Snakes and lizards

Male squamates (all "lizards," including snakes) are distinguished by the presence of paired copulatory structures called hemipenes. These are inverted in invaginations at the base of the tail and one or the other is everted into the cloaca of a female to achieve copulation. In many taxa, the presence of hemipenes is indicated by a thickened tail base or prominent paired ventrolateral bulges immediately posterior to the cloaca, such that sex can be confidently assigned by visual examination. Hemipenes of many squamate taxa can be manually everted by pressure on the underside of the tail posterior to the cloaca. This technique is effective for small lizards and juvenile snakes, but is difficult for larger species and species with muscular tails. This technique may cause temporary or permanent injury when employed with excessive enthusiasm. Female snakes often have caudal scent glands, the papillae of which can be partially everted and mistaken for hemipenes. These papillae can be

differentiated from hemipenes by color, size, and shape, as scent glands are usually small, white, and somewhat pointed when everted, whereas hemipenes are larger, red or pinkish, and blunt when everted (Klauber 1956). Female squamates may also possess hemiclitori that become visible when pressure is applied to the tail; these tiny organs may conceivably be mistaken for hemipenes. When manual eversion is not possible, sex often can be determined by sliding a blunt probe into the cloacal region to detect an inverted hemipenis in the tail (Blanchard and Finster 1933; Schaefer 1934). If the probe can be inserted an appreciable distance, then the individual is a male (Fig. 9-13).

Sarah Foster

Figure 9-13. A snake being probed to determine sex.

Although probing sounds easy, in practice it can be problematic. Hemipenis length varies among taxa, as does the strength of surrounding muscle. Furthermore, females of many species have invaginations in the tail that can be mistaken for hemipenes. Schildger and Wicker (1989) showed that the depth of "hemipeneal" invaginations did not differ between sexes of lizards of three families. Ball-tipped metal probes are preferred for probing squamates, and are available in sets of various sizes. Plastic-tipped, straightened bobby pins have been advocated as ideal probing tools, as they are disposable after a single use and thus minimize risk of pathogen transfer between individuals (DeNardo 1996).

More advanced techniques may be used to determine sex when necessary. These techniques include ultrasonography (Morris and Alberts 1996; Morris et al. 1996; Wright et al. 1995), laparoscopy (Schildger and Wicker 1989; Cree et al. 1991), karyotypy (Bull 1980), and radiography (Shea and Reddacliff 1986; Card and Kluge 1995; Card and Mehaffey 1994).

The numbers, position, and morphology of dermal scales often varies by sex within a species. Male lizards of many taxonomic groups exhibit femoral pores that are larger or more numerous than those of females (Rivas and Avila 1996; note that pore size may be seasonally variable in males and sexually dimorphic only among adults), and may have additional sex-specific scalation characters (e.g., a pair of enlarged post-cloacal scales in many phrynosomatids, polychrotids, and relatives). Among some snakes, the number of ventral or subcaudal scales varies between sexes, as does the number of mid-body scale rows.

Joe Mitchell

The sex of some lizards can be determined by appearance alone. This is the case with the fence lizard, *Sceloporus undulatus*. On the left is a female with wavy, dark crosslines on the back. On the right is a male with a plain back and dark blue throat patch (throat patch not pictured).

Turtles

Determining the sex of an adult turtle is simple if secondary sexual characteristics are present. Adult males typically have a broader and longer tail with a more distally located vent than females (Fig. 9-14). Tail characteristics are unreliable with immature animals, however. With some turtles, the males bear a concave plastron whereas the females have a flat to slightly convex plastron (Fig. 9-14). Other species-specific marks of sex include eye color (e.g., *Terrapene carolina*, *Clemmys guttata;* Fig. 9-15), size dimorphism (e.g., *Graptemys*), nail elongation in adult males (e.g., *Terrapene*, *Trachemys scripta*), roughened patches

on the inner surface of the hind legs (*Kinosternon*), and melanism among older males (e.g., *Trachemys scripta*; Fig. 9-16); however, it is important to note that none of these characters by itself is foolproof as an indicator of sex.

When specimens are sacrificed or preserved, direct observations of the gonads will confirm sex. Non-destructive methods include laparoscopy, ultrasonography, or blood samples taken for hormonal assays

Jamie Bettaso

Figure 9-14. Female (on left) and male (on right) western pond turtle. Note longer, broader tail and concave plastron on male.

Gabrielle Graeter

Figure 9-15. Female (on left, brown eyes) and male (on right, red eyes) eastern box turtle.

John Jensen

Figure 9-16 Melanism is common in old male turtles, as seen in this male pond slider (*Trachemys scripta*).

Crocodilians

Sex can be determined by checking within the cloaca. With the animal safely restrained, a finger probed anteriorly into the cloaca can palpate for the presence of a penis, which is a larger structure than the clitoris. For hatchlings or yearlings, a blunt probe can be used as a speculum to gently spread the cloaca and enable inspection of the genitalia. In small specimens, females have an unpigmented and tapered cliteropenis, whereas a male penis will have a pigmented, bulbous tip (Webb et al. 1984). Males are generally larger than females, with a wider head and snout, a larger body size, and a thicker tail at the base.

DETERMINATION OF REPRODUCTIVE CONDITION

Frogs and toads

Age at first reproduction has been recorded for many species and Duellman and Trueb (1986) is a good starting point for anyone interested in a basic review of this topic for amphibians. Fortunately, determining the reproductive condition of most anurans can be accomplished relatively easily in the field with little to no harm to the subject.

Nearly all reproductively active male anurans call readily during their specific breeding season (e.g., Fig. 9-17). However, non-calling satellite males can be found in the general area of calling males and it cannot be assumed that a non-calling individual is female. Many times, a male or female anuran will illicit a release call or an alarm call when grabbed. *Bufo terrestris*, for example, will give off a chirping release call when grasped around the waist and many *Rana* give off alarm calls when they are disturbed before jumping into a body of water.

Tom Luhring

Figure 9-17. Pine woods treefrog, *Hyla femoralis*, calling from a branch.

Reproductive females often have egg masses visible underneath the skin (see determination of sex section) and recently spent females will appear to be unusually skinny with loose skin folds. Aside from estimating age or stage of development based on size, one of the only other ways to be sure of an individual anuran's reproductive condition outside of the breeding season is to sacrifice the animal for dissection and examine the ovaries or testes (see Duellman and Trueb 1986 for information on testicular and ovarian development as well as reproductive cycles in anurans).

Males exhibit a suite of secondary sexual characteristics when they are in breeding condition (see the determining sex section). While one can usually make a safe assumption that any animal displaying male secondary sex characteristics is a sexually mature male, failure to display such characteristics is by no means a guarantee that an individual is a female. However, in some species where the male is much smaller than the female and "male" characteristics are not always clear cut, size can be used to make an educated guess as to the sex of an individual. For example, some female *Gastrophryne carolinensis* will have a darkish colored throat. However, the males are usually somewhat smaller and a large individual with a questionable throat coloration is most likely a female. Caution should be used in making such inferences and we suggest working with someone that is familiar with each species and differentiating sexes before making inferences on one's own.

Salamanders

Determining reproductive condition in salamanders is basically the same as determining reproductive condition in frogs and toads. Secondary sexual characteristics are exhibited by males (see determining sex section) and breeding females are often full of eggs that can sometimes be seen through the skin. Unlike anuran males, male caudates do not advertise their presence with audible sounds. However, in species like the pond-breeding *Ambystoma*, mass immigration of reproductive adults into a wetland accompanies breeding activity. Mass emigrations from wetlands of metamorphosed juveniles also occur and these animals should not be confused with reproductive adults. Generally, if salamanders are entering a wetland en masse, they are likely reproductive adults.

As with frogs and toads, secondary sexual characteristics are markedly more pronounced in male salamanders during the height of that species' breeding season (see determining sex section). Nearly all reproductive terrestrial and semi-terrestrial male salamanders develop enlarged cloacal regions that are the result of the swelling of the cloacal glands (Duellman and Trueb 1986). Nuptial excrescences, hedonic glands, mental glands, fin structure and coloration, skin texture, enlarged premaxillary teeth and enlarged cirri are also signs of reproductive condition in salamanders (see Duellman and Trueb 1986 for a general overview of these characteristics). Petranka (1998) provides excellent species accounts of all North American salamanders that includes much information about each species' reproductive biology.

The secondary sexual characteristics that provide an indicator of an individual's reproductive condition during peak breeding activity are usually not discernable outside of the breeding season. Dissection of a sacrificed individual and observation of its testes or ovaries (see Duellman and Trueb 1986) can provide an investigator with information on that individual's development outside the breeding season. Of course, any technique that can efficiently provide such information without harm to the study animal is preferred. Sonograms may be a future non-lethal direction for gathering such information.

Snakes and lizards

Squamate reptiles display a bewildering variety of reproductive modes, seasons, and physiologies. Saint Girons (1985) succinctly summed up this variety by stating, "…the modes of lepidosaurian reproduction are diverse. Thus, some species are monestrous, whereas others are polyestrous; some are seasonal, whereas others are not; some are oviparous and others are viviparous; some are bisexual and others are parthenogenetic. Also, the time of embryonic development…is very variable, as is the length of time between mating and ovulation." Given the range of reproductive variation and a lack of solid reproductive data for the majority of species, generalizations about determination of sexual maturity and reproductive status of lepidosaurs are few and far between.

Color changes are frequently associated with the breeding season in squamates, such that reproductive condition can be determined visually (Cooper and Greenberg 1992; Fig. 9-18). Male lizards commonly exhibit seasonally striking color change, especially in the family Iguanidae (*sensu lato*). However, females may experience color changes during some reproductive seasons as well; for example, striking orange spots develop on the sides of adult females in the genera *Gambelia* and *Crotaphytus* during the

breeding season. Although various other factors may contribute to seasonal or short-term color changes (e.g., disruptive coloration, aggressive encounters, defensive behavior, background matching, ontogenetic changes, mimicry), seasonally bright and visually obvious color changes in lizards generally occur during times of reproductive activity, and are a good indicator of sexual maturity.

Tom Luhring

Figure 9-18. A male green anole, (*Anolis carolinensis*), displaying his dewlap is a clear indication of reproductive condition.

Direct observation of mating and confirmed fertilization of ova comprise the ideal indicator of sexual maturity, but mating is rarely observed in the field for most species. For male squamates, therefore, sexual maturity is usually confirmed via dissection. Measurement of testis size from a time-series of individuals may reveal seasonal changes in testis volume or mass relative to body size, presumably revealing reproductive season in those species wherein sperm production occurs during the mating season (Shine 1977). Mature sperm can be collected from males (and recently-mated females) by smearing cloacal fluid or the liquid resulting from a cloacal wash (using Ringer's solution or comparable saline fluid) onto a microscope slide. The stage of spermatogenesis in males can be determined via dissection and histological examination of testes and associated tissues from sacrificed males (sample methodologies can be found in Wilhoft 1963; Aldridge 1979; Zug et al. 1982; Aldridge and Brown 1995; and Ramirez-Pinilla et al. 2002).

Male squamates are characterized by a sexual segment of the kidney (SSK; Regaud and Policard 1903; Bishop 1959; Fox 1977; Sever et al. 2002, Sever and Hopkins 2005). The SSK is hypertrophied during the mating season in many species (Saint Girons 1982). Because spermiogenesis is often temporally dissociated from the mating season in snakes (Saint Girons 1982), examination of the SSK may be more reliable as an indicator of the mating season than is the presence of mature sperm. Examination of the SSK has thus far been performed exclusively via dissection of museum specimens, road-killed snakes, and sacrificed males.

Among female snakes and lizards, the presence of developing offspring is often easily detected. The presence of shelled eggs can be observed visually through the ventral skin of many thin-skinned lizards, although these must be distinguished from ventral fat bodies. Gravid females of diverse taxa may appear heavy-bodied, with noticeable distention in the posterior portions of the body. Among snakes, gentle palpation of the abdomen can reveal the number and size of follicles or embryos. This is accomplished by holding the female belly up and gently running a thumb in one direction along the venter, allowing the eggs to slide under the thumb one by one (Fitch 1987). Dissection of females can produce additional information on reproductive status. Females that have previously reproduced exhibit thickened muscular oviducts that often appear "stretched" as compared to virgin females, indicating that they have attained sexual maturity even if maturing follicles or embryos are not currently present. Counting and measuring the diameter of developing ovarian follicles can provide information on timing of follicular development. Once follicles have matured into mature ova, the ovarian walls rupture and ova migrate to the oviduct. They leave behind corpora lutea (structures that develop on the ruptured follicular walls in the ovary after extrusion of ova, also known as yellow bodies) indicating the number of ova released in the previous reproductive bout.

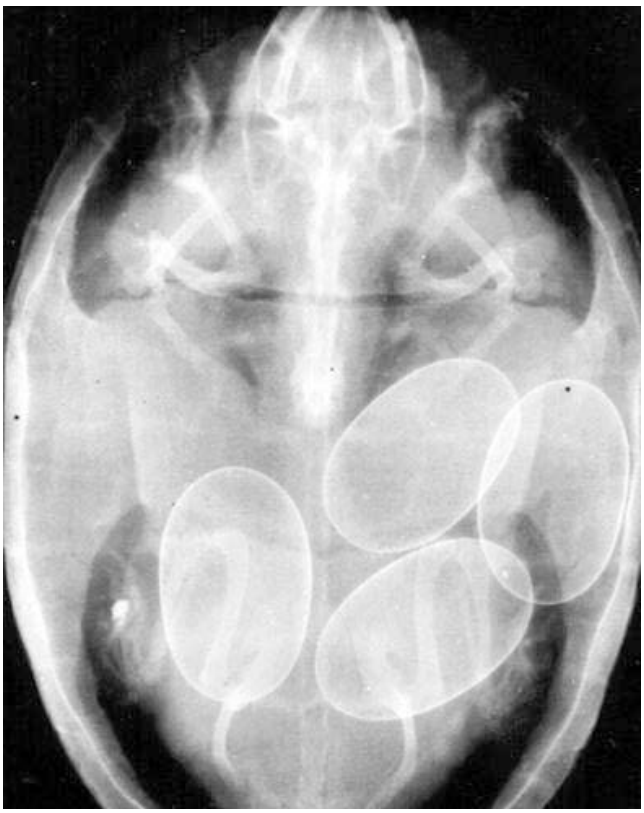
Turtles

Sexual maturity can be determined from the appearance of external sexual characteristics, by direct or indirect inspection of the gonads, or by following the status of gamete production and storage. For female turtles, maturity can be defined as a capacity for egg production in the current season, and for males, maturity is defined by the production of spermatozoa. However, neither size nor ages are absolute determinants of maturity per se, given the wide variance in minimum age or size at maturity across broad geographic scales or even within a population.

Palpation can detect eggs and assess sexual maturity in small, gravid females. With the turtle held upright, fingers are inserted into the inguinal space anterior to the hind limb, and the female is gently jiggled so that any oviductal eggs are felt as a tapping against the fingertips.

Male maturity is generally inferred once the tail elongates, but conclusive validation requires a direct visual or biopsy inspection of the gonad. Laparoscopy is preferred over sacrificing an animal for histological sectioning of the gonad. Semen collection by electroejaculation has been applied in a representative cross-section of sea turtles, tortoises, and freshwater turtles (Platz et al. 1980).

X-radiography has long been used to study shelled oviducal eggs in gravid females, to measure linear dimensions of eggs, and to count clutch size (Burbidge 1967; Gibbons and Greene 1979; see Fig. 9-19). These methods are thought to pose negligible threat to chelonians if radiation doses are reduced during exposure by use of cassette films with rare earth screens instead of Ready Pack film (Hinton et al. 1997). The recommended distances, duration, and intensity of x-ray exposures are species-specific but rules of thumb are available for small to medium-sized animals (Silverman and Janssen 1996; Hinton et al. 1997).



Whit Gibbons

Figure 9-19. X-ray of a female turtle, showing the number of developing eggs.

Ultrasound as a technique for assessing maturity is covered comprehensively by several authors (Rostal et al. 1990, Kuchling 1999). Laparoscopy is a surgical insertion of an optical scope through the body wall into the body cavity to permit direct viewing of the gonads in situ (Wood et al. 1983; Limpus and Reed 1985). With appropriately sized equipment, even individuals as small as 50 mm can be sexed by laparoscopy (Rostal et al. 1994). Although laparoscopy is a minimally invasive surgery, it is time intensive, requires specialized equipment, and should not be attempted without proper veterinary training and a thorough knowledge of turtle internal anatomy. A good overview of laparoscopy (and a variant known as soft tissue laparatomy) is found in Kuchling (1999).

Crocodilians

Reproductive maturity can be determined via ultrasound, laparoscopy, cloacal examinations (for females), and penile swabs (for males). The following section on techniques is biased toward *Crocodylus johnstoni* (Webb et al. 1983; Limpus 1984; Tucker and Limpus 1997) as a subject for all the methods mentioned. Nevertheless, the general anatomical appearance and methods will be similar for most crocodilians (Richardson et al. 2002).

Cloacal examinations consist of inserting a finger via the cloaca into the lower intestine, voiding any fluids, and palpating through the intestinal wall for follicles or eggs (Limpus 1984). Field exams for ultrasound or laparoscopies are performed with the animal restrained in dorsal recumbency to a board or ladder, or if the animal is large, some creative adjustments can be made to work in the prone position. Some researchers prefer to work with a local anaesthetic for smaller specimens (~2 m total length) that can be restrained securely. Larger specimens may be sedated (see Lloyd (1998) for the topic of anaesthesia) and at least 24 hr allotted for recovery after surgery.

Females are recorded as sexually mature if soft or hard-shelled eggs can be felt by palpation through the cloaca, the cloaca is flaccid as a result of recent egg passage, the oviduct is enlarged and highly convoluted, or if vitellogenic follicles or corpora lutea are observed via laparoscopy. A convoluted oviduct but no enlargement of previtellogenic follicles identifies a pubescent female. A white straight oviduct identifies a juvenile female.

Male maturity is confirmed by visual examination of the testis and associated ducts by laparoscopy. Adult males have a pendulous, sausage-shaped testis that protrudes from the body wall and a convoluted epididymis enlarged with sperm. Pubescent males have an elongated and ellipsoidal testis with little evidence of a convoluted epididymis. Juvenile males have an elongated, flat testis and a small, straight vas deferens. Males are recorded as adults when a mucus smear from the penile groove shows active spermiogenesis (Tucker et al. 1997).

APPENDIX III. MARKING AMPHIBIANS AND REPTILES

Xavier A. Glaudas and Kurt A. Buhlmann

Marking amphibians and reptiles is necessary to investigate migration patterns, survival rate, age, growth rate, and population size (Emery and Wydoski 1987; Donnelly et al. 1994). Marks can be temporary or permanent and can identify a captured animal as being from a particular time period or as a specific individual (Donnelly et al. 1994). Several methods can be used to mark amphibians and reptiles and many factors have to be considered before choosing the best-adapted technique (e.g., species, size, habitat, time, and resources).

Although some marking techniques are specific to a particular group of herpetofauna (e.g., notch marks on turtles), there is considerable overlap in the use of some marking methods (e.g., PIT tags and toe- or scale-clipping) amongst amphibians, squamates, crocodilians, and turtles.

For resource managers embarking on inventory projects, whether or not to mark animals is one of the first questions they should ask themselves. What is the goal? Will the rapid assessment inventory evolve into a long-term monitoring program? Are there research questions to be answered about a population in the future that would justify individually identifying animals during the initial inventory? For comprehensive inventories and population-level analysis monitoring programs, individually marking animals is a necessity.

AMPHIBIANS

Options abound for marking amphibians, but four methods currently stand out as the most widely used and accepted. The primary marking methods for marking and identifying amphibians are toe-clipping, visible implant elastomer (VIE) marking, PIT (passive integrated transponder) tagging, and pattern identification. See Donnelly et al. (1994) for a thorough historical summary of the many marking techniques employed with amphibians.

Toe-clipping

Toe-clipping consists of removing toes according to a marking scheme that allows for identification of captured individuals. Many different clipping schemes have been proposed for salamanders (Twitty 1966; D. B. Wake, in Donnelly 1994) and frogs (Martof 1953; Donnelly 1989; Hero 1989; Waichman 1992). To be effective, sharp scissors or nail clippers should be used, although nail clippers are usually less effective with small individuals because it is difficult to cut individual toes. In addition, rubbing alcohol is recommended for cleaning the scissors between animals (Donnelly et al. 1994). Some researchers suggest using antibiotic or antifungal creams or powders to ward off infections and to anesthetize salamanders before clipping toes (e.g., Donnelly et al. 1994). Ideally, this protocol would be followed (and certainly would for captive animals), but the demands of a study may not always allow enough time to follow this protocol (e.g., high number of captures on one day).

The advantages of toe-clipping include that individuals can be marked rapidly, the technique is relatively inexpensive because no complex equipment is required, and it allows for unique cohort or individual marks. However, this method is limited in several ways, including the potential harm to amphibians through infection and decreased growth, survival, or mobility (Clarke 1972; Golay and Durrer 1994; Halliday 1994, 1995; Reaser 1995; McCarthy and Parris 2004). Likewise, some amphibians regenerate their toes quite quickly (e.g., ambystomatid salamanders), so that reading the toe marks can become very challenging, especially for researchers with less experience identifying regenerated toes of a particular species. Furthermore, although toe-clipping is likely the most common marking technique used, it can be very difficult in some situations to obtain the proper permits to use this technique (e.g., some universities and organizations have relatively strict animal-use guidelines).

Visible implant elastomer (VIE)

The VIE marking technique involves implanting a small amount of fluorescent, non-toxic latex under the skin with a needle in a location where the fluorescent dye can be seen through the skin (Anholt et al. 1998; Binckley et al. 1998, www.pwrc.usgs.gov; Northwest Marine Technology 2000; see Fig. 10-1). Usually the tag is implanted on the back, abdomen, or inside forearm of an animal. Considerations for tag location include skin pigmentation and the looseness of the skin, which affects how much the tag moves around under the skin. This method may be preferred over toe-clipping because it does not involve removing toes and for the possible longevity of the marks. However, it has its own limitations, including that marks can be lost if they work their way out of the skin or move to an area where they cannot be read, that the injection

process and mark itself can be harmful to the animal (Bailey 2004), and that this technique may be difficult or ineffective to use with some species because of dark skin pigmentation or especially loose skin. Other disadvantages involve the time required when anesthetization is necessary, the limited amount of time one can work with mixed elastomers, and the inability to mark as many individuals at a time as with toe-clipping. A monitoring protocol has been established for long-toed salamanders (*Ambystoma macrodactylum*) using VIE (Pretzlaw et al. 2002).

John Jensen

Figure 10-1. Biologist injecting VIE (visible implant elastomer) into a gopher frog (*Rana capito*) tadpole.

A similar technique is the use of visible implant alphanumeric (VIA) tags for marking (Buchan et al. 2005), although it has been used less frequently than VIE for amphibians. This technique has the advantage of providing individual-specific marks.

PIT (passive integrated transponder) tagging

PIT tagging involves inserting a radio-frequency tag under the skin that transmits a numeric or alphanumeric code to a portable tag reader that displays the code (see Fig. 10-2). The tags are used for identifying a particular individual, not for long-range tracking, and the reader will only detect a signal when the tag is within 18 inches or less. PIT tags are helpful in that they give a definite positive identification of each captured individual and have an incredible number of unique codes available (up to 34 billion codes; Donnelly et al. 1994). To insert the tag, an implanter or spring-loaded syringe with a trigger can be used, but some cases will require a surgical incision. Tags and the implanter should be sterilized before use, and superglue or dissolvable sutures may be necessary to ensure proper healing after insertion.

The main limitation of this technique is its expensiveness (approximately $5 - $7 per tag and greater than $450 for each PIT tag reader), although some researchers have had difficulty with loss of tags via exiting out the incision opening, and the potential for problems related to the incision and insertion of the tag always exists. Another limitation is that some species and juveniles may be too small to safely use PIT tags in (Gibbons and Andrews 2004). Carefully considering the location of the tag and thoroughly learning the insertion methodology will help in reducing the likelihood of tag loss, damage to organs, and infections. Some studies have examined the effects of PIT tagging on the condition of the animals (e.g., Jehle and Hodl 1998; Ott and Scott 1999).

Gabrielle Graeter

Figure 10-2. PIT tag reader and PIT tag injector

Pattern identification

This technique is applied to some amphibians by recording their patterns with photographs or sketches (see citations in Donnelly et al. 1994; see Fig. 10-3), although a description-based technique has also been employed (e.g., Loafman 1991). More recently, pattern identification has gained attention because of the new possibilities that digital photography and computer-based image analysis programs offer (Church 2003; Bailey 2004)

A major advantage of this technique over all previously discussed marking methods is that it causes no harm to the animals. It can provide individual identification with some species, particularly those with unique patterns or spots, and identification is always possible because marks cannot be lost as with methods that involve tags (e.g., PIT tags, VIE) and

digit regeneration is not an issue (i.e., toe-clipping). However, in addition to a relatively longer handling time, simply identifying each individual can be time-consuming, especially when there are large numbers of identified individuals. The software development stage for image analysis programs can also be complex, difficult, and time-consuming. Furthermore, this technique is limited to those species with distinct patterns that do not change over time.

Jeff Humphries

Figure 10-3. Unique patterns on green salamanders, *Aneides aeneus*, can be used to identify individuals within a population.

SQUAMATES

Some of the same techniques for marking amphibians are applicable to squamates with some minor modifications to the marking scheme and methodology. For example, toe-clipping is used with lizards, but this technique is modified for snakes (e.g., scale-clipping). Likewise, PIT tagging and VIE have been used for lizards and snakes. However, freeze-branding and hot-branding are probably more commonly used with snakes than with amphibians. Temporary marks are described here for squamates, but have also been used some with amphibians.

Toe- and scale-clipping

Toe clipping is the standard method used on lizards. Each foot and toe is assigned a letter or a number and hundreds of unique combinations can be made (Woodbury 1956). Since snakes do not have limbs, one can clip either the ventral or subcaudal scales using a pair of scissors. The anal plate is used as the point of reference, and plates anterior or posterior to the anal plate are given numbers (Brown 1976). However, periodic sheds of marked individuals may obscure the identification number and the ID may disappear over time (Conant 1948; Tinkle 1957).

Freeze- and heat-branding

Scale clipping juveniles and small snakes may be problematic (Weary 1969). In this case, freeze-branding (Lewke 1974) or hot-branding (Weary 1969; Clark 1971) may be preferred (see Fig. 10-4). For freeze-branding, the instrument can be made of copper iron dipped into a liquid solution composed of equal parts of dry ice and 95% ethyl alcohol, liquid freon 12, or liquid Freon 22 (see Lewke 1974 for more information). For hot-branding, several devices have been used: pyrographic needles (Weary 1969), Hoskins Chromel "A" resistance wire (The Malin and Co., Cleveland, Ohio; Clark 1971), and High Temperature Medical Cautery Units (Aaron Medical Industries, Winne et al. 2006).

Sarah Foster

Figure 10-4. Hot branding equipment can be used for marking snakes

PIT tagging and VIE

Another alternative for marking squamates although costly, is the use of passive integrated transponder tags (PIT tags; see amphibian marking section, above) that are implanted into the animal's body (Gibbons and Andrews 2004) and allow for definite individual identification. Many of the limitations of this technique for amphibians are also true for reptiles (see amphibian marking section above). Visible implant elastomer (VIE tagging) has also been used to mark reptiles (Northwest Marine Technology, 2007).

Temporary marks

Whereas the methods mentioned above are permanent and necessitate catching the animal to identify the mark, some marking techniques allow researchers to identify an individual from a distance. Using a marker that can be seen from a distance, such as tape (Zwickel and Allison 1983), paint (Brown et al. 1984), plastic bird bands (Paulissen 1986), or beads (Fisher

and Muth 1989), has the advantage of not stressing the animal and eliminates the need to re-capture the animal. Beads are stitched to a surgical steel monofilament and then inserted with a hypodermic needle through the lizard's tail (see Fisher and Muth 1989 for detailed procedures).

However, there are at least two major drawbacks to using these temporary markers: some of these markers will only be effective for a short-term period since squamate reptiles shed their skin regularly, and juveniles do so more than adults. Second, it may affect the fitness of the individual even though several studies suggest markers do not affect survivorship in lizards and alligators (Jones and Ferguson 1980; Simon and Bissinger 1983; Jennings and David 1991).

CROCODILIANS

Crocodilians are generally marked using the same method of clipping toes or scutes as outlined above for lizards. Toe-clipping is usually used on juveniles whereas scute-clipping is more frequently performed on adults (Chabreck 1963; Murphy 1977). The location of scute-clipping is variable and some researchers have used the dorsal scutes (Chabreck 1963) or the ventral caudal scales (Gorzula 1978). Scute-clipping follows the same technique used for scale clipping: scutes are given letters or numbers and some are removed, providing the animal with a unique code. A distinction is made between one-edged and two-edged scutes, and the first one-edged scute (from the base of the tail) is used as the point of reference.

TURTLES

Turtles are perhaps the most easily monitored animal. The hard shell and scute pattern allows for a variety of permanent identification schemes. The turtle's shell is also suited to carry various devices including radio transmitters and temperature-sensing dataloggers. Turtles are long-lived animals, with longevities known to exceed 70 years in some species, based on actual field studies (Congdon et al. 1993). Marking turtles for individual identification has been standard practice for many herpetologists conducting field studies. A variety of identification systems have been used, and each researcher seems to have his or her preferred method.

Scute notching

Most North American hard-shelled turtles can be individually marked by notching or drilling a combination of the marginal scutes. The marginal scutes are found around the edges of the carapace (top shell). In most species, 12 scutes are found on each side of the carapace, not including the nuchal scute (the scute centered directly behind the head. Thus, there are 12 (left) and 12 (right) markable scutes. However, some genetic variation occurs in individuals and some turtles may have 11 or 13 scutes on a side. Whether that is important or not depends on the marking system the researcher implements. In some species the anal scute (directly above the tail) is divided, meaning that half can be attributed to each side of the carapace. In gopher tortoises (*Gopherus polyphemus*), the anal scute is not divided and is centered directly over the tail.

Each scute on each side of the carapace can be assigned a unique number or letter. Fig. 10-5 illustrates a marking system using letters, while Fig. 10-6 illustrates a system using numbers. Other systems exist and many variations on each system have been developed.

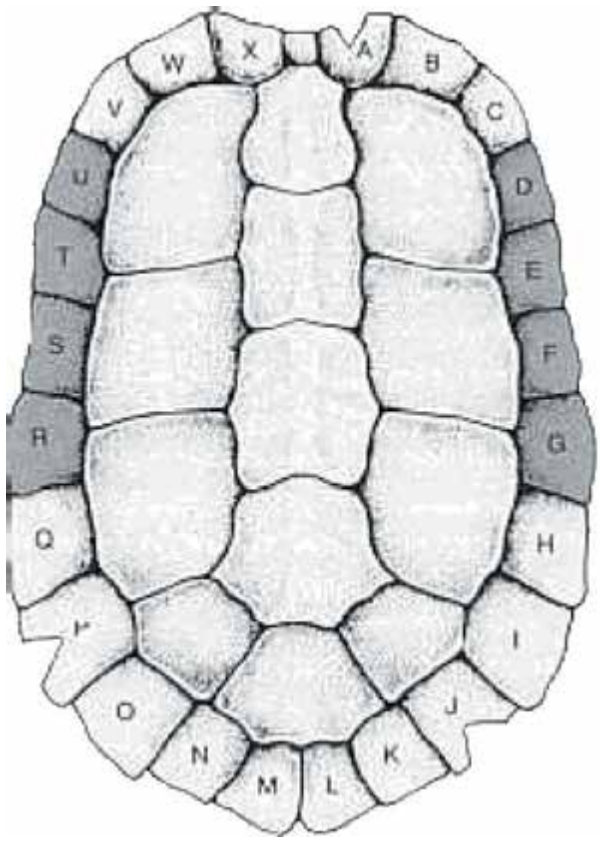
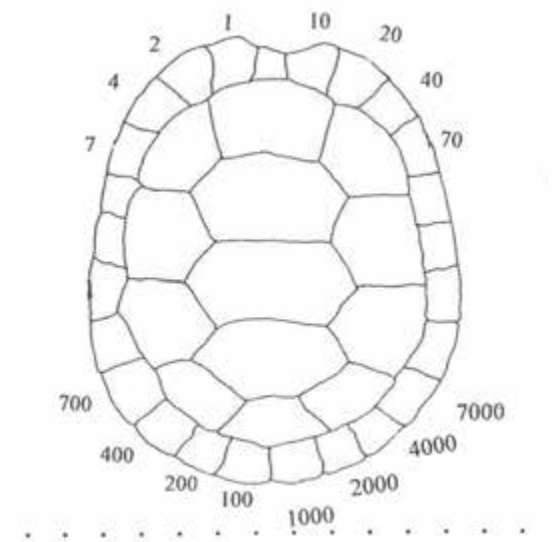


Figure 10-5. A lettering system for use on most hard-shelled turtles. The identification code for this turtle is AJP. Note that the shaded scutes should not be notched or drilled on this species, as the turtle's shell is rounded at those points. Cutting or drilling may cause injury by entering the body cavity. Variation in shell marking protocols occurs among species, and sometimes among individuals within a species. Consult with an experienced herpetologist before initiating a marking program. Reprinted from *Buhlmann, K., T. Tuberville, and W. Gibbons. 2008. Turtles of the Southeast. The University of Georgia Press, Athens, GA*. Used with permission of the authors.



Kurt Buhlmann

Figure 10-6. A numbering system that works well on most hard-shelled turtles. Using this system it is possible to mark 9,999 individual turtles. Using the 1-2-4-7 system, any number from 1-9 can be identified by notching no more than two scutes. The same pattern follows for the 10s, 100s, and 1000s. Some researchers prefer to locate the 1s and 10s posteriorly. Again, work with a herpetologist who has conducted mark-recapture studies with turtles before initiating your own marking scheme. Reprinted from *Buhlmann, K., T. Tuberville, and W. Gibbons. 2008. Turtles of the Southeast. The University of Georgia Press, Athens, GA.* Used with permission of the authors.

In most turtle species, hatchling turtles may also be uniquely marked and identified by using nail clippers to cut tiny v-shaped notches in the marginal scutes. The coding system used should, of course, be the same as those used on adults. Damage to the shell and its development or death can occur from improperly applied markings, so work with an experienced herpetologist.

Unique plastral marks, old injuries, or patterns on individual turtles, much like a human fingerprint, will assist in identification. Thus, where feasible, such as when turtles are brought into a laboratory from the field for processing, a photocopy can be made of a turtle's plastron. Photocopies of plastrons should not be used as the only source of marking identification. Photocopies of hatchlings are helpful as backup identification in some species because hatchling shell notches become difficult to reliably identify as the hatchling grows. In this situation, photocopies are most helpful when a hatchling plastral pattern is evident and persists into the juvenile years.

Whit Gibbons

A scalpel, file, or drill can be used to notch the scutes.

Kurt Buhlmann

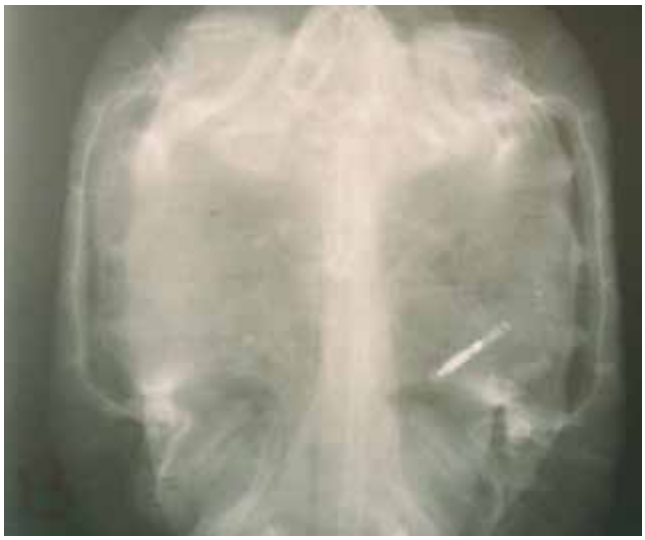
The notches cut with nail clippers are evident on the marginal scutes of these three hatchling chicken turtles (*Deirochelys reticularia*)

Whit Gibbons

With turtles, you can make a photocopy to record their temporary marks and those originating from injuries.

PIT tagging

The use of PIT-tags has been extended to turtles with success (see amphibian marking section). Tags are usually inserted into the body cavity through the inguinal region. However, some researchers have injected PIT tags into legs of turtles. Gluing PIT tags to turtle shells is not recommended as they will be lost when the turtle sheds its scutes, or worn off in species that do not shed scutes (e.g., wood turtles [*Clemmys insculpta*], gopher tortoises [*Gopherus polyphemus*]). PIT tags have several advantages and disadvantages to traditional shell-notching marking schemes. PIT-tagging is very useful for identifying softshell turtles (*Apalone* sp.) that do not have bony shells or marginal scutes to notch or drill. PIT tags may also be employed so that law enforcement personnel may discreetly identify individual turtles, particularly rare or endangered species (e.g., bog turtles [*Glyptemys*], yellow-blotched map turtles [*Graptemys flavimaculata*]) that may be illegally collected for the domestic or overseas pet trade (Buhlmann and Tuberville 1998).



Kurt Buhlmann

X-ray of turtle with a PIT-tag.

Kurt Buhlmann

A Blanding's turtle (*Emydoidea blandingii*) with temporary marks written on plastron.

Disadvantages of PIT tags include cost (see amphibian marking section). The high cost of PIT tag readers makes it unlikely that every researcher will have one with them when a turtle is captured. For long-term studies, it is likely that recaptured turtles without visible shell marks will be recorded as new captures. Also, the shells of deceased turtles are frequently found in the field. The shell marks that identified that animal when it was alive are usually still plainly visible. PIT tags are injected into the soft body parts, thus the ability to identify that animal is quickly lost when the animal dies. In conclusion, when PIT tags are used in turtle monitoring programs, it must be in supplement to shell notching, but not in place of.

Temporary marks

Various colored disks, numbered stickers, or painted numbers may be used as temporary identification on turtles. Florescent-colored plastic disks can be glued to the shells of river cooters, so that individual basking turtles are identifiable at great distances (Buhlmann and Vaughan 1991). Similarly, numbers can be taped onto the shell to temporarily identify nesting female turtles to avoid disturbing them (Congdon et al., 2000). For very short-term inventories, such as a single nesting season, numbers may be painted on the shells of turtles. Under no circumstances should long-term monitoring projects be initiated using these temporary marking systems.

CONCLUSIONS AND RECOMMENDATIONS

An incredible variety of techniques for marking amphibians and reptiles have been developed, used, and improved over the years, and new techniques are currently being developed. Factors such as potential risks, funding, resources, time available, study objectives, habitat, and characteristics of the target species (e.g., size, patterns) should all be carefully considered when deciding on a marking technique to use. When selecting a marking method, we recommend consulting an expert about the technique you are interested in using because they may know of limitations specific to your area or a particular species.

APPENDIX IV. COLLECTING TISSUE FOR BIOCHEMICAL ANALYSIS

Dean A. Croshaw

INTRODUCTION

Collecting tissue samples for later laboratory use is a necessary component of many studies concerning conservation genetics, phylogeography, population and behavioral ecology, systematics, physiology, and molecular biology of amphibians and reptiles. In general, two categories of tissue collection techniques may be used:

1. lethal collection of entire specimens followed by necropsy or

2. non-lethal tissue sampling (biopsy)

If the former option is desired, accepted euthanasia techniques (as recommended by the American Veterinary Medical Association Panel of Euthanasia http://www.avma.org; also see Cooper et al. 1989; Frye

1984; Wright and Whitaker 2001; ASIH 2004; Appendix V in this volume) for amphibians and reptiles should be used before preservation. In both cases, collectors should follow standard necropsy and biopsy procedures (see below).

NECROPSY AND SPECIMEN COLLECTION

Humane techniques for euthanizing reptiles and amphibians include:

1. lethal injection (see below for accepted compounds)
2. submersion of amphibians in a lethal solution
3. inhalant anesthetics
4. gunshot directly through the brain
5. decapitation of unconscious animals followed by pithing
6. rapid freezing of unconscious animals

The most preferred technique is injection of (reptiles) or submersion in (amphibians) a lethal dose of one of the following:

1. sodium pentobarbital
2. hydrous chlorobutanol
3. tricaine methanesulfonate
4. chloretone
5. ethanol
6. other anesthetics

Necessary doses vary widely across species and agent. Pentobarbital should be administered at a dose of 40-90 mg/kg body weight and tricaine methanesulfonate at 200-600 mg/kg body weight (Dessauer et al. 1996). Tissue samples should be taken before fixing the specimen in formalin. Injection of barbiturates can damage internal organs, so this method should be avoided in studies requiring the preservation of internal anatomy. All specimens should be labeled with care, cross-referenced with corresponding tissue samples and the collector's personal field notes, and deposited into a permanent collection using generally accepted procedures (e.g., Dessauer et al. 1996, see appendix section on preparing scientific specimens).

For some research questions, such as studies concerning tissue-specific gene expression, whole-carcass necropsies must be performed (Frye 1984; Jacobs and Heyer 1994). Animals should first be euthanized (see appendix section on preparing scientific specimens) then, a quick dissection can be performed in a cool location. Selected tissue types are removed and cut into small pieces before placement in cryovial tubes for cryopreservation. If freezing options are not possible, tissue should be cut into very small pieces before immersion in a preservative (e.g., ethanol). Samples should be labeled to indicate from which voucher specimen they were taken. Be sure to clean all instruments between individuals and avoid contamination with human tissue.

BIOPSY

If non-lethal tissue sampling is desired, researchers must decide on the type and quantity of tissue to harvest. People commonly take blood samples to be used for later DNA analyses, a technique that, when performed properly, may be less invasive than harvesting of other tissues. Only very small amounts of blood or solid tissue are needed when using highly efficient DNA extraction techniques such as standard phenol-chloroform isolation (Sambrook and Russell 2001). Blood samples are also frequently required for physiological studies, although much greater volume is often required. Obtaining adequate amounts of blood tissue may be difficult, especially with small animals, which includes most amphibians. In these cases, tail clips may be harvested from salamanders, lizards, snakes, or turtles. These samples are usually sufficient for most purposes. Toe clips may be used as well, especially in salamanders, frogs, and lizards, and probably do not result in considerable detriment to survival (e.g., Ott and Scott 1999 for salamanders). This sampling technique should be particularly useful to researchers who need to mark animals for later group or individual identification. Clipping scutes off the tail region of crocodilians is also an accepted tissue collection method (Fig. 11-1).

Cris Hagen

Figure 11-1. Removing a scute from an alligator's tail for a tissue sample.

Procedures for taking blood samples vary widely across taxa and among individual investigators. For genetic analyses, generally very small amounts of blood or tissue are needed. Microgram quantities of amplifiable DNA may be obtained from fewer than 100 microliters of blood or a few milligrams of biopsy (Dessauer et al. 1996). However, for other types of studies, a greater volume may be necessary. Unfortunately, it is very difficult to collect reasonable quantities of blood from many small amphibians. In most of these cases, collecting blood will require sacrificing animals by heart puncture. Blood is collected with a heparinized capillary tube. Larger amphibians can endure blood collection from vessels such as the lingual venous plexus which supplies the tongue in frogs. The midline abdominal vein can also be used in large anurans. In salamanders, the anterior (midline) abdominal vein and the ventral caudal vein are good sources. Heart puncture (Sooter 1955) is a viable and common option for most relatively large reptiles and amphibians. When performed properly, animals survive the technique, but it may cause mortality when performed by inexperienced workers (Reinert and Bushar 1991). When working with large reptiles and most snakes, blood may be sampled from caudal vessels (Gorzula 1978; Fig. 11-2); Reinert and Bushar 1991). Turtle blood is sampled from a femoral or jugular vein, a carotid artery, the retroorbital space, or the paired dorsal cervical sinuses (Dessauer 1970 and references therein; Owens and Ruiz 1980; Fig. 11-3). Blood from lizards of a wide size range may be collected from the orbital sinuses (e.g., Haenel et al. 2003; MacLean et al. 1973). Crocodilian blood is typically taken from the internal jugular or caudal veins or the dorsal sinuses (C. Hagen and T. C. Glenn, pers. comm.). Refer to Table 11-1 for a summary of the most common locations for taking tissue samples from the major amphibian and reptilian groups of the United States.

Cris Hagen

Figure 11-2. How to take a blood sample from the caudal vein of a turtle.

Cris Hagen

Figure 11-3. How to take a blood sample from the dorsal cervical sinus of a turtle.

Jamie Bettaso

Biologists collect tissue samples for Bd testing using a cotton swab, (see Appendix VI).

Nontraditional sources of DNA

Several studies have purported to obtain DNA via much less invasive sampling methods than those outlined in previous sections, feces being the most relevant for reptilian and amphibian subjects (e.g., Frantz et al. 2003). Although it is clear that DNA is available from body secretions and fecal material, it is likely to be of low molecular weight and concentration, rendering it difficult or impossible to analyze. Unless noninvasive sampling techniques are required, traditional sampling, which usually yields large amounts of high quality DNA, is recommended. Of course, when no other method is possible, researchers may well find some success with feces, orifice swabs (e.g., Poschadel and Moller 2004), and shell or scale remnants.

Tissue Type	Frogs	Salamanders	Turtles	Crocodilians
Blood	Lingual venous plexus	Tail Vein	Cervical sinuses	Dorsal sinuses
Solid	Toe clips	Toe/tail clips	Tail clips	Scutes
Tissue Type	**Lizards**	**Snakes**		
Blood	Orbital sinuses	Caudal vessel		
Solid	Toe/Tail clips	Tail clips		

Table 11-1. Common techniques used for tissue collection.

TISSUE PRESERVATION

Solid tissue

For any tissue collection, whether whole-animal or a tissue sample, a simple preservation option is to place the collection on ice in a cooler or other container for quick transport to an ultra-cold freezer (-70° to -80° C) where it can be stored for long periods. However, this is not recommended unless the transport time is short (minutes to hours). DNA from some organisms may degrade extremely quickly (< 1 hour, personal observation). In most cases, it will be desirable to use an immediate preservation technique in the field.

Several tissue preservatives may be used, but cryopreservation is probably the most generally effective and commonly used option. As soon as possible, samples can be placed in liquid nitrogen or on dry ice (solid CO_2) for long-term storage or transport to an ultra-cold freezer (see Dessauer et al. 1996 and Jacobs and Heyer 1994 for field procedures). Placing tissue immediately into ethanol is another option that is usually practical in field situations. It is best to use undiluted ethanol (i.e., > 95%) and cut the tissue into very small pieces to insure that the preservative quickly reaches all parts of the sample. Tissue should be placed in at least twice its volume of ethanol. After a few hours of preservation, the ethanol should be changed, as it has been diluted by fluid from the tissue. This technique is standard for tissue to be used for future DNA extraction (Sibley and Ahlquist 1981). Several other preservatives may be used in a similar fashion, including isopropanol, propanol, dimethyl sulfoxide (DMSO), and salts of ethylenediaminetetraacetic acid (EDTA), acetone, diethyl ether, and ethyl acetate. Seutin et al. (1991) showed that a DMSO-salt solution (20% DMSO, 0.25 M sodium-EDTA, and NaCl to saturation, pH 7.5) preserved DNA in tissue samples as well as cryopreservation and better than 70% ethanol. Fukatsu (1999) showed that acetone is superior to ethanol at preserving DNA in samples contaminated with water.

Solid desiccants (e.g., silica gel) are sometimes used to preserve animal tissue. Indicator desiccants, which change color when no longer effective, are safest to use because tissues are susceptible to rehydration. Below is a list of several solid tissue preservation techniques in order of decreasing frequency of use:

1. cryopreservation
2. ethanol
3. DMSO-salt solutions
4. other alcohols
5. DMSO
6. EDTA
7. Solid desiccants

Tissue may also be dried in an oven or freeze dryer before long-term storage. Freeze drying is recommended because it avoids subjecting samples to high temperatures that may accelerate the breakdown of macromolecules. After drying, it is essential to store samples in a cool dry place. Because of the omnipresent risk of rehydration and subsequent degradation, the above techniques are more prudent.

Blood

Blood samples should be placed immediately in an anticoagulant such as heparin or EDTA or dried onto

filter paper before long-term storage. Special cards, of various trade names (e.g., FTA, Isocode), are commercially available for dried blood spot sampling. The cards consist of high quality filter paper, covered with a special matrix. Chemicals in the matrix cause blood cells to lyse and stabilize nucleic acids by inactivating enzymes and other agents that could degrade them. DNA and RNA are stable for very long time periods on cards stored at room temperature. A drop of blood is placed directly onto the card and allowed to air dry, before storing in plastic bags with silica gel desiccant if available. The use of heat-assisted drying of blood samples is not recommended. When ready to use, a portion of the sample is punched out of the card and eluted into a buffer using a special protocol. Techniques have been developed to perform polymerase chain reaction directly from the card, without the elution step.

For storing blood as a liquid, use about one mg of heparin or one millimole of EDTA for every 100 mL of blood (Dessauer et al. 1996). It is often desirable to use a collecting device (capillary tube or syringe) that already contains the anticoagulant. Samples may be cryopreserved or stored in a lysis buffer. Numerous unique chemical compositions have been used successfully to preserve and store blood tissue. Lysis buffers generally contain EDTA, tris(hydroxymethyl) aminomethane hydrochloride (Tris-HCl), and sodium chloride (NaCl). Several other additives are sometimes used (e.g., urea, sodium dodecyl sulfate (SDS), DMSO, diaminocyclohexanetetraacetate). A detergent in the buffer lyses the cells, causing the DNA to go into solution. The salts inactivate residual nucleases, preventing DNA degradation. A study by Seutin et al. (1991) found that "Queen's" lysis buffer (0.01 M Tris-HCl, 0.01 M NaCl, 0.01 M sodium-EDTA, and 1.0% n-lauroylsarcosine, pH 8.0) performed extremely well in preserving DNA in blood samples stored at room temperature.

Listed below are the most common lysis buffer additives:

1. EDTA
2. Tris-HCl
3. NaCl
4. SDS
5. DMSO

OBTAINING PRESERVATIVES

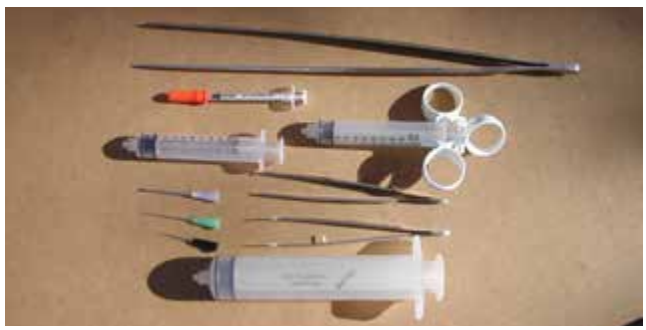
All the chemical preservatives that will be needed for tissue collection may be easily obtained from most chemical suppliers, including the following industry leaders: Sigma-Aldrich (www.sigmaaldrich.com), Fisher Scientific (www.fishersci.com), and Amresco (www.amresco-inc.com). Dry ice may be purchased from frozen food distributors and cryogenic specialists (Jacobs and Heyer 1994). The Dry Ice Ideas website (www.dryiceideas.com) provides a directory of dry ice suppliers. Liquid nitrogen may be obtained from a variety of liquid gas suppliers.

APPENDIX V. PREPARING SCIENTIFIC SPECIMENS

Brian D. Todd and Peri A. Mason

INTRODUCTION

Preserved specimens, accompanied by properly composed field notes, are excellent resources for scientists and can be useful in investigations spanning every branch of biology. Of particular relevance to this book are the historical data that preserved specimens provide, which enable researchers to detect and assess changes in biodiversity in an area over time. Likewise, specimens provide evidence that a species exists or existed in a particular area. This information can be used to create range maps, document changes over time, and can be consulted for many different purposes (also see Appendix VII on vouchering with photos). Here, we review preferred methods of preparing voucher specimens. For more information and alternative treatments, refer to Pisani (1973) and McDiarmid (1994b).



Joe Mitchell

Tools commonly used during the preservation process includes needles, syringes, and forceps.

RECORDING FIELD DATA

Field notes are essential if a specimen is to be useful. Date and locality should be recorded at a minimum. Other pertinent information to record includes time of day, names of all collectors present, weather data, including temperature, cloud cover, moisture, and recent weather changes, a list of all species collected at the given locality including observed species, microhabitat, and field number for the specimen in question. The field number should match the number assigned to any associated materials such as photos, tissue samples, regurgitated prey, color descriptors, and the preserved animal. These are important to note because preservatives will fade the pigment in the animal's integument and coloration will not always be observable after preservation. Photos of the specimen may also prove invaluable because photographs record the coloration of the animal at the time of capture (see Appendix VII on vouchering with photos). Waterproof ink and paper should be used for field notes. As long as the identities of the animals can remain certain, tags can be applied upon returning from the field. Otherwise, tags can be added in the field. Commercial tags are available in numerical sequence. They are relatively expensive, but save time and prevent errors and they are often resistant to fading in preservatives.

Joe Mitchell

Heading out to collect specimens.

EUTHANIZING ANIMALS

It is possible that some voucher specimens in an inventory of herpetofaunal biodiversity will be salvaged from already dead animals, possibly from trap or road mortality. However, live specimens will occasionally need to be sacrificed and preserved depending on the objectives of the study. Zug et al (2001) suggests the preservation of 20-30 animals for scientific study or a minimum of 4 for a voucher specimen. Obviously, it is recommended that you collect the minimum number of animals needed to meet the objectives of the study. When necessary, euthanasia must be humane and should preserve the condition of the animal (see the appendix section on collecting tissue for biochemical analysis for more information on euthanasia). Make sure that you follow any applicable institutional guidelines and obtain proper approval and permits if required.

Joe Mitchell

Road-killed herpetofauna with minimal damage can be preserved as specimens. This garter snake was killed on the road and later preserved.

Amphibians

Because amphibians have highly permeable skin, application of liquid solutions to the skin or immersion in anesthetic solutions are the most frequently used forms of euthanasia. Tricaine methanesulfonate (TMS), also known as MS-222®, is a commonly used agent for euthanizing amphibians (>500 mg/L). It is acidic when dissolved in water and should be buffered to minimize discomfort to the animals. Benzocaine hydrochloride is similar to TMS and can also be used in a bath (>250 mg/L) for euthanizing amphibians. Ten percent (10%) ethanol or Chloretone solutions can be used in the same fashion (McDiarmid 1994b). Chloretone solution is made by dissolving 1 tsp. hydrous chlorobutanol crystals in 1 L water. Benzocaine-containing gels for toothache remedies are also readily available over-the-counter and can be used on short notice for euthanizing small amphibians (Altig 1980). Spread the gel on the head between the eyes and on the pelvic patch in the case of anurans. Lastly, sodium

pentobarbital (60-100 mg/kg of body weight) can be administered intravenously, intra-abdominally, or intrapleuroperitoneally. Other barbiturates may cause pain on injection and should be avoided. Species will differ in the length of time it takes for any method to completely euthanize the animal. Be sure that the animal has stopped respiring and is dead before proceeding to fix and preserve the animal.

Reptiles

Sodium pentobarbital (60-100 mg/kg of body weight) can be injected intravenously, intra-abdominally, or intra-pleuroperitoneally, or may be injected into the heart. But Nembutal is difficult to obtain and requires a permit that may not be easily available. Injection of alcohol to the heart also works (25-30% solution; 1 mL for each cm of SVL or 1cc per cm of plastron length; all types [ethyl, methyl, isopropyl] will work here). Confinement in a sealed container with chloroform is recommended for turtles only, as soft bodied reptiles will contort. The turtle should be placed in a closed container with chloroform dampened rags or cotton. Chemicals with similar properties, such as Trichloroethylene and ether are suggested for other reptiles and a similar method of confinement should be used. Large specimens (≥5 lbs) should always be injected instead of confined. Some reptiles, especially turtles and crocodilians, can hold their breath a long time and survive long periods without oxygen. This may increase the amount of time it takes for an inhalant to cause death. Such problems are less likely to exist with lizards and snakes.

FIXING

Chemical fixation causes the denaturation and hardening of proteins in the tissue of specimens. Therefore, specimens must be fixed in a position that preserves their morphological state and that is conducive to observation of key characters such as scale count or number of toes. For example, *Rhineura floridana* would be fixed with something propping its mouth open, as oral characters are important in distinguishing it from other amphisbaenids. A chicken turtle (*Deirochelys reticularia*) would be fixed with its neck entirely distended because its long neck is unique among southeastern emydids.

Chemical supplies

A solution of 10% commercial formalin should be used for fixing reptiles and amphibians. Formalin is a mixture of 37-40% formaldehyde, water, and occasionally 10% methanol. Formalin is carcinogenic, flammable, and dangerous if fumes are inhaled so appropriate precautions should be taken, including using a fume hood or working in a well-ventilated area. It is also advisable to use safety goggles and gloves when working with formalin. If formalin is not available, 70% ethyl alcohol can be used. Other alcohols are not recommended (McDiarmid 1994b).

Lucas Wilkinson

The proper safety equipment must be used when preserving specimens.

Procedures for fixing amphibians

Most amphibians can be fixed in a preserving tray after they have been euthanized. Long shallow plastic trays with tight fitting lids are appropriate for fixing specimens. The bottom of the tray is lined with undyed paper towels soaked with formalin (colored paper towels will stain the specimen). The limp specimen is then placed on the paper towel and arranged in the desired position. Optimum positions are ones that facilitate later examination and measurement of key characters as well as make long-term storage easy. Salamanders can typically be arranged in a life-like state with their ventral side facing down (Fig. 12-1). Their tails should extend straight behind them. Caecilians and fully aquatic salamanders such as amphiumas or sirens can be preserved in a loose, flat loop or with their body straight. Some researchers recommend the preservation of frogs and toads with their hind limbs extended (Pisani 1973), whereas others find that tag loss and long-term storage can be difficult with animals in this position and instead recommend storing frogs with both fore and hind limbs tucked into a natural position (McDiarmid 1994b, Fig. 12-2). After arranging the animal in the appropriate position, more formalin-soaked paper towels can be placed over the specimens. Amphibians will generally harden within hours or days and field tags can be affixed to the animals. In contrast to reptiles, most amphibians will not need to be injected with formalin. Fixation is usually

complete within 4 days, whereupon specimens are ready to be rinsed (see below). Then the animals can be transferred to their long-term storage solutions. Eggs and larvae can usually be fixed by placing the specimens directly into the formalin solution. Because of the amount of water contained in many egg masses and in many larvae, you may require a stronger formalin solution for fixation; McDiarmid (1994b) has suggested using pure formalin.

Joe Mitchell

Figure 12-1. Salamanders should be individually labeled, positioned as shown here, and covered with an additional layer of paper towels during the fixing process.

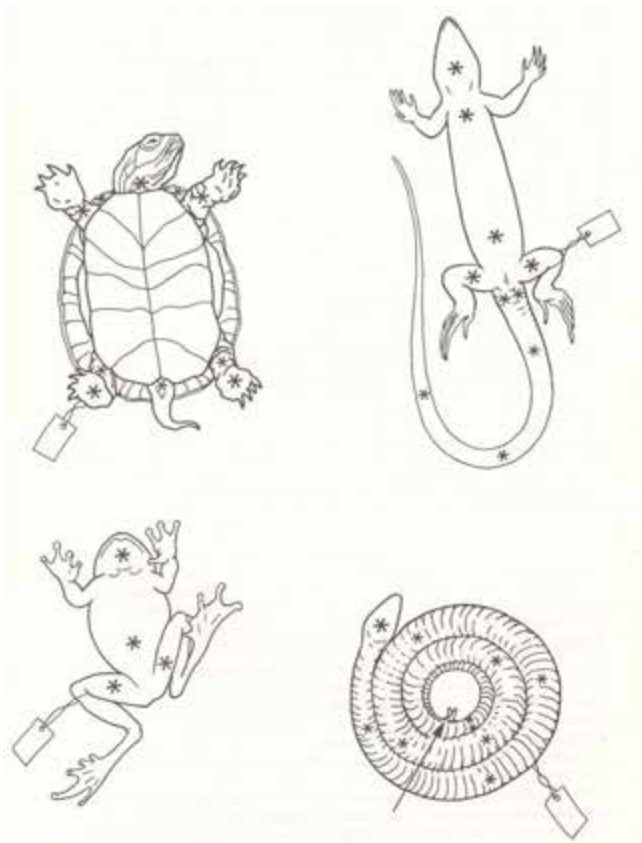


Figure. 12-2. Proper position for preserving turtles, lizards, anurans, and snakes.

Procedures for fixing reptiles

Reptiles should be injected with as much formalin as possible before the body becomes distorted because injection prevents internal decay. Turtles should be injected with 1/3 to 1/2 of their body cavity volume worth of formalin. All appendages including the tail should be injected generously. Necks should be distended entirely, which can be done by hanging the specimen from a hook by the jaw for injecting. Mouths should be held open (a cork works well). Snakes should be injected about every 20 cm for the body cavity and as far down on the tail as the needle can puncture (Zug et al 2001). The underside of the lower portion of the snake's tail should be slit using a razor blade so that it will take in formalin when it is submerged. Snakes should be coiled neatly. Specimens should be placed in the proper position on a layer of formalin soaked paper towels which have been placed in a container or tray with a cover. Tags can be added now or after fixation, but prior to long-term storage. Care must be taken that all specimen museum numbers are recorded in a museum record book with all associated information (as delineated above). When the bottom of the container is covered, another layer of formalin soaked, undyed paper towels can be added. Specimens can be layered atop, stacked in this manner as long as the layering does not upset the position of any specimen. The container will be covered and left for 24 hours. Larger turtles and snakes should be flipped over (on back first, then belly) after 24 hours and then left for another 24 hours in order to ensure that both the muscle of the dorsum and the viscera of the venter have been adequately permeated by the fixative. Specimens can then be moved to a jar and submerged in formalin. You can place as many reptiles per jar as will fit because they are already fixed in position (Fig. 12-3). The reptiles then stay submerged in formalin for 4-5 days. Turtles may take longer. The formalin may be reused until it becomes "dirty" with foreign material. It must be disposed of according to regulations.

Lucas Wilkinson

Figure 12-3. Snakes can be coiled up in a preserving jar until the jar is filled.

STORAGE AND PRESERVATION

Following fixation, specimens should be soaked in water for 1-2 days and the water should be changed a few times daily. After removing the specimens from their rinse, they should be stored in 50-70% ethanol with specimens of similar sizes to prevent larger specimens from damaging smaller ones (Zug et al. 2001). Other forms of alcohol are not recommended. Stored specimens should be kept cool and dry. Although specimens store well in formalin, it is often undesirable as a long-term preservative because the noxiousness of formalin may preclude working with the preserved specimens. Monitor your collection at least every six months for evaporation. If a lot of evaporation has occurred, replace all of the alcohol rather than topping off the remaining solution as the concentration may have changed and specimens may begin to dehydrate.

SHIPPING AND TRANSPORTATION

When transporting or shipping specimens, they should be wrapped in cheesecloth or durable undyed paper towels before being soaked and carefully bagged. This will prevent some damage during transport. Sealed bags of preserved specimens should be placed inside of sealed plastic containers instead of glass to prevent breakage and leaks from happening. Vials should be carefully shipped in special containers or boxes that minimize the jostling of the contents and prevent breakage. It is not advisable to ship specimens in formalin due to the noxiousness of formalin and the risk of leakage. Instead, ship specimens double bagged in 50-70% ethyl alcohol. **Please check with your specific shipping or airline carrier to ensure that appropriate carrier-specific requirements and regulations are met before transporting any preserved specimens.**

APPENDIX VI. DECONTAMINATION OF FIELD GEAR TO MINIMIZE POTENTIAL PATHOGEN TRANSMISSION ACROSS STUDY SITES

Jamie B. Bettaso

INTRODUCTION

Pathogens and parasites can pose a threat to amphibians and reptiles. Pathogens can be bacterial, viral, or fungal, whereas parasites are usually protozoa or helminths (i.e., parasitic worms). Many of these organisms may be a part of the natural world yet become a problem for amphibians and reptiles when they are introduced to a naïve population or if the population is under stress from another source (e.g., contaminants reducing the immune system of a species).

A bacterial infection of *Aeromonas hydrophila* was associated with a die-off of Mountain Yellow-legged Frogs. *Rana muscosa* (Bradford 1991). This bacterial pathogen is known as “red-legged” disease in amphibians and is the same pathogen that causes “red-mouth” disease in fish (Hazen et al. 1978). Viral infections associated with iridoviruses have been linked to several die-off events in North America (Green et al. 2002; Docherty et al. 2003). An iridovirus called “Redwood Creek Virus” is the first iridovirus described to have infected both an amphibian and a fish (Moa et al. 1999).

Chytridiomycosis is a fungal pathogen caused by the etiological agent *Batrachochytrium dendrobatidis* (often referred to as “chytrid” or “Bd”) that has been associated with both die-off events and declines of amphibians in North America, Central America, and Australia (Berger et al. 1998; Longcore et al. 2007; Morgan et al. 2007). The following protocol for decontamination of field gear was developed with a focus on limiting the spread of chytrid.

DECONTAMINATION OF FIELD GEAR FOR HERPETOFAUNA SURVEYS

Biologists should consider the decontamination of field gear for at least these three scenarios:

1) Sites to have known infections or die-offs

2) Sites across a watershed level, which is between major lotic systems

3) Sites across watersheds at a basin level.

If wildlife biologists, researchers, or land managers have sites where they have witnessed die-offs of amphibians or suspect declines of amphibians, they should decontaminate their field gear before and after a site visit. When biologists are working in aquatic systems such as streams and rivers, effort should be made, especially during first time visits, to work from sites in the upper watershed to sites in the lower watersheds and to decontaminate field gear between

sites that are widely separated. Decontamination of field gear between watersheds should be conducted. For biologists working in wetland habitats, such as areas in the Prairie Pothole Region or in mountainous areas with lake and pond habitats, biologists should decontaminate field gear between basins or catchments. The following websites provide additional information on decontamination: www.separc.org (see "Disease/Pathogens/Parasites page), www.ccadc.us (see "Field Decontamination Guide").

Chemical disinfectants used for decontamination of field gear

A good first step for decontamination of field gear is to remove all visible contaminants such as mud or aquatic vegetation from stream boots, waders, water shoes, dip-nets, seine nets, or any gear used in the aquatic environment. Next, place equipment into a decontamination solution for the appropriate concentration and time [see Table 13-1]. Equipment can be washed with tap water to remove residual decontamination solution, but should not be washed in the field at the site most recently visited.

All disinfectants in Table 13-1 are adapted from the published work of Johnson et al. (2003) and Webb et al. (2007) and are for concentrations with 100% killing efficiencies of *Batrachochytrium dendrobatidis*. These disinfectants may extend to other pathogens but the studies cited were for control of Bd, therefore further research would be required to determine the effectiveness of these disinfectants to Ranaviruses and *Aeromonas* species.

TABLE 13-1.

Chemical disinfectants, concentrations, and exposure times for complete killing effectiveness against *Batrachochytrium dendrobatidis* (Adapted from Johnson et al. 2003 and Webb et al. 2007).

Chemical/Product	Disenfectant concentration	Exposure Time of Gear in Disinfectant	100% Effectiveness
Sodium chloride	10%	10 min, 2min	Yes
Household bleach[1]	4 to 1%	10 min, 5 min, 2 and 1 min, 30 sec	Yes
	0.2% to 0.1%	10 min	Yes
Potassium permanganate	2%	10 min, 5 min	Yes
	1%	10 min	Yes
Formaldehyde solution	1%	10 min, 5 min	Yes
	0.1%	10 min	Yes
Path-X™ agricultural disenfectant	1 x 10^{-2} to 1 x 10^{-3}%	5 min, 2 min, 1 min and 30 sec	Yes
	1 x 10^{-4} %	5 min, 2 min	Yes
Quaternary ammonium compound 128[2]	Full strength to 1 x 10^{-3} %	5 min, 2 min, 1 min and 30 sec	Yes
Virkon	1 mg ml^{-1}	5 min, 20 sec	Yes
Ethanol	70%	5 min, 20 sec	Yes
Benzalkonium chloride	1 mg ml^{-1}	5 min, 20 sec	Yes
F103	4 ml l^{-1}	10 min, 5min and 1 min	Yes
	1 ml l^{-1}	10 min, 5min and 1 min	Yes
	0.33 ml l-1	10 min, 5min and 1 min	Yes
	0.28 ml l-1	10 min	Yes
Betadine	Full strength (10% povidone iodine)	10 min, 5min and 1 min	Yes
	500 ml l-1 (5% povidone iodine)	10 min, 5min and 1 min	Yes
	100 ml l-1 (1% povidone iodine)	10 min, 5min and 1 min	Yes
TriGene4	50 ml l-1	10 min, 5min and 1 min	Yes
	2 ml l-1	10 min, 5min and 1 min	Yes
	1 ml l-1	10 min, 5min and 1 min	Yes
	0.2 ml l-1	10 min, 5min and 1 min	Yes
	0.1 ml l-1	10 min, 5min and 1 min	Yes

1 Active ingredient = sodium hypochlorite; 2 Active ingredient = DDAC (didecyl dimethyl ammonium chloride); 3 Active ingredients = benzalkonium chloride and polyhexamethylene biguanide hydrochloride ; 4 Active ingredients = dodecylamine sulphamate, octyldecyl dimethyl ammonium chloride, poly (hexamethylene) biguanide hydrochloride, 4 nonyl phenyl-w-hydroxyl-poly (oxythylene) and ethanol.

APPENDIX VII. PHOTO-VOUCHERING

John B. Jensen

INTRODUCTION

Photo-vouchering, the use of photographs to document the occurrence of encountered wildlife, has gained increased acceptance in the herpetology community, especially when careful consideration is made to take photographs that accent identifying features of amphibian and reptile species. Although sacrificing a limited number of specimens and placing them in natural history museums is still considered by most herpetologists to be the best way to properly document species' occurrences (see Appendix V on Preparing Scientific Specimens), growing conservation and ethical concerns have led, or in some cases required, many to use non-lethal means of validating amphibian and reptile observations. Herein we provide information to assist those in deciding whether to sacrifice or photograph voucher specimens, and if photo-vouchering, what methodology to follow to ensure adequate documentation.

John Jensen

By itself, this photograph of a juvenile Broadhead Skink is not suitable for vouchering purposes. The scale characteristics needed to distinguish it from Five-lined and Southeastern Five-lined skinks, which co-occur where this individual was found, are not accented sufficiently. Close-up photographs of the side of the head and the underside of the tail would be necessary to reveal its indentity.

WHY IS VOUCHERING NEEDED?

Vouchering the occurrence of particular species in particular places provides long-term evidence that those species exist, or existed, there. This information is used to support where lines are drawn on range maps, where rare species surveys may be required in planning and mitigating for proposed construction projects, to evaluate changes in species' distributions, whether expansion or shrinkage, and many other purposes. Long-term and verifiable evidence is necessary so that future scientists, land managers, developers, and others have confidence that previously reported occurrences made by others were accurate. A literature record not supported by any verifiable evidence of the pine barrens treefrog (*Hyla andersonii*) in Georgia, for example, is the only information that the species is, or was, native to the state. Many herpetologists have surveyed the reported site and have not rediscovered the species, nor have they found appropriate habitat. As a result, most herpetologists question the validity of the occurrence. A photograph of this very distinctive species would have satisfied many of the doubters.

PHOTOGRAPH OR SACRIFICE, OR BOTH?

While sacrificing an individual or two from a particular locality followed by proper fixation, preservation, and accessioning in a reputable natural history museum remains the surest way to provide long-term verifiable evidence of a species' occurrence (see Appendix V), many species' occurrences can be adequately documented with sufficient photographs. Photography may be the best, or in some cases the only option for documenting rare, threatened, and endangered species. Quite understandably, many landowners or land managers may not want any of the amphibians or reptiles on their property sacrificed for science, but taking photographs of them is almost always possible. Some species, such as freshwater turtles, may be difficult to capture, but photographing them with a telephoto lens while they are basking on a distant log, for example, is much easier.

Unfortunately, quite a few species of amphibians or reptiles have very subtle characters that are rarely sufficiently captured in photographs to distinguish them from closely related or similar-looking species. For these species, at least in areas where they may co-exist with their look-alikes, sacrificing voucher individuals may be the only adequate way to document their occurrence. Please refer to the table of photograph priorities (Table 14-1) to determine which species are unlikely to be verifiable with photographs alone.

Many specimen vouchers would actually benefit from additionally having photographs accompany them. Preserved specimens eventually lose color and pattern, which, for some species, aid in identification. Having both the specimen, to examine subtle characters, and an accompanying photograph, to show color and pattern in life, may be the ideal way to document the occurrence of certain species.

HOW TO PROPERLY PHOTO-VOUCHER A SPECIMEN

Photographs of amphibians and reptiles may be taken using several media – digital, slide, or print. Digital photographs have the advantage of quick review to make sure the photographs are adequate, they are cost-efficient, can be easily and quickly shared via e-mail or websites, and if properly done can be archived in original condition for the long-term. However, digital photograph files can be easily corrupted or deleted if the proper precautions are not taken. Slides and prints have some advantages, but both lose quality with age. Consul the natural history museum or other repository that the photo-vouchers are ultimately intended for to determine the preferred or required medium, and the preferred or required size and resolution for digital photographs.

John Jensen

Where the ranges of Cope's gray and bird-voiced treefrogs overlap, photo-vouchers of either must include shots of the back of the thighs. Cope's gray treefrogs have a wash of bright yellow or orange coloration on the back of the thighs, while this area is greenish on bird-voiced treefrogs. Thus, the photograph of the bird-voiced treefrog (top left photo) is not a sufficient voucher.

Proper photo-documentation requires that the photographer captures the identifying features of particular species. Refer to Table 14-1 to determine which features of each species need to be accented by the photographs to help ensure verifiable identification. Again, in certain areas occupied by two or more similar-looking species, photographs may not be sufficient. In much of the southeastern United States, for example, the "blue-tailed skinks," including the five-lined (*Eumeces fasciatus*), southeastern five-lined (*E. inexpectatus*), and broad-headed skinks (*E. laticeps*), co-occur, and depending on the quality of the photographs and the abilities of the photographer, the often subtle differences in certain scale characteristics may not be captured well-enough to determine the exact species. However, if a blue-tailed skink is photographed in southern Florida where only the southeastern five-lined skink occurs, a single coarse-scale, full-body photograph may be all that is necessary to document the species' occurrence. Thus, it is important to know what species possibly occur in a given geographic area, and using Table 14-1, what characters should be accented by photographs if other similar-looking species co-occur there.

John Jensen

Many amphibians change color and/or pattern soon after capture. Thus, whenever possible, photograph individuals as soon as possible after capture to ensure that natural coloration and patterns are depicted. Both of these photographs (above) are of upland chorus frogs, but the light-colored and patternless individual (top right) was held in a white container prior to being photographed. In areas where this species co-occurs with the mountain chorus frog, this photograph may not be a suitable voucher for the species.

Some helpful tips to consider:

- Ideally, include some sort of scale in the photographs to indicate relative size of the animal. Obviously, a ruler is a suitable scale, but

anything that is of known approximate size can be used. This can include a coin, a lens-cap, a pen or pencil, etc. Photographs used for vouchering purposes do not need to have National Geographic-quality backgrounds – photographs of hand-held amphibians and reptiles provide sufficient scale and allow the captor to position the animals in ways that show distinguishing features.

- When possible, photograph specimens soon after capture. Many herpetofauna, especially certain amphibians, rapidly change color and pattern once captured and placed in containers. Amphibians placed in light-colored containers often become light-colored themselves, and may lose all traces of discernible pattern. Conversely, placing amphibians in dark containers may cause them to become so dark that distinctive patterns are obscured.

- Photographing quick and slippery amphibians can quite often be an exercise in extreme patience. To make this easier, especially when the aesthetic value of the photograph is not an issue, place the animal in a deflated Ziploc bag with a small amount of water and photograph it through the clear plastic. This is especially helpful for species that require photographs of the belly that are otherwise difficult to obtain since live amphibians rarely remain motionless when flipped on their back. Photograph "bagged" specimens in the shade whenever possible. Be sure not to leave any animal in a plastic bag exposed to sunlight for more than a few seconds – the greenhouse effect doesn't take long to raise the temperature in the bag to lethal levels.

- Another technique for photo-vouchering otherwise hyperactive herpetofauna is to chill (do not freeze!) them in a refrigerator or cooler for a short time prior to photographing. Make sure the animal has returned to normal activity temperatures before releasing it so it does not fall prey to opportunistic predators.

WHAT TO DO WITH PHOTO-VOUCHERS

We recommend that photo-vouchers be placed in natural history museums that are dedicated to long-term storage and maintenance of amphibian and reptile collections. If suitable repositories are not convenient, contact a local herpetologist to assist you with photo-voucher placement or to recommend alternatives. All photographs should be accompanied with exact locality of capture/observation data, date of capture/observation, the name of the captor/observer, and other notes that may be helpful, such as the habitat type where the specimen was found and the number of individuals observed (see "recording field data" in Appendix V).

John Jensen

Some species, like this spotted salamander, are not likely to be confused for any other species. For these, a single dorso-lateral photograph of the whole indvidual is all that is needed to provide a sufficient voucher.

TABLE 14-1: PHOTO-VOUCHERING TABLE

TABLE 14-1. PHOTO VOUCHERING TABLE
This table provides a checklist of the major anatomical features and photographic angles that should be captured to ensure that images are sufficient for species identification or verification (adult forms only). It is important that you use the information provided specific to the particular region of interest since each region may have a different suite of species that can be confused with one another and therefore may require more or different photographs/images.

It is also important to consult with your state's laws and regulations concerning capture and handling of amphibian and reptile species, since even short-term handling may be regarded as a form of illegal take with some species in certain states without proper permits. For more information on species vouchering, please refer to the summary in Appendix VII. Please see the Taxonomic Synonyms Table (Chapter 5, after *Species x Techniques Table*) for information about official changes in scientific and common names of given species (Crother et al. 2008).

FROGS AND TOADS

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
SOUTHEAST REGION														
Acris crepitans	Northern Cricket Frog	x				x	x		x					
Acris gryllus	Southern Cricket Frog	x				x	x		x					
Bufo americanus	American Toad	x			x		x							
Bufo fowleri	Fowler's Toad	x			x		x							17
Bufo quercicus	Oak Toad	x												
Bufo terrestris	Southern Toad	x			x		x							
Bufo valliceps	Gulf Coast Toad	x			x		x							
Bufo woodhousii	Woodhouse's Toad	x			x		x							17
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	x			x									
Gastrophryne olivacea	Western Narrow-mouthed Toad	x			x									
Hyla andersonii	Pine Barrens Treefrog	x			x									
Hyla avivoca	Bird-voiced Treefrog	x			x	x								
Hyla chrysoscelis	Cope's Gray Treefrog	x			x	x								1
Hyla cinerea	Green Treefrog	x			x									
Hyla femoralis	Pine Woods Treefrog	x			x	x								
Hyla gratiosa	Barking Treefrog	x			x									
Hyla squirella	Squirrel Treefrog	x			x									
Hyla versicolor	Gray Treefrog	x			x	x								1
Pseudacris brachyphona	Mountain Chorus Frog	x			x									
Pseudacris brimleyi	Brimley's Chorus Frog	x			x	x								
Pseudacris crucifer	Spring Peeper	x			x									
Pseudacris feriarum	Southeastern Chorus Frog	x			x		x							
Pseudacris illinoensis	Illinois Chorus Frog	x			x									
Pseudacris nigrita	Southern Chorus Frog	x			x		x							
Pseudacris ocularis	Little Grass Frog	x			x									
Pseudacris ornata	Ornate Chorus Frog	x			x									
Pseudacris streckeri	Strecker's Chorus Frog	x			x									
Pseudacris triseriata	Western Chorus Frog	x			x									
Rana areolata	Crawfish Frog	x			x									
Rana blairi	Plains Leopard Frog	x			x									
Rana capito	Gopher Frog	x			x									
Rana catesbeiana	American Bullfrog	x			x		x		x					
Rana clamitans	Green Frog	x			x				x					
Rana grylio	Pig Frog	x			x		x		x					
Rana heckscheri	River Frog	x			x			x						
Rana okaloosae	Florida Bog Frog	x			x				x					
Rana palustris	Pickerel Frog	x		x	x									
Rana pipiens	Northern Leopard Frog	x			x			x						
Rana sevosa	Dusky Gopher Frog	x			x									
Rana sphenocephala	Southern Leopard Frog	x			x			x						
Rana sylvatica	Wood Frog	x			x									
Rana virgatipes	Carpenter Frog	x			x				x					
Scaphiopus holbrookii	Eastern Spadefoot	x			x									

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Scaphiopus hurterii	Hurter's Spadefoot	x			x		x							
Spea bombifrons	Plains Spadefoot	x			x		x							
NORTHEAST REGION														
Acris crepitans	Northern Cricket Frog	x				x	x		x					
Acris gryllus	Southern Cricket Frog	x				x	x		x					
Bufo americanus	American Toad	x			x		x							
Bufo fowleri	Fowler's Toad	x			x		x							
Bufo quercicus	Oak Toad	x					x							
Bufo terrestris	Southern Toad	x			x		x							
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	x			x		x	x						
Hyla andersonii	Pine Barrens Treefrog	x			x	x								
Hyla chrysoscelis	Cope's Gray Treefrog	x			x	x		x						1
Hyla cinerea	Green Treefrog	x	x		x									
Hyla femoralis	Pine Woods Treefrog	x			x	x								
Hyla gratiosa	Barking Treefrog	x			x			x						
Hyla squirella	Squirrel Treefrog	x			x			x						
Hyla versicolor	Gray Treefrog	x			x	x		x						1
Pseudacris brachyphona	Mountain Chorus Frog	x			x		x			x				
Pseudacris brimleyi	Brimley's Chorus Frog	x		x	x	x								
Pseudacris crucifer	Spring Peeper	x			x				x	x				
Pseudacris feriarum	Upland Chorus Frog	x			x		x							
Pseudacris kalmi	New Jersey Chorus Frog	x		x	x			x			x			
Pseudacris nigrita	Southern Chorus Frog	x			x	x	x	x						
Pseudacris ocularis	Little Grass Frog	x		x	x			x						
Pseudacris triseriata	Western Chorus Frog	x			x	x								
Rana catesbeiana	American Bullfrog	x			x		x		x					
Rana clamitans	Green Frog	x			x		x		x					
Rana palustris	Pickerel Frog	x		x	x	x		x						
Rana pipiens	Northern Leopard Frog	x			x			x						
Rana septentrionalis	Mink Frog	x			x	x			x					
Rana sphenocephala	Southern Leopard Frog	x			x		x	x						
Rana sylvatica	Wood Frog	x			x			x						
Rana virgatipes	Carpenter Frog	x			x				x					
Scaphiopus holbrookii	Eastern Spadefoot	x			x			x	x					
SOUTHWEST REGION														
Acris crepitans	Northern Cricket Frog	x			x		x		x					
Ascaphus truei	Coastal Tailed Frog	x			x									
Bufo americanus	American Toad	x			x		x							
Bufo boreas	Western Toad	x		x	x		x	x						
Bufo californicus	Arroyo Toad	x			x		x	x						
Bufo canorus	Yosemite Toad	x			x									
Bufo cognatus	Great Plains Toad	x			x		x	x						
Bufo debilis	Green Toad	x			x		x	x						
Bufo exsul	Black Toad	x			x									
Bufo hemiophrys	Canadian Toad	x			x									
Bufo houstonensis	Houston Toad	x			x		x							

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Bufo microscaphus	Arizona Toad	x			x		x	x						
Bufo nelsoni	Amargosa Toad	x			x		x	x						
Bufo punctatus	Red-spotted Toad	x			x		x	x						
Bufo retiformis	Sonoran Green Toad	x			x		x	x						
Bufo speciosus	Texas Toad	x			x		x	x						
Bufo woodhousii	Woodhouse's Toad	x		x			x							
Bufo alvarius	Colorado River Toad	x			x	x	x	x						
Bufo valliceps	Gulf Coast Toad	x			x		x	x						
Eleutherodactylus augusti	Barking Frog	x		x	x				x					
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	x		x	x									
Gastrophryne olivacea	Western Narrow-mouthed Toad	x		x	x		x							
Hyla arenicolor	Canyon Treefrog	x			x				x	x				
Hyla chrysoscelis	Cope's Gray Treefrog	x			x									1
Hyla cinerea	Green Treefrog	x			x									
Hyla squirella	Squirrel Treefrog	x			x									
Hyla versicolor	Gray Treefrog	x			x									1
Hyla wrightorum	Arizona Tree Frog	x			x				x	x				
Hypopachus variolosus	Sheep Frog	x			x					x				
Leptodactylus fragilis	Mexican White-lipped Frog	x			x					x				
Pseudacris cadaverina	California Tree Frog	x			x									
Pseudacris clarkii	Spotted Chorus Frog	x			x			x						
Pseudacris crucifer	Spring Peeper	x			x									
Pseudacris hypochondriaca	Baja California Treefrog	x			x									
Pseudacris maculata	Boreal Chorus Frog				x					x				
Pseudacris regilla	Northern Pacific Chorus Frog	x			x									
Pseudacris streckeri	Strecker's Chorus Frog	x			x			x						
Pseudacris triseriata	Western Chorus Frog	x			x				x	x				
Pternohyla fodiens	Lowland Burrowing Treefrog	x			x									
Rana areolata	Crawfish Frog	x			x									
Rana berlandieri	Rio Grande Leopard Frog	x			x			x						
Rana blairi	Plains Leopard Frog	x			x	x		x						
Rana catesbeiana	American Bullfrog	x			x		x	x	x					
Rana chiricahuensis	Chiricahua Leopard Frog	x			x	x		x						
Rana clamitans	Green Frog	x			x				x					
Rana grylio	Pig Frog	x			x		x		x					
Rana onca	Relict Leopard Frog	x			x			x						
Rana palustris	Pickerel Frog	x		x	x									
Rana pipiens	Northern Leopard Frog	x			x	x		x						
Rana sphenocephala	Southern Leopard Frog	x			x		x							
Rana sylvatica	Wood Frog				x			x						
Rana tarahumarae	Tarahumara Frog	x			x			x						
Rana yavapaiensis	Lowland Leopard Frog	x			x	x		x						
Rana aurora	Northern Red-legged Frog	x			x									
Rana boylii	Foothill Yellow-legged Frog	x			x									
Rana cascadae	Cascades Frog	x			x									
Rana draytonii	California Red-legged Frog	x			x									
Rana luteiventris	Columbia Spotted Frog	x			x			x						
Rana muscosa	Mountain Yellow-legged Frog	x			x			x						
Rana pretiosa	Oregon Spotted Frog	x			x									
Rana sierrae	Sierra Nevada Yellow-legged Frog	x			x									

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Rhinophrynus dorsalis	Burrowing Toad	x			x		x			x				
Scaphiopus couchii	Couch's Spadefoot	x			x			x		x				
Scaphiopus hurterii	Hurter's Spadefoot	x			x									
Smilisca baudinii	Mexican Treefrog	x			x			x						
Spea bombifrons	Plains Spadefoot	x			x		x	x		x				
Spea hammondi	Western Spadefoot	x			x									
Spea intermontana	Great Basin Spadefoot	x			x			x						
Spea multiplicata	Mexican Spadefoot	x			x		x	x		x				
Syrrhophus cystignathoides	Rio Grande Chirping Frog	x			x					x				
Syrrhophus gutillatus	Spotted Chirping Frog	x			x		x							
Syrrhophus marnockii	Cliff Chirping Frog	x			x		x			x				
NORTHWEST REGION														
Ascaphus montanus	Rocky Mountain Tailed Frog				x			x		x				
Ascaphus truei	Coastal Tailed Frog				x			x		x				
Bufo baxteri	Wyoming Toad			x	x									
Bufo boreas	Western Toad				x		x							
Bufo cognatus	Great Plains Toad				x			x						
Bufo hemiophrys	Canadian Toad			x	x			x						
Bufo woodhousii	Woodhouse's Toad				x		x							
Pseudacris maculata	Boreal Chorus Frog	x	x					x		x				
Pseudacris regilla	Northern Pacific Chorus Frog	x	x					x		x				14
Pseudacris sierra	Sierran Chorus Frog	x	x					x		x				14
Rana catesbeiana	American Bullfrog	x	x	x	x		x	x		x	x			
Rana clamitans	Green Frog	x	x	x	x		x	x		x	x			
Rana pipiens	Northern Leopard Frog	x	x	x	x		x	x		x	x			
Rana sylvatica	Wood Frog	x		x	x		x	x		x	x			
Rana aurora	Northern Red-legged Frog	x	x	x	x		x	x		x	x			13
Rana boylii	Foothill Yellow-legged Frog	x	x	x	x		x	x		x	x			
Rana cascadae	Cascades Frog	x	x	x	x		x	x		x	x			13
Rana draytonii	California Red-legged Frog	x	x	x	x		x	x		x	x			13
Rana luteiventris	Columbia Spotted Frog	x	x	x	x		x	x		x	x			
Rana pretiosa	Oregon Spotted Frog	x	x	x	x		x	x		x	x			13
Spea bombifrons	Plains Spadefoot	x	x		x		x	x						
Spea hammondii	Western Spadefoot	x	x		x		x	x						
Spea intermontana	Great Basin Spadefoot	x	x		x		x	x						
MIDWEST REGION														
Acris crepitans	Northern Cricket Frog	x	x				x							
Bufo americanus	American Toad	x	x								x			
Bufo cognatus	Great Plains Toad	x	x								x			
Bufo debilis	Green Toad	x	x											
Bufo fowleri	Fowler's Toad	x	x			x					x			17
Bufo hemiophrys	Canadian Toad	x	x				x							
Bufo punctatus	Red-spotted Toad	x	x				x							
Bufo woodhousii	Woodhouse's Toad	x	x								x			17
Gastrophryne carolinensis	Eastern Narrow-mouthed Toad	x	x	x			x							
Gastrophryne olivacea	Western Narrow-mouthed Toad	x	x	x			x							

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Hyla avivoca	Bird-voiced Treefrog	x	x	x		x								
Hyla chrysoscelis	Cope's Gray Treefrog	x	x			x		x						1
Hyla cinerea	Green Treefrog	x	x	x	x									
Hyla versicolor	Gray Treefrog	x	x			x		x						1
Pseudacris brachyphona	Mountain Chorus Frog	x	x		x		x							
Pseudacris clarkii	Spotted Chorus Frog	x	x		x		x							
Pseudacris crucifer	Spring Peeper	x	x		x			x						
Pseudacris feriarum	Upland Chorus Frog	x	x					x						
Pseudacris illinoensis	Illinois Chorus Frog	x	x	x		x		x						
Pseudacris maculata	Boreal Chorus Frog	x	x	x										18
Pseudacris streckeri	Strecker's Chorus Frog	x	x	x		x		x						
Pseudacris triseriata	Western Chorus Frog	x	x	x										18
Rana areolata	Crawfish Frog	x	x	x										
Rana blairi	Plains Leopard Frog	x	x	x	x		x							
Rana catesbeiana	American Bullfrog	x	x	x	x			x	x		x			
Rana clamitans	Green Frog	x	x	x	x			x	x		x			
Rana palustris	Pickerel Frog	x	x	x	x	x								
Rana pipiens	Northern Leopard Frog	x	x	x	x	x								
Rana septentrionalis	Mink Frog	x	x	x	x	x			x					
Rana sphenocephala	Southern Leopard Frog	x	x	x	x	x	x	x						
Rana sylvatica	Wood Frog	x	x		x			x						
Scaphiopus holbrookii	Eastern Spadefoot	x	x	x			x			x				
Spea bombifrons	Plains Spadefoot	x	x	x			x	x		x				

SALAMANDERS

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-web bing	toes	chin / throat	under-side of tail	side of tail	Comment code
SOUTHEAST REGION														
Ambystoma annulatum	Ringed Salamander	x			x									
Ambystoma barbouri	Streamside Salamander	x			x									2
Ambystoma bishopi	Reticulated Flatwoods Salamander	x			x									
Ambystoma cingulatum	Frosted Flatwoods Salamander	x			x									
Ambystoma jeffersonianum	Jefferson Salamander	x			x									
Ambystoma laterale	Blue-spotted Salamander	x			x									
Ambystoma mabeei	Mabee's Salamander	x			x									
Ambystoma maculatum	Spotted Salamander	x			x									
Ambystoma opacum	Marbled Salamander	x			x									
Ambystoma talpoideum	Mole Salamander	x			x									
Ambystoma texanum	Small-mouthed Salamander	x			x									2
Ambystoma tigrinum	Tiger Salamander	x			x									
Amphiuma means	Two-Toed Amphiuma	x			x					x				
Amphiuma pholeter	One-Toed Amphiuma	x			x					x				
Amphiuma tridactylum	Three-Toed Amphiuma	x			x					x				
Aneides aeneus	Green Salamander	x			x									
Cryptobranchus alleganiensis	Hellbender	x			x									
Desmognathus abditus	Cumberland Dusky Salamander	x			x									3
Desmognathus aeneus	Seepage Salamander	x			x		x							3
Desmognathus apalachicolae	Apalachicola Dusky Salamander	x		x	x									3
Desmognathus auriculatus	Southern Dusky Salamander	x		x	x									3
Desmognathus brimleyorum	Ouachita Dusky Salamander	x			x									3
Desmognathus carolinensis	Carolina Mountain Dusky Salamander	x			x									3
Desmognathus conanti	Spotted Dusky Salamander	x			x									3
Desmognathus folkertsi	Dwarf Black-bellied Salamander	x		x	x									3
Desmognathus fuscus	Northern Dusky Salamander	x			x									3
Desmognathus imitator	Imitator Salamander	x			x			x						3
Desmognathus marmoratus	Shovel-Nosed Salamander	x		x	x									3
Desmognathus monticola	Seal Salamander	x			x									3
Desmognathus ochrophaeus	Allegheny Mountain Dusky Salamander	x			x									3
Desmognathus ocoee	Ocoee Salamander	x			x									3
Desmognathus orestes	Blue Ridge Dusky Salamander	x			x									3
Desmognathus quadramaculatus	Black-bellied Salamander	x		x	x									3

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-web bing	toes	chin / throat	under-side of tail	side of tail	Comment code
Desmognathus santeetlah	Santeetlah Dusky Salamander	x		x	x									3
Desmognathus welteri	Black Mountain Salamander	x			x									3
Desmognathus wrighti	Pygmy Salamander	x			x		x							3
Eurycea aquatica	Brown-backed Salamander	x			x									
Eurycea chamberlaini	Chamberlain's Dwarf	x		x	x									
Eurycea cirrigera	Southern Two-lined Salamander	x			x								x	
Eurycea guttolineata	Three-lined Salamander	x			x									
Eurycea junaluska	Junaluska Salamander	x			x								x	
Eurycea longicauda	Long-tailed Salamander	x			x								x	
Eurycea lucifuga	Cave Salamander	x			x									
Eurycea multiplicata	Many-ribbed Salamander	x		x	x									
Eurycea quadridigitata	Dwarf Salamander	x		x	x									
Eurycea tynerensis	Oklahoma Salamander	x			x									
Eurycea wilderae	Blue Ridge Two-lined Salamander	x			x								x	
Gyrinophilus gulolineatus	Berry Cave Salamander	x			x		x				x			
Gyrinophilus palleucus	Tennessee Cave Salamander	x			x		x							
Gyrinophilus porphyriticus	Spring Salamander	x			x		x							
Haideotriton wallacei	Georgia Blind Salamander	x			x									
Hemidactylium scutatum	Four-toed Salamander	x			x									
Necturus alabamensis	Black Warrior River Waterdog	x		x	x									
Necturus beyeri	Gulf Coast Waterdog	x		x	x									
Necturus lewisi	Neuse River Waterdog	x		x	x									
Necturus maculosus	Mudpuppy	x		x	x									
Necturus punctatus	Dwarf Waterdog	x		x	x									
Notophthalmus perstriatus	Striped Newt	x			x									
Notophthalmus viridescens	Eastern Newt	x			x									
Phaeognathus hubrichti	Red Hills Salamander	x			x									
Plethodon ainsworthi	Bay Springs Salamander	x			x									
Plethodon amplus	Blue Ridge Gray-cheeked Salamander	x			x									
Plethodon angusticlavius	Ozark Zigzag Salamander	x			x									4, 19
Plethodon aureolus	Tellico Salamander	x			x						x			
Plethodon caddoensis	Caddo Mountain Salamander	x			x									
Plethodon chattahoochee	Chattahoochee Slimy Salamander	x			x									5
Plethodon cheoah	Cheoah Bald Salamander	x			x									
Plethodon cinereus	Eastern Red-backed Salamander	x			x									4, 19
Plethodon dorsalis	Northern Zigzag Salamander	x			x									4, 19
Plethodon electromorphus	Northern Ravine Salamander	x			x									
Plethodon fourchensis	Fourche Mountain Salamander	x			x									
Plethodon glutinosus	Northern Slimy Salamander	x			x									5

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-web bing	toes	chin / throat	under-side of tail	side of tail	Comment code
Plethodon grobmani	Southeastern Slimy Salamander	x			x									5
Plethodon jordani	Red-cheeked Salamander	x			x									
Plethodon kentucki	Cumberland Plateau Salamander	x		x	x					x				
Plethodon kisatchie	Louisiana Slimy Salamander	x			x									5
Plethodon meridianus	South Mountain Gray-cheeked Salamander	x			x									
Plethodon metcalfi	Southern Gray-cheeked Salamander	x			x									
Plethodon mississippi	Mississippi Slimy Salamander	x			x									5
Plethodon ocmulgee	Ocmulgee Slimy Salamander	x			x									5
Plethodon ouachitae	Rich Mountain Salamander	x			x									
Plethodon petraeus	Pigeon Mountain Salamander	x			x									
Plethodon richmondi	Southern Ravine Salamander	x		x	x						x			
Plethodon savannah	Savannah Slimy Salamander	x			x									5
Plethodon serratus	Southern Red-backed Salamander	x			x									4, 19
Plethodon shermani	Red-legged Salamander	x			x									
Plethodon teyahalee	Southern Appalachian Salamander	x			x									
Plethodon variolatus	South Carolina Slimy Salamander	x			x									5
Plethodon ventralis	Southern Zigzag Salamander	x			x									4, 19
Plethodon websteri	Webster's Salamander	x			x									4, 19
Plethodon wehrlei	Wehrle's Salamander	x			x									
Plethodon welleri	Weller's Salamander	x			x									
Plethodon yonahlossee	Yonahlossee Salamander	x			x									
Pseudobranchus axanthus	Southern Dwarf Siren	x			x					x				
Pseudobranchus striatus	Northern Dwarf Siren	x			x					x				
Pseudotriton montanus	Mud Salamander	x			x		x							
Pseudotriton ruber	Red Salamander	x			x		x							
Siren intermedia	Lesser Siren	x			x					x				6
Siren lacertina	Greater Siren	x			x					x				6
Stereochilus marginatus	Many-lined Salamander	x	x		x									
Typhlotriton spelaeus	Grotto Salamander	x			x									
Urspelerpes brucei	Patch-nosed Salamander	x			x		x							
NORTHEAST REGION														
Ambystoma barbouri	Streamside Salamander	x							x					
Ambystoma jeffersonianum	Jefferson Salamander	x		x							x			
Ambystoma laterale	Blue-spotted Salamander	x		x										
Ambystoma mabeei	Mabee's Salamander	x	x				x							
Ambystoma maculatum	Spotted Salamander	x												
Ambystoma opacum	Marbled Salamander	x												
Ambystoma talpoideum	Mole Salamander	x	x											

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-web bing	toes	chin / throat	under-side of tail	side of tail	Comment code
Ambystoma texanum	Small-mouthed Salamander	x							x					
Ambystoma tigrinum	Tiger Salamander	x												
Amphiuma means	Two-toed Amphiuma	x									x			
Aneides aeneus	Green Salamander	x												
Cryptobranchus alleganiensis	Hellbender	x	x											
Desmognathus auriculatus	Southern Dusky Salamander	x												3
Desmognathus fuscus	Northern Dusky Salamander	x		x										3
Desmognathus marmoratus	Shovel-nosed Salamander	x					x	x						3
Desmognathus monticola	Seal Salamander	x		x										3
Desmognathus ochrophaeus	Allegheny Mountain Dusky Salamander	x						x						3
Desmognathus orestes	Blue Ridge Dusky Salamander	x												3
Desmognathus quadramaculatus	Black-bellied Salamander	x	x	x										3
Desmognathus welteri	Black Mountain Salamander	x	x	x										3
Desmognathus wrighti	Pygmy Salamander	x		x										3
Eurycea bislineata	Northern Two-lined Salamander	x	x											
Eurycea cirrigera	Southern Two-lined Salamander	x												
Eurycea guttolineata	Three-lined Salamander	x												
Eurycea longicauda	Long-tailed Salamander	x												
Eurycea lucifuga	Cave Salamander	x												
Eurycea wilderae	Blue Ridge Two-lined Salamander	x	x											
Gyrinophilus porphyriticus	Spring Salamander	x					x	x						
Gyrinophilus subterraneus	West Virginia Spring Salamander	x												
Hemidactylium scutatum	Four-toed Salamander	x		x										
Necturus maculosus	Mudpuppy	x	x	x										
Necturus punctatus	Dwarf Waterdog	x	x	x										
Notophthalmus viridescens	Eastern Newt	x												
Plethodon chlorobryonis	Atlantic Coast Slimy Salamander	x												5
Plethodon cinereus	Eastern Red-backed Salamander	x												10
Plethodon cylindraceus	White-spotted Slimy Salamander	x												5
Plethodon electromorphus	Northern Ravine Salamander	x	x											
Plethodon glutinosus	Northern Slimy Salamander	x		x										5
Plethodon hoffmani	Valley and Ridge Salamander	x		x										
Plethodon hubrichti	Peaks of Otter Salamander	x	x											
Plethodon kentucki	Cumberland Plateau Salamander	x	x											5
Plethodon montanus	Northern Gray-cheeked salamander	x					x							

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-web bing	toes	chin / throat	under-side of tail	side of tail	Comment code
Plethodon nettingi	Cheat Mountain Salamander	x	x											
Plethodon punctatus	Cow Knob Salamander	x												
Plethodon richmondi	Southern Ravine Salamander	x	x											
Plethodon shenandoah	Shenandoah Salamander	x		x										10
Plethodon sherando	Big Levels Salamander	x												10
Plethodon ventralis	Southern Zigzag Salamander	x												
Plethodon virginia	Shenandoah Mountain Salamander	x												
Plethodon wehrlei	Wehrle's Salamander	x	x											
Plethodon welleri	Weller's Salamander	x	x											
Plethodon yonahlossee	Yonahlossee Salamander	x												
Pseudotriton montanus	Mud Salamander	x						x						
Pseudotriton ruber	Red Salamander	x						x						
Siren Intermedia	Lesser Siren	x												
Siren lacertina	Greater Siren	x												
Stereochilus marginatus	Many-lined Salamander	x	x				x							
SOUTHWEST REGION														
Ambystoma californiense	California Tiger Salamander	x	x			x								
Ambystoma gracile	Northwestern Salamander	x	x			x								
Ambystoma macrodactylum	Long-toed Salamander	x	x			x								
Ambystoma maculatum	Spotted Salamander	x	x			x								
Ambystoma mavortium	Barred Tiger Salamander	x	x			x								
Ambystoma opacum	Marbled Salamander	x	x			x								
Ambystoma talpoideum	Mole Salamander	x	x			x								
Ambystoma texanum	Small-mouthed Salamander	x	x			x								
Ambystoma tigrinum	Tiger Salamander	x	x		x	x								
Amphiuma tridactylum	Three-toed Salamander	x	x			x					x			
Aneides ferreus	Clouded Salamander	x	x		x									
Aneides flavipunctatus	Black Salamander	x	x		x									
Aneides hardii	Sacramento Mountains Salamander				x									
Aneides lugubris	Arboreal Salamander	x	x		x									
Aneides vagrans	Wandering Salamander	x	x		x									
Batrachoseps attenuatus	California Slender Salamander	x	x											15
Batrachoseps campi	Inyo Mountains Salamander	x	x											
Batrachoseps diabolicus	Hell Hollow Slender Salamander	x	x											
Batrachoseps gabrieli	San Gabriel Mountains Slender Salamander	x	x											
Batrachoseps gavilanensis	Gabilan Mountains Slender Salamander	x	x											
Batrachoseps gregarius	Gregarious Slender Salamander	x	x											
Batrachoseps incognitus	San Simeon Slender Salamander	x	x											
Batrachoseps kawia	Sequoia Slender Salamander	x	x											

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-web bing	toes	chin / throat	under-side of tail	side of tail	Comment code
Batrachoseps luciae	Santa Lucia Mountains Slender Salamander	x	x											
Batrachoseps major	Garden Slender Salamander	x	x											
Batrachoseps minor	Lesser Slender Salamander	x	x											
Batrachoseps nigriventris	Black-bellied Slender Salamander	x	x											15
Batrachoseps pacificus	Channel Islands Slender Salamander	x	x											
Batrachoseps regius	Kings River Slender Salamander	x	x											
Batrachoseps relictus	Relictual Slender Salamander	x	x											16
Batrachoseps robustus	Kern Plateau Salamander	x	x											
Batrachoseps simatus	Kern Canyon Slender Salamander	x	x											16
Batrachoseps stebbinsi	Tehachapi Slender Salamander	x	x											
Desmognathus auriculatus	Southern Dusky Salamander	x	x		x	x								
Dicamptodon ensatus	California Giant Salamander	x	x											
Dicamptodon tenebrosus	Coastal Giant Salamander	x	x											
Ensatina eschscholtzii	Ensatina	x	x											
Eurycea chisholmensis	Salado Salamander	x	x			x								
Eurycea latitans	Cascade Caverns Salamander	x	x			x								
Eurycea nana	San Marcos Salamander	x	x			x								
Eurycea naufragia	Georgetown Salamander	x	x			x								
Eurycea neotenes	Texas Salamander	x	x			x								
Eurycea pterophila	Fern Bank Salamander	x	x			x								
Eurycea quadridigitata	Dwarf Salamander	x	x		x	x								
Eurycea rathbuni	Texas Blind Salamander	x	x			x		x						
Eurycea robusta	Blanco Blind Salamander	x	x			x		x						
Eurycea sosorum	Barton Springs Salamander	x	x			x								
Eurycea tonkawae	Jollyville Plateau Salamander	x	x			x								
Eurycea tridentifera	Comal Blind Salamander	x	x			x								
Eurycea troglodytes	Valdina Farms Salamander	x	x			x								
Eurycea waterlooensis	Austin Blind Salamander	x	x			x								
Hydromantes brunus	Limestone Salamander	x	x											
Hydromantes platycephalus	Mount Lyell Salamander	x	x											
Hydromantes shastae	Shasta Salamander	x	x											
Necturus beyeri	Gulf Coast Waterdog	x	x		x	x								
Notophthalmus meridionalis	Black-spotted Newt	x	x			x								
Notophthalmus viridescens	Eastern Newt	x	x			x								
Plethodon albagula	Western Slimy Salamander	x	x			x						x		
Plethodon asupak	Scott Bar Salamander	x	x			x								
Plethodon dunni	Dunn's Salamander	x	x			x								
Plethodon elongatus	Del Norte Salamander	x	x			x								
Plethodon kiamichi	Kiamichi Slimy Salamander	x	x			x								
Plethodon neomexicanus	Jemez Mountains Salamander	x	x		x									

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-web bing	toes	chin / throat	under-side of tail	side of tail	Comment code
Plethodon sequoyah	Sequoyah Slimy Salamander	x	x			x								
Plethodon serratus	Southern Red-backed Salamander	x	x			x								
Plethodon stormi	Siskiyou Mountains Salamander	x	x			x								
Rhyacotriton variegatus	Southern Torrent Salamander	x	x											
Siren intermedia	Lesser Siren	x	x			x						x		
Taricha granulosa	Rough-skinned Newt	x	x											
Taricha rivularis	Red-bellied Newt	x	x											
Taricha torosa	California Newt	x	x											
NORTHWEST REGION														
Ambystoma californiense	California Tiger Salamander	x	x		x		x			x				
Ambystoma gracile	Northwestern Salamander	x	x		x		x			x				
Ambystoma macrodactylum	Long-toed Salamander	x	x		x		x			x				
Ambystoma mavortium	Barred Tiger Salamander	x	x		x		x			x				
Aneides ferreus	Clouded Salamander			x	x					x				
Aneides flavipunctatus	Black Salamander			x	x					x				
Aneides lugubris	Arboreal salamander			x	x					x				
Aneides vagrans	Wandering Salamander			x	x					x				
Batrachoseps attenuatus	California Slender Salamander			x	x					x				
Batrachoseps wrighti	Oregon Slender Salamander			x	x					x				
Dicamptodon aterrimus	Idaho Giant Salamander	x					x							
Dicamptodon copei	Cope's Giant Salamander	x					x							12
Dicamptodon ensatus	California Giant Salamander	x					x	x						
Dicamptodon tenebrosus	Coastal Giant Salamander	x					x							12
Ensatina eschscholtzii	Ensatina				x								x	
Hydromantes shastae	Shasta Salamander		x		x					x				
Plethodon asupak	Scott Bar Salamander	x	x	x						x				
Plethodon dunni	Dunn's Salamander	x	x	x						x				11
Plethodon elongatus	Del Norte Salamander	x	x	x						x				
Plethodon idahoensis	Coeur d'Alene Salamander	x	x	x						x	x			
Plethodon larselli	Larch Mountain Salamander	x	x	x						x	x			
Plethodon stormi	Siskiyou Mountains Salamander	x	x	x						x				
Plethodon vandykei	Van Dyke's Salamander	x	x	x						x	x			
Plethodon vehiculum	Western Red-backed Salamander	x	x	x						x				11
Rhyacotriton cascadae	Cascade Torrent Salamander			x	x									
Rhyacotriton kezeri	Columbia Torrent Salamander			x	x									
Rhyacotriton olympicus	Olympic Torrent Salamander			x	x									
Rhyacotriton variegatus	Southern Torrent Salamander			x	x									
Taricha granulosa	Rough-skinned Newt				x									
Taricha rivularis	Red-bellied Newt				x		x	x						
Taricha torosa	California Newt				x		x	x						

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-web bing	toes	chin / throat	under-side of tail	side of tail	Comment code
MIDWEST REGION														
Ambystoma annulatum	Ringed Salamander	x	x	x										
Ambystoma barbouri	Streamside Salamander	x	x	x										2
Ambystoma jeffersonianum	Jefferson Salamander	x	x	x						x		x		
Ambystoma laterale	Blue-spotted Salamander	x	x	x						x		x		
Ambystoma maculatum	Spotted Salamander	x	x	x										
Ambystoma mavortium	Barred Tiger Salamander	x	x	x				x						
Ambystoma opacum	Marbled Salamander	x	x	x										
Ambystoma talpoideum	Mole Salamander	x	x	x								x		
Ambystoma texanum	Small-mouthed Salamander	x	x	x				x		x				2
Ambystoma tigrinum	Tiger Salamander	x	x	x				x						
Amphiuma tridactylum	Three-toed Amphiuma	x	x					x		x				
Aneides aeneus	Green Salamander	x	x	x										
Cryptobranchus alleghaniensis	Hellbender	x	x											
Desmognathus conanti	Spotted Dusky Salamander	x	x	x									x	3
Desmognathus fuscus	Northern Dusky Salamander	x	x	x									x	3
Desmognathus ochrophaeus	Allegheny Mountain Dusky Salamander	x	x	x									x	3
Eurycea bislineata	Northern Two-lined Salamander	x	x	x									x	
Eurycea cirrigera	Southern Two-lined Salamander	x	x	x									x	
Eurycea longicauda	Long-tailed Salamander	x	x	x									x	
Eurycea lucifuga	Cave Salamander	x	x	x										
Eurycea multiplicata	Many-ribbed Salamander	x	x	x										
Eurycea tynerensis	Oklahoma Salamander	x	x	x				x					x	
Gyrinophilus porphyriticus	Spring Salamander	x	x	x				x						
Hemidactylium scutatum	Four-toed Salamander	x	x	x						x				
Necturus maculosus	Mudpuppy	x	x	x										
Notophthalmus viridescens	Eastern Newt	x	x	x										
Plethodon albagula	Western Slimy Salamander	x	x	x										5, 20
Plethodon angusticlavius	Ozark Zigzag Salamander	x	x	x										19
Plethodon cinereus	Eastern Red-backed Salamander	x	x	x										19
Plethodon dorsalis	Northern Zigzag Salamander	x	x	x										19
Plethodon electromorphus	Northern Ravine Salamander	x	x	x										20
Plethodon glutinosus	Northern Slimy Salamander	x	x	x										5, 20
Plethodon serratus	Southern Red-backed Salamander	x	x	x										19
Plethodon wehrlei	Wehrle's Salamander	x	x	x										20
Pseudotriton montanus	Mud Salamander	x	x	x				x						
Pseudotriton ruber	Red Salamander	x	x	x				x						
Siren intermedia	Lesser Siren	x	x	x				x		x				
Typhlotriton spelaeus	Grotto Salamander	x	x	x			x	x						

TURTLES

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-web bing	toes	chin / throat	under-side of tail	side of tail	Comment code
SOUTHEAST REGION														
Apalone ferox	Florida Softshell	x			x									
Apalone mutica	Smooth Softshell	x			x									
Apalone spinifera	Spiny Softshell	x			x									
Caretta caretta	Loggerhead Sea Turtle	x			x		x							
Chelonia mydas	Green Sea Turtle	x			x		x							
Chelydra serpentina	Snapping Turtle	x			x									
Chrysemys dorsalis	Southern Painted Turtle	x		x	x									
Chrysemys picta	Painted Turtle	x		x	x									
Clemmys guttata	Spotted Turtle	x			x									
Deirochelys reticularia	Chicken Turtle	x		x	x	x								
Dermochelys coriacea	Leatherback Sea Turtle	x			x									
Eretmochelys imbricata	Hawksbill Sea Turtle	x			x		x							
Glyptemys muhlenbergii	Bog Turtle	x			x									
Gopherus polyphemus	Gopher Tortoise	x			x									
Graptemys barbouri	Barbour's Map Turtle	x		x	x		x				x			
Graptemys ernsti	Escambia Map Turtle	x		x	x		x				x			
Graptemys flavimaculata	Yellow-blotched Map Turtle	x		x	x									
Graptemys geographica	Northern Map Turtle	x		x	x		x							
Graptemys gibbonsi	Pascagoula Map Turtle	x		x	x		x							
Graptemys nigrinoda	Black-knobbed Map Turtle	x		x	x									
Graptemys oculifera	Ringed Map Turtle	x		x	x									
Graptemys ouachitensis	Ouachita Map Turtle	x		x	x		x							
Graptemys pearlensis	Pearl River Map Turtle	x		x	x		x							
Graptemys pseudogeographica	False Map Turtle	x		x	x		x							
Graptemys pulchra	Alabama Map Turtle	x		x	x		x							
Kinosternon baurii	Striped Mud Turtle	x		x	x			x						
Kinosternon subrubrum	Eastern Mud Turtle	x		x	x			x						
Lepidochelys kempii	Kemp's Ridley Sea Turtle	x			x		x							
Macroclemys temminckii	Alligator Snapping Turtle	x			x									
Malaclemys terrapin	Diamond-backed Terrapin	x			x									
Pseudemys alabamensis	Alabama Red-Bellied Cooter	x		x	x									
Pseudemys concinna	River Cooter	x		x	x									
Pseudemys nelsoni	Florida Red-bellied Cooter	x		x	x									
Pseudemys peninsularis	Peninsula Cooter	x		x	x									
Pseudemys rubriventris	Northern Red-bellied Cooter	x		x	x									
Pseudemys suwanniensis	Suwanee Cooter	x		x	x									
Sternotherus carinatus	Razor-backed Musk Turtle	x		x	x			x						
Sternotherus depressus	Flattened Musk Turtle	x		x	x									
Sternotherus minor	Loggerhead Musk Turtle	x		x	x			x						
Sternotherus odoratus	Eastern Musk Turtle	x		x	x			x						
Terrapene carolina	Eastern Box Turtle	x		x	x									
Trachemys scripta	Pond Slider	x		x	x	x								

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-web bing	toes	chin / throat	under-side of tail	side of tail	Comment code
NORTHEAST REGION														
Apalone mutica	Smooth Softshell	x		x										
Apalone spinifera	Spiny Softshell	x		x										
Caretta caretta	Loggerhead Sea Turtle	x	x		x		x	x						
Chelonia mydas	Green Sea Turtle	x	x		x		x	x						
Chelydra serpentina	Snapping Turtle			x										
Chrysemys picta	Painted Turtle	x		x										
Clemmys guttata	Spotted Turtle	x		x										
Deirochelys reticularia	Chicken Turtle	x		x	x	x		x						
Dermochelys coriacea	Leatherback Sea Turtle	x	x		x		x	x						
Emydoidea blandingii	Blanding's Turtle	x		x										
Eretmochelys imbricata	Hawksbill Sea Turtle	x	x		x		x	x						
Glyptemys insculpta	Wood Turtle	x		x										
Glyptemys muhlenbergii	Bog Turtle	x		x				x						
Graptemys geographica	Northern Map Turtle	x		x				x						
Graptemys pseudogeographica	False Map Turtle	x		x				x						
Kinosternon baurii	Striped Mud Turtle	x						x						
Kinosternon subrubrum	Eastern Mud Turtle	x		x										
Lepidochelys kempii	Kemp's Ridley Sea Turtle	x	x		x		x	x						
Malaclemys terrapin	Diamond-backed Terrapin	x		x										
Pseudemys concinna	River Cooter	x		x	x		x	x						
Pseudemys rubriventris	Northern Red-bellied Cooter	x		x			x	x						
Sternotherus minor	Loggerhead Musk Turtle	x					x	x						
Sternotherus odoratus	Eastern Musk Turtle	x		x				x			x			
Terrapene carolina	Eastern Box Turtle	x		x										
Trachemys scripta	Pond Slider	x		x		x		x						
SOUTHWEST REGION														
Actinemys marmorata	Western Pond Turtle	x			x			x						
Apalone mutica	Smooth Softshell	x	x		x			x						
Apalone spinifera	Spiny Softshell	x	x		x			x						
Caretta caretta	Loggerhead Sea Turtle	x	x		x		x							
Chelonia mydas	Green Sea Turtle	x	x				x	x						
Chelydra serpentina	Snapping Turtle	x	x		x									
Chrysemys picta	Painted Turtle	x	x		x		x							
Deirochelys reticularia	Chicken Turtle	x	x	x	x	x	x	x	x					
Dermochelys coriacea	Leatherback Sea Turtle	x	x				x							
Eretmochelys imbricata	Hawksbill Sea Turtle	x	x				x	x						
Gopherus agassizii	Desert Tortoise	x			x									
Gopherus berlandieri	Texas Tortoise	x	x				x							
Graptemys caglei	Cagle's Map Turtle	x	x		x		x	x	x			x		
Graptemys ouachitensis	Ouachita Map Turtle	x	x		x		x	x	x			x		
Graptemys pseudogeographica	False Map Turtle	x	x		x		x	x	x			x		
Graptemys versa	Texas Map Turtle	x	x		x		x	x	x			x		
Kinosternon arizonense	Arizona Mud Turtle													

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-web bing	toes	chin / throat	under-side of tail	side of tail	Comment code
Kinosternon flavescens	Yellow Mud Turtle	x	x		x		x	x						
Kinosternon hirtipes	Rough-footed Mud Turtle	x	x		x		x							
Kinosternon sonoriense	Sonoran Mud Turtle	x			x			x						
Kinosternon subrubrum	Eastern Mud Turtle	x	x		x		x							
Lepidochelys kempii	Kemp's Ridley Sea Turtle	x	x		x		x							
Lepidochelys olivacea	Olive Ridley Sea Turtle	x	x		x		x							
Macroclemys temminckii	Alligator Snapping Turtle	x	x				x							
Malaclemys terrapin	Diamond-backed Terrapin	x	x		x		x	x	x					
Pseudemys concinna	River Cooter	x	x				x		x					
Pseudemys gorzugi	Rio Grande Cooter	x	x		x		x	x	x					
Pseudemys texana	Texas Cooter	x	x				x		x					
Sternotherus carinatus	Razor-backed Musk Turtle	x	x		x		x		x					
Sternotherus odoratus	Eastern Musk Turtle	x	x		x		x		x					
Terrapene carolina	Eastern Box Turtle	x	x				x							
Terrapene ornata	Ornate Box Turtle	x	x		x		x							
Trachemys gaigae	Mexican Plateau Slider	x	x		x		x	x	x					
Trachemys scripta	Pond Slider	x			x	x		x						
NORTHWEST REGION														
Actinemys marmorata	Western Pond Turtle	x		x	x		x	x			x	x		
Apalone spinifera	Spiny Softshell	x			x		x	x						
Caretta caretta	Loggerhead Sea Turtle	x	x		x		x	x						
Chelonia mydas	Green Sea Turtle	x	x	x	x		x	x						
Chelydra serpentina	Snapping Turtle	x			x		x	x						
Chrysemys picta	Painted Turtle	x		x	x		x	x			x	x		
Dermochelys coriacea	Leather back Sea Turtle	x	x		x		x	x						
Lepidochelys olivacea	Olive Ridley Sea Turtle	x	x		x		x	x						
Terrapene ornata	Ornate Box Turtle	x		x	x	x	x	x	x	x	x	x		
Trachemys scripta	Pond Slider	x		x	x	x	x	x	x	x	x	x		
MIDWEST REGION														
Apalone mutica	Smooth Softshell	x					x	x						
Apalone spinifera	Spiny Softshell	x					x	x						
Chelydra serpentina	Snapping Turtle	x	x		x			x			x		x	
Chrysemys dorsalis	Southern Painted Turtle	x	x	x				x						
Chrysemys picta	Painted Turtle	x	x	x				x						
Clemmys guttata	Spotted Turtle	x	x	x				x						
Deirochelys reticularia	Chicken Turtle	x	x	x								x		
Emydoidea blandingii	Blanding's Turtle	x	x	x				x			x			
Glyptemys insculpta	Wood Turtle	x	x	x				x						
Graptemys geographica	Northern Map Turtle	x	x	x			x	x			x			
Graptemys ouachitensis	Ouachita Map Turtle	x	x	x			x	x			x			
Graptemys pseudogeographica	False Map Turtle	x	x	x			x	x			x			
Kinosternon flavescens	Yellow Mud Turtle	x	x	x				x						
Kinosternon subrubrum	Eastern Mud Turtle	x	x	x				x						
Macrochelys temminckii	Alligator Snapping Turtle	x	x		x			x			x		x	

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-web bing	toes	chin / throat	under-side of tail	side of tail	Comment code
Pseudemys concinna	River Cooter	x	x	x				x						
Sternotherus odoratus	Eastern Musk Turtle	x	x	x				x						
Terrapene carolina	Eastern Box Turtle	x	x	x										
Terrapene ornata	Ornate Box Turtle	x	x	x										
Trachemys scripta	Pond Slider	x	x	x				x						

LIZARDS

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
SOUTHEAST REGION														
Amphisbaenians														
Rhineura floridana	Florida Worm Lizard	x			x									
Lizards														
Anolis carolinensis	Green Anole	x			x									
Cnemidophorus sexlineatus	Six-lined Racerunner	x			x									
Eumeces anthracinus	Coal Skink	x			x									
Eumeces egregius	Mole Skink	x			x									
Eumeces fasciatus	Common Five-lined Skink	x			x			x					x	
Eumeces inexpectatus	Southeastern Five-lined Skink	x			x			x					x	
Eumeces laticeps	Broad-headed Skink	x			x			x					x	
Neoseps reynoldsi	Florida Sand Skink	x			x									
Ophisaurus attenuatus	Slender Glass Lizard	x	x				x							7
Ophisaurus compressus	Island Glass Lizard	x	x				x							
Ophisaurus mimicus	Mimic Glass Lizard	x	x				x							7
Ophisaurus ventralis	Eastern Glass Lizard	x	x				x							
Sceloporus undulatus	Eastern Fence Lizard	x			x									
Sceloporus woodi	Florida Scrub Lizard	x			x									
Scincella lateralis	Little Brown Skink	x			x									
Sphaerodactylus notatus	Reef Gecko	x			x									
NORTHEAST REGION														
Cnemidophorus sexlineatus	Six-lined Racerunner	x												
Eumeces anthracinus	Coal Skink	x												
Eumeces fasciatus	Common Five-lined Skink	x					x					x		
Eumeces inexpectatus	Southeastern Five-lined Skink	x					x					x		
Eumeces laticeps	Broad-headed Skink	x					x					x		
Ophisaurus attenuatus	Slender Glass Lizard	x												
Ophisaurus ventralis	Eastern Glass Lizard	x												
Sceloporus undulatus	Eastern Fence Lizard	x												
Scincella lateralis	Little Brown Skink	x			x									
SOUTHWEST REGION														
Anniella pulchra	California Legless Lizard	x	x											
Anolis carolinensis	Green Anole	x	x			x						x		
Callisaurus draconoides	Zebra-tailed Lizard	x		x	x							x		
Cnemidophorus arizonae	Arizona Striped Whiptail	x	x			x	x	x						
Cnemidophorus burti	Canyon Spotted Whiptail	x	x		x		x	x			x			
Cnemidophorus dixoni	Gray Checkered Whiptail	x	x		x	x	x	x			x			
Cnemidophorus exsanguis	Chihuahuan Spotted Whiptail	x	x		x	x	x	x			x	x		
Cnemidophorus flagellicauda	Gila Spotted Whiptail	x	x	x	x		x	x			x			
Cnemidophorus gularis	Common Spotted Whiptail	x	x	x	x	x	x	x			x			

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Cnemidophorus gypsi	Little White Whiptail	x	x			x	x	x						
Cnemidophorus hyperythra	Orange-throated Whiptail	x	x	x		x	x	x			x			
Cnemidophorus inornata	Little Striped Whiptail	x	x	x	x	x	x	x			x	x		
Cnemidophorus laredoensis	Laredo Striped Whiptail	x	x			x							x	
Cnemidophorus marmorata	Marbled Whiptail	x	x		x	x	x	x			x			
Cnemidophorus neomexicana	New Mexico Whiptail	x	x		x	x	x	x			x			
Cnemidophorus neotesselata	Colorado Checkered Whiptail	x	x			x	x	x						
Cnemidophorus pai	Pai Striped Whiptail	x	x			x	x	x						
Cnemidophorus scalaris	Plateau Spotted Whiptail	x	x			x						x		
Cnemidophorus sexlineatus	Six-lined Racerunner	x	x		x	x	x	x			x			
Cnemidophorus sonorae	Sonoran Spotted Whiptail	x	x	x	x		x	x			x			
Cnemidophorus tesselata	Common Checkered Whiptail	x	x		x	x	x	x			x			
Cnemidophorus tigris	Tiger Whiptail	x	x		x		x	x			x			
Cnemidophorus uniparens	Desert Grassland Whiptail	x	x		x	x	x	x			x			
Cnemidophorus velox	Plateau Striped Whiptail	x	x	x	x		x	x			x			
Cnemidophorus xanthonota	Red-backed Whiptail	x	x			x	x	x						
Coleonyx brevis	Texas Banded Gecko	x	x		x	x								
Coleonyx reticulatus	Reticulated Gecko	x	x			x								
Coleonyx switaki	Switak's Banded Gecko	x	x		x									
Coleonyx variegatus	Western Banded Gecko	x	x		x									
Cophosaurus texanus	Greater Earless Lizard	x	x	x	x			x				x		
Crotaphytus bicinctores	Great Basin Collared Lizard	x			x									
Crotaphytus collaris	Eastern Collared Lizard	x	x		x	x								
Crotaphytus nebrius	Sonoran Collared Lizard	x	x		x	x								
Crotaphytus reticulatus	Reticulate Collared Lizard	x	x			x								
Crotaphytus vestigium	Baja California Collared Lizard	x	x		x	x								
Dipsosaurus dorsalis	Desert Iguana	x			x									
Elgaria coerulea	Northern Alligator Lizard	x		x	x			x						9
Elgaria kingii	Madrean Alligator Lizard	x			x			x						
Elgaria multicarinata	Southern Alligator Lizard	x		x	x									9
Elgaria panamintina	Panamint Alligator Lizard	x			x			x						
Eumeces anthracinus	Coal Skink	x	x			x		x				x		
Eumeces callicephalus	Mountain Skink				x		x							
Eumeces fasciatus	Common Five-lined Skink	x	x			x			x					
Eumeces gilberti	Gilbert's Skink	x			x									
Eumeces laticeps	Broad-headed Skink	x	x			x			x					
Eumeces multivirgatus	Many-lined Skink	x	x		x	x								
Eumeces obsoletus	Great Plains Skink	x	x	x	x			x						
Eumeces septentrionalis	Prairie Skink	x	x			x		x				x		
Eumeces skiltonianus	Western Skink	x	x		x									
Eumeces tetragrammus	Four-lined Skink	x	x			x		x	x					
Gambelia copeii	Cope's Leopard Lizard	x	x		x	x								
Gambelia silus	Blunt-nosed Leopard Lizard	x	x		x	x					x			
Gambelia wislizenii	Long-nosed Leopard Lizard	x	x		x	x					x			
Gerrhonotus infernalis	Texas Alligator Lizard	x	x	x										
Heloderma suspectum	Gila Monster				x									
Holbrookia elegans	Elegant Earless Lizard		x		x									
Holbrookia lacerata	Spot-tailed Earless Lizard	x	x			x							x	
Holbrookia maculata	Common Lesser Earless Lizard	x	x		x	x		x				x	x	

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Holbrookia propinqua	Keeled Earless Lizard	x	x			x							x	
Ophisaurus attenuatus	Slender Glass Lizard	x	x	x										
Petrosaurus mearnsi	Banded Rock Lizard	x			x								x	
Phrynosoma cornutum	Texas Horned Lizard	x	x		x	x	x	x						
Phrynosoma coronatum	Blainville's Horned Lizard	x	x		x		x	x			x			
Phrynosoma douglasii	Pygmy Short-horned Lizard	x			x		x							
Phrynosoma goodei	Goode's Horned Lizard	x	x		x									
Phrynosoma hernandesi	Greater Short-horned Lizard	x	x		x		x	x						
Phrynosoma mcallii	Flat-tailed Horned Lizard	x			x		x	x					x	
Phrynosoma modestum	Round-tailed Horned Lizard	x	x		x	x	x	x					x	
Phrynosoma platyrhinos	Desert Horned Lizard	x			x		x				x			
Phrynosoma solare	Regal Horned Lizard	x			x		x							
Phyllodactylus xanti	Peninsular Leaf-toed Gecko	x	x		x					x				
Sauromalus obesus	Common Chuckwalla	x	x		x									
Sceloporus arenicolus	Dunes Sagebrush Lizard	x	x	x	x	x		x				x		
Sceloporus bimaculosus	Twin-spotted Spiny Lizard	x	x		x	x		x						
Sceloporus clarkii	Clark's Spiny Lizard		x	x	x		x	x			x			
Sceloporus consobrinus	Prairie Lizard			x	x	x		x				x		
Sceloporus cowlesi	Southwestern Fence Lizard			x	x	x		x				x		
Sceloporus cyanogenys	Blue Spiny Lizard	x	x		x	x		x						
Sceloporus graciosus	Common Sagebrush Lizard	x	x	x	x	x					x			
Sceloporus grammicus	Graphic Spiny Lizard	x	x		x	x			x			x		
Sceloporus jarrovii	Yarrow's Spiny Lizard				x			x						
Sceloporus magister	Desert Spiny Lizard	x	x	x	x	x		x			x			
Sceloporus merriami	Canyon Lizard	x	x		x	x		x				x		
Sceloporus occidentalis	Western Fence Lizard	x		x	x	x					x			
Sceloporus olivaceus	Texas Spiny Lizard	x	x		x	x		x						
Sceloporus orcutti	Granite Spiny Lizard	x	x	x	x	x		x			x			
Sceloporus poinsettii	Crevice Spiny Lizard	x	x		x	x		x						
Sceloporus slevini	Slevin's Bunchgrass Lizard			x	x	x		x						
Sceloporus tristichus	Plateau Fence Lizard	x	x	x	x	x		x				x		
Sceloporus undulatus	Eastern Fence Lizard	x	x		x	x		x						
Sceloporus uniformis	Yellow-backed Spiny Lizard	x	x	x	x			x			x			
Sceloporus variabilis	Rose-bellied Lizard	x	x	x	x	x								
Sceloporus virgatus	Striped Plateau Lizard			x	x	x					x			
Scincella lateralis	Little Brown Skink	x	x			x		x						
Uma inornata	Coachella Fringe-toed Lizard	x		x	x					x	x			
Uma notata	Colorado Fringe-toed Lizard	x	x		x					x	x			
Uma rufopunctata	Yuman Fringe-toed Lizard	x			x					x				
Uma scoparia	Mohave Fringe-toed Lizard	x			x					x	x			
Urosaurus graciosus	Long-tailed Brush Lizard	x			x			x						
Urosaurus nigricaudus	Baja California Brush Lizard	x	x		x	x		x						
Urosaurus ornatus	Ornate Tree Lizard	x	x		x	x		x						
Uta stansburiana	Common Side-blotched Lizard	x	x		x	x		x						
Xantusia arizonae	Arizona Night Lizard	x	x		x			x						
Xantusia bezyi	Bezy's Night Lizard	x	x		x			x						
Xantusia henshawi	Granite Night Lizard	x	x		x			x						
Xantusia riversiana	Island Night Lizard	x	x		x			x						

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Xantusia sierrae	Sierra Night Lizard	x	x		x			x						
Xantusia vigilis	Desert Night Lizard	x	x		x			x						
Xantusia wigginsi	Wiggins' Night Lizard	x	x		x			x						
NORTHWEST REGION														
Cnemidophorus tigris	Tiger Whiptail	x	x		x			x			x			
Cnemidophorus velox	Plateau Striped Whiptail	x	x		x			x			x			
Crotaphytus bicinctores	Great Basin Collared Lizard	x	x		x			x						
Elgaria coerulea	Northern Alligator Lizard	x	x		x			x						9
Elgaria multicarinata	Southern Alligator Lizard	x	x		x			x						9
Eumeces gilberti	Gilbert's Skink	x	x		x			x						
Eumeces multivirgatus	Many-lined Skink	x	x		x			x						
Eumeces skiltonianus	Western Skink	x	x		x			x						
Gambelia wislizenii	Long-nosed Leopard Lizard	x	x		x			x						
Holbrookia maculata	Common Lesser Earless Lizard	x	x		x			x						
Phrynosoma coronatum	Blainville's Horned Lizard	x	x		x			x						
Phrynosoma douglasii	Pygmy Short-horned Lizard	x	x		x			x						
Phrynosoma hernandesi	Greater Short-horned Lizard	x	x		x			x						
Phrynosoma platyrhinos	Desert Horned Lizard	x	x		x			x						
Sceloporus graciosus	Common Sagebrush Lizard	x	x	x	x	x		x						
Sceloporus occidentalis	Western Fence Lizard	x	x	x	x	x		x						
Sceloporus undulatus	Eastern Fence Lizard	x	x	x	x	x		x						
Urosaurus ornatus	Ornate Tree Lizard	x	x		x	x		x						
Uta stansburiana	Common Side-blotched Lizard	x	x		x	x		x						
MIDWEST REGION														
Cnemidophorus sexlineatus	Six-lined Racerunner	x	x	x										
Crotaphytus collaris	Eastern Collared Lizard	x	x	x										
Eumeces anthracinus	Coal Skink	x	x		x		x				x			
Eumeces fasciatus	Common Five-lined Skink	x	x					x						
Eumeces laticeps	Broad-headed Skink	x	x					x						
Eumeces multivirgatus	Many-lined Skink	x	x								x			
Eumeces obsoletus	Great Plains Skink	x	x											
Eumeces septentrionalis	Prairie Skink	x	x					x			x			
Holbrookia maculata	Common Lesser Earless Lizard	x	x	x				x				x		
Ophisaurus attenuatus	Slender Glass Lizard	x	x					x						
Phrynosoma cornutum	Texas Horned Lizard	x	x				x	x						
Phrynosoma hernandesi	Greater Short-horned Lizard	x	x				x	x						
Sceloporus consobrinus	Prairie Lizard	x	x	x							x			
Sceloporus graciosus	Common Sagebrush Lizard	x	x	x							x			
Sceloporus undulatus	Eastern Fence Lizard	x	x	x		x					x			
Scincella lateralis	Little Brown Skink	x	x		x			x						

SNAKES

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
SOUTHEAST REGION														
Agkistrodon contortrix	Copperhead	x			x									
Agkistrodon piscivorus	Cottonmouth	x			x									
Carphophis amoenus	Eastern Worm Snake	x			x		x							
Carphophis vermis	Western Worm Snake	x			x		x							
Cemophora coccinea	Scarlet Snake	x		x	x									
Clonophis kirtlandii	Kirtland's Snake	x			x									
Coluber constrictor	North American Racer	x			x									
Crotalus adamanteus	Eastern Diamond-backed Rattlesnake	x			x									
Crotalus atrox	Western Diamond-backed Rattlesnake	x			x									
Crotalus horridus	Timber Rattlesnake	x			x									
Diadophis punctatus	Ring-necked Snake	x			x									
Drymarchon couperi	Eastern Indigo Snake	x			x									
Drymarchon melanurus	Central American Indigo Snake	x			x									
Elaphe guttata	Red Corn Snake	x			x									
Elaphe obsoleta	Eastern Rat Snake	x			x									
Elaphe slowinskii	Slowinski's Corn Snake	x			x									
Farancia abacura	Red-bellied Mud Snake	x			x									
Farancia erytrogramma	Rainbow Snake	x			x									
Heterodon platirhinos	Eastern Hog-nosed Snake	x		x	x			x						
Heterodon simus	Southern Hog-nosed Snake	x		x	x			x						
Lampropeltis calligaster	Yellow-bellied Kingsnake	x			x									
Lampropeltis getula	Common Kingsnake	x			x									
Lampropeltis triangulum	Milk Snake	x			x									
Masticophis flagellum	Coachwhip	x			x									
Micrurus fulvius	Harlequin Coral Snake	x			x									
Nerodia clarkii	Saltmarsh Watersnake	x			x									
Nerodia cyclopion	Mississippi Green Watersnake	x		x	x									
Nerodia erythrogaster	Plain-bellied Watersnake	x		x	x									
Nerodia fasciata	Southern Watersnake	x		x	x									
Nerodia floridana	Florida Green Watersnake	x		x	x									
Nerodia rhombifer	Diamond-backed Watersnake	x		x	x									
Nerodia sipedon	Northern Watersnake	x		x	x									
Nerodia taxispilota	Brown Watersnake	x		x	x									
Opheodrys aestivus	Rough Green Snake	x			x									
Opheodrys vernalis	Smooth Green Snake	x			x									
Pituophis melanoleucus	Pine Snake	x			x									
Pituophis ruthveni	Louisiana Pine Snake	x			x									
Regina alleni	Striped Crayfish Snake	x			x									
Regina grahamii	Graham's Crayfish Snake	x		x	x									
Regina rigida	Glossy Crayfish Snake	x		x	x									
Regina septemvittata	Queen Snake	x		x	x									
Rhadinaea flavilata	Pine Woods Snake	x			x									

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Seminatrix pygaea	Black Swamp Snake	x		x	x									
Sistrurus miliarius	Pygmy Rattlesnake	x			x									
Sonora semiannulata	Western Ground Snake	x			x									
Stilosoma extenuatum	Short-tailed Snake	x			x									
Storeria dekayi	Dekay's Brown Snake	x			x									
Storeria occipitomaculata	Red-bellied Snake	x		x	x									
Storeria victa	Florida Brown Snake	x			x									
Tantilla coronata	Southeastern Crowned Snake	x			x			x						
Tantilla gracilis	Flat-headed Snake	x			x			x						
Tantilla oolitica	Rim Rock Crowned Snake	x			x									
Tantilla relicta	Florida Crowned Snake	x			x			x						
Thamnophis proximus	Western Ribbon Snake	x			x		x							
Thamnophis sauritus	Eastern Ribbon Snake	x			x			x						
Thamnophis sirtalis	Common Garter Snake	x			x			x						
Virginia striatula	Rough Earth Snake	x			x		x							
Virginia valeriae	Smooth Earth Snake	x			x		x							
NORTHEAST REGION														
Agkistrodon contortrix	Copperhead	x						x						
Agkistrodon piscivorus	Cottonmouth							x						
Carphophis amoenus	Eastern Worm Snake	x		x									x	
Cemophora coccinea	Scarlet Snake	x		x				x						
Clonophis kirtlandii	Kirtland's Snake	x		x										
Coluber constrictor	North American Racer	x		x				x						
Crotalus horridus	Timber Rattlesnake	x						x					x	
Diadophis punctatus	Ring-necked Snake	x		x										
Elaphe guttata	Red Corn Snake	x		x										
Elaphe obsoleta	Eastern Rat Snake	x		x										
Farancia abacura	Red-bellied Mud Snake	x	x											
Farancia erytrogramma	Rainbow Snake	x												
Heterodon platirhinos	Eastern Hog-nosed Snake	x		x				x						
Lampropeltis calligaster	Yellow-bellied Kingsnake	x												
Lampropeltis getula	Common Kingsnake	x												
Lampropeltis triangulum	Milk Snake	x					x							
Nerodia erythrogaster	Plain-bellied Watersnake	x		x										
Nerodia sipedon	Northern Watersnake	x		x										
Nerodia taxispilota	Brown Watersnake	x												
Opheodrys aestivus	Rough Green Snake	x												
Opheodrys vernalis	Smooth Green Snake	x												
Pituophis melanoleucus	Pine Snake	x		x										
Regina rigida	Glossy Crayfish Snake	x		x										
Regina septemvittata	Queen Snake	x	x											
Sistrurus catenatus	Massasauga	x						x					x	
Storeria dekayi	Dekay's Brown Snake	x						x						
Storeria occipitomaculata	Red-bellied Snake	x		x			x							
Tantilla coronata	Southeastern Crowned Snake	x					x							
Thamnophis brachystoma	Short-headed Garter Snake	x					x							
Thamnophis sauritus	Eastern Ribbon Snake	x	x											

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Thamnophis sirtalis	Common Garter Snake	x												
Virginia striatula	Rough Earth Snake	x					x							
Virginia valeriae	Smooth Earth Snake	x						x						
SOUTHWEST REGION														
Agkistrodon contortrix	Copperhead	x	x											
Agkistrodon piscivorus	Cottonmouth	x	x											
Arizona elegans	Glossy Snake	x	x		x									
Carphophis vermis	Western Worm Snake	x	x					x						
Cemophora coccinea	Scarlet Snake	x	x		x									
Charina bottae	Northern Rubber Boa	x			x		x	x						
Charina trivirgata	Rosy Boa	x	x		x		x	x						
Chilomeniscus cinctus	Burrowing Sand Snake	x	x		x		x							
Chionactus occipitalis	Western Shovel-nosed Snake	x			x		x	x						
Chionactus palarostris	Sonoran Shovel-nosed Snake	x	x		x		x	x						
Coluber constrictor	North American Racer	x			x									
Coniophanes imperialis	Regal Black-striped Snake	x	x											
Contia tenuis	Sharp-tailed Snake	x	x	x	x								x	
Crotalus atrox	Western Diamond-backed Rattlesnake	x	x		x		x						x	
Crotalus cerastes	Sidewinder	x			x			x						
Crotalus cerberus	Arizona Black Rattlesnake	x			x									
Crotalus lepidus	Rock Rattlesnake	x	x		x									
Crotalus mitchellii	Speckled Rattlesnake	x			x		x	x						
Crotalus molossus	Black-tailed Rattlesnake	x	x		x									
Crotalus oreganus	Western Rattlesnake	x			x									8
Crotalus pricei	Twin-spotted Rattlesnake	x	x		x		x							
Crotalus ruber	Red Diamond Rattlesnake	x	x		x		x	x					x	
Crotalus scutulatus	Mohave Rattlesnake	x	x		x		x	x						
Crotalus stephensi	Panamint Rattlesnake	x			x		x	x						
Crotalus tigris	Tiger Rattlesnake	x	x		x		x							
Crotalus viridis	Prairie Rattlesnake	x	x		x		x							8
Crotalus willardi	Ridge-nosed Rattlesnake	x			x			x						
Diadophis punctatus	Ring-necked Snake	x		x	x		x					x		
Drymarchon melanurus	Central American Indigo Snake	x	x											
Drymobius margaritiferus	Speckled Racer	x	x											
Elaphe bairdi	Baird's Rat Snake	x	x											
Elaphe emoryi	Great Plains Rat Snake	x	x		x									
Elaphe guttata	Red Corn Snake	x	x				x							
Elaphe obsoletus	Texas Rat Snake	x	x		x									
Elaphe rosaliae	Baja California Rat Snake	x	x		x									
Elaphe subocularis	Trans-Pecos Rat Snake	x	x		x									
Elaphe triaspis	Green Rat Snake	x			x									
Ficimia streckeri	Tamaulipan Hook-nosed Snake	x	x											
Gyalopion canum	Chihuahuan Hook-nosed Snake	x	x		x		x	x						
Gyalopion quadrangulare	Thornscrub Hook-nosed Snake	x	x		x		x	x						
Heterodon kennerlyi	Mexican Hog-nosed Snake	x	x	x	x			x						
Heterodon nasicus	Plains Hog-nosed Snake	x	x	x	x		x	x						
Heterodon platirhinos	Eastern Hog-nosed Snake	x	x											

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Hypsiglena torquata	Coast Night Snake	x	x											
Hypsiglena torquata	Desert Night Snake	x			x		x	x						
Hypsiglena torquata	Chihuahuan Night Snake	x			x		x	x						
Lampropeltis alterna	Gray-banded Kingsnake	x	x		x									
Lampropeltis calligaster	Yellow-bellied Kingsnake	x	x											
Lampropeltis getula	Common Kingsnake	x	x		x									
Lampropeltis pyromelana	Sonoran Mountain Kingsnake	x	x		x			x						
Lampropeltis triangulum	Milk Snake	x	x	x	x			x						
Lampropeltis zonata	California Mountain Kingsnake	x			x			x						
Leptodeira septentrionalis	Cat-eyed Snake	x	x											
Leptotyphlops dissectus	New Mexico Thread Snake	x	x		x		x							
Leptotyphlops dulcis	Texas Thread Snake	x	x		x		x	x						
Leptotyphlops humilis	Western Thread Snake	x			x		x							
Masticophis bilineatus	Sonoran Whipsnake	x			x							x		
Masticophis flagellum	Coachwhip	x	x	x	x			x				x		
Masticophis fuliginosus	Baja California Coachwhip	x			x									
Masticophis lateralis	Striped Racer	x	x		x							x		
Masticophis schotti	Schott's Whipsnake	x	x											
Masticophis taeniatus	Striped Whipsnake	x	x	x	x									
Micruroides euryxanthus	Sonoran Coral Snake	x			x									
Micrurus tener	Texas Coral Snake	x	x											
Nerodia clarkii	Saltmarsh Watersnake	x	x		x									
Nerodia cyclopion	Mississippi Green Watersnake	x	x		x									
Nerodia erythrogaster	Plain-bellied Watersnake	x	x	x	x									
Nerodia fasciata	Southern Watersnake	x	x		x									
Nerodia harteri	Brazos River Watersnake	x	x		x									
Nerodia paucimaculata	Concho Watersnake	x	x		x									
Nerodia rhombifer	Diamond-backed Watersnake	x	x		x									
Nerodia sipedon	Northern Watersnake	x	x	x	x									
Opheodrys aestivus	Rough Green Snake	x	x											
Opheodrys vernalis	Smooth Green Snake	x	x		x									
Oxybelis aeneus	Brown Vine Snake	x	x		x		x	x						
Phyllorhynchus browni	Saddled Leaf-nosed Snake	x	x		x		x	x						
Phyllorhynchus decurtatus	Spotted Leaf-nosed Snake	x			x		x	x						
Pituophis catenifer	Gopher Snake	x	x		x		x	x						
Pituophis ruthveni	Louisiana Pine Snake	x	x											
Regina grahamii	Graham's Crayfish Snake	x	x		x									
Regina rigida	Glossy Crayfish Snake	x	x		x									
Rhinocheilus lecontei	Long-nosed Snake	x	x		x		x							
Salvadora grahamiae	Eastern Patch-nosed Snake	x	x		x		x	x			x			
Salvadora hexalepis	Western Patch-nosed Snake	x			x		x	x						
Sistrurus catenatus	Massasauga	x	x		x		x	x						
Sistrurus miliarius	Pygmy Rattlesnake	x	x											
Sonora semiannulata	Western Ground Snake	x	x		x									
Storeria dekayi	DeKay's Brown Snake	x	x											
Storeria occipitomaculata	Red-bellied Snake	x	x	x			x							
Tantilla atriceps	Mexican Black-headed Snake	x	x					x						
Tantilla cucullata	Trans-Pecos Black-headed Snake	x	x					x						
Tantilla gracilis	Flat-headed Snake	x	x		x			x						

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Tantilla hobartsmithi	Smith's Black-headed Snake	x			x		x							
Tantilla nigriceps	Plains Black-headed Snake	x	x		x		x							
Tantilla planiceps	Western Black-headed Snake	x	x	x	x		x	x						
Tantilla wilcoxi	Chihuahuan Black-headed Snake	x	x		x		x							
Tantilla yaquia	Yaqui Black-headed Snake	x			x		x							
Thamnophis atratus	Aquatic Garter Snake	x	x		x		x	x						
Thamnophis couchii	Sierra Garter Snake	x			x		x	x						
Thamnophis cyrtopsis	Black-necked Garter Snake	x	x		x		x							
Thamnophis elegans	Terrestrial Garter Snake	x	x	x	x			x						
Thamnophis eques	Mexican Garter Snake	x	x		x		x	x						
Thamnophis gigas	Giant Garter Snake	x	x		x			x						
Thamnophis hammondii	Two-striped Garter Snake	x	x		x									
Thamnophis marcianus	Checkered Garter Snake	x	x		x									
Thamnophis ordinoides	Northwestern Garter Snake	x	x	x	x			x						
Thamnophis proximus	Western Ribbon Snake	x	x		x			x						
Thamnophis radix	Plains Garter Snake	x	x		x			x						
Thamnophis rufipunctatus	Narrow-headed Garter Snake	x	x		x			x						
Thamnophis sirtalis	Common Garter Snake	x	x	x	x			x						
Trimorphodon biscutatus	Western Lyre Snake	x			x		x	x						
Trimorphodon vilkinsonii	Texas Lyre Snake	x	x		x		x	x						
Tropidoclonion lineatum	Lined Snake	x	x	x	x			x						
Virginia striatula	Rough Earth Snake	x	x		x									
Virginia valeriae	Smooth Earth Snake	x	x		x									
NORTHWEST REGION														
Charina bottae	Northern Rubber Boa	x		x				x				x		
Coluber constrictor	North American Racer	x	x	x	x									
Contia tenuis	Sharp-tailed Snake	x		x	x							x	x	
Crotalus oreganus	Western Rattlesnake	x			x		x	x					x	8
Crotalus viridis	Prairie Rattlesnake	x			x		x	x					x	8
Diadophis punctatus	Ring-necked Snake	x		x	x		x	x						
Heterodon nasicus	Plains Hog-nosed Snake	x			x		x	x						
Hypsiglena torquata	Desert Night Snake	x			x			x						
Lampropeltis getula	Common Kingsnake	x			x									
Lampropeltis pyromelana	Sonoran Mountain Kingsnake	x	x		x									
Lampropeltis triangulum	Milk Snake	x	x		x		x							
Lampropeltis zonata	California Mountain Kingsnake	x	x		x		x							
Masticophis lateralis	Striped Racer	x			x									
Masticophis taeniatus	Striped Whipsnake	x			x									
Nerodia fasciata	Southern Watersnake	x		x	x									
Nerodia rhombifer	Diamond-backed Watersnake	x		x	x									
Opheodrys vernalis	Smooth Green Snake	x		x	x									
Pituophis catenifer	Gopher Snake	x												
Rhinocheilus lecontei	Long-nosed Snake	x	x		x									
Salvadora hexalepis	Western Patch-nosed Snake	x			x		x	x						
Sonora semiannulata	Western Ground Snake	x	x		x									
Thamnophis atratus	Aquatic Garter Snake	x	x		x									
Thamnophis couchii	Sierra Garter Snake	x	x											

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Thamnophis elegans	Terrestrial Garter Snake	x	x		x									
Thamnophis gigas	Giant Garter Snake	x	x		x									
Thamnophis ordinoides	Northwestern Garter Snake	x	x		x									
Thamnophis radix	Plains Garter Snake	x	x		x									
Thamnophis sirtalis	Common Garter Snake	x	x		x									
MIDWEST REGION														
Agkistrodon contortrix	Copperhead	x	x				x	x				x		
Agkistrodon piscivorus	Cottonmouth	x	x				x	x				x		
Arizona elegans	Glossy Snake	x	x	x								x		
Carphophis amoenus	Eastern Worm Snake	x	x	x			x					x		
Carphophis vermis	Western Worm Snake	x	x	x	x		x					x		
Cemophora coccinea	Scarlet Snake	x	x	x			x	x				x		
Clonophis kirtlandii	Kirtland's Snake	x	x	x								x		
Coluber constrictor	North American Racer	x	x	x								x		
Crotalus horridus	Timber Rattlesnake	x	x				x	x				x	x	
Crotalus viridis	Prairie Rattlesnake	x	x				x	x				x		
Diadophis punctatus	Ring-necked Snake	x	x	x			x					x		
Elaphe emoryi	Great Plains Rat Snake	x	x	x			x					x		
Elaphe gloydi	Eastern Fox Snake	x	x	x								x		
Elaphe guttata	Red Corn Snake	x	x	x			x	x				x		
Elaphe obsoleta	Eastern Rat Snake	x	x	x			x	x			x	x		
Elaphe spiloides	Gray Rat Snake	x	x	x				x			x	x		
Elaphe vulpina	Western Fox Snake	x	x	x								x		
Farancia abacura	Red-bellied Mud Snake	x	x	x	x							x		
Heterodon gloydi	Dusty Hog-nosed Snake	x	x	x			x	x				x		
Heterodon nasicus	Plains Hog-nosed Snake	x	x	x			x	x				x		
Heterodon platirhinos	Eastern Hog-nosed Snake	x	x	x			x	x				x		
Hypsiglena torquata	Chihuahuan Night Snake	x	x	x			x	x				x		
Lampropeltis calligaster	Yellow-bellied Kingsnake	x	x	x			x					x		
Lampropeltis getula	Common Kingsnake	x	x	x								x		
Lampropeltis triangulum	Milk Snake	x	x	x			x					x		
Leptotyphlops dulcis	Texas Thread Snake	x	x	x								x		
Masticophis flagellum	Coachwhip	x	x	x								x	x	
Nerodia cyclopion	Mississippi Green Watersnake	x	x	x				x				x		
Nerodia erythrogaster	Plain-bellied Watersnake	x	x	x				x				x		
Nerodia fasciata	Southern Watersnake	x	x	x				x				x		
Nerodia rhombifer	Diamond-backed Watersnake	x	x	x				x				x		
Nerodia sipedon	Northern Watersnake	x	x	x				x				x		
Opheodrys aestivus	Rough Green Snake	x	x	x								x		
Opheodrys vernalis	Smooth Green Snake	x	x	x								x		
Pituophis catenifer	Gopher Snake	x	x	x			x	x				x		
Regina grahamii	Graham's Crayfish Snake	x	x	x								x		
Regina septemvittata	Queen Snake	x	x	x								x		
Rhinocheilus lecontei	Long-nosed Snake	x	x	x				x				x		
Sistrurus catenatus	Massasauga	x	x	x			x	x				x		
Sistrurus miliarius	Pygmy Rattlesnake	x	x	x			x	x				x		
Sonora semiannulata	Western Ground Snake	x	x	x			x	x				x		

Scientific Name	Common Name	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
Storeria dekayi	Dekay's Brown Snake	x	x	x			x	x				x		
Storeria occipitomaculata	Red-bellied Snake	x	x	x			x	x				x		
Tantilla coronata	Southeastern Crowned Snake	x	x	x			x	x				x		
Tantilla gracilis	Flat-headed Snake	x	x	x			x	x				x		
Tantilla nigriceps	Plains Black-headed Snake	x	x	x			x	x				x		
Thamnophis butleri	Butler's Garter Snake	x	x		x		x	x				x		
Thamnophis elegans	Terrestrial Garter Snake	x	x		x			x				x		
Thamnophis marcianus	Checkered Garter Snake	x	x		x			x				x		
Thamnophis proximus	Western Ribbon Snake	x	x	x	x		x	x				x		
Thamnophis radix	Plains Garter Snake	x	x		x		x	x				x		
Thamnophis sauritus	Eastern Ribbon Snake	x	x	x	x		x	x				x		
Thamnophis sirtalis	Common Garter Snake	x	x		x		x	x				x		
Tropidoclonion lineatum	Lined Snake	x	x	x	x		x	x				x		
Virginia striatula	Rough Earth Snake	x	x	x			x	x				x		
Virginia valeriae	Smooth Earth Snake	x	x	x			x	x				x		

CROCODILIANS														
Scientific Name	**Common Name**	whole body - dorsal	whole body - lateral	belly	whole body - dorso-lateral	back of thigh	head - dorsal	head - lateral	toe-webbing	toes	chin / throat	under-side of tail	side of tail	Comment code
SOUTHEAST REGION														
Alligator mississippiensis	American Alligator	x			x		x							
Crocodylus acutus	American Crocodile	x			x		x							
SOUTHWEST REGION														
Alligator mississippiensis	American Alligator	x			x		x							

Comments and their relevant codes for the *Photo Vouchering Table*:

Code	Comments
1	In areas where Cope's Gray and Gray Treefrogs geographically overlap, photographs are not sufficient to distinguish species
2	In areas where Streamside and Smallmouth Salamanders geographically overlap, photographs are not sufficient to distinguish species
3	*Desmognathus* spp. often difficult to distinguish; photo may not be sufficient
4	In dark color phase, photograph may not be sufficient
5	In areas of range overlap, species within the Slimy Salamander complex are often impossible to distinguish by photographs
6	In areas where Greater and Lesser Sirens geographically overlap, photographs are often insufficient to distinguish species
7	In areas where Mimic and Slender Glass Lizards geographically overlap, photographs are often insufficient to distinguish species
8	In areas where Prairie and Western Rattlesnakes geographically overlap, photographs are often insufficient to distinguish species
9	In areas where Northern and Southern Alligator Lizards geographically overlap, photographs are often insufficient to distinguish species without view of tail length vs. body length
10	In areas where Eastern Red-backed and either Big Levels or Shenandoah Salamanders geographically overlap, photographs are often insufficient to distinguish species
11	In areas where Western Red-backed and Dunn's Salamanders geographically overlap, photographs are often insufficient to distinguish species
12	In areas where Cope's and Coastal Giant Salamanders geographically overlap, photographs are often insufficient to distinguish species
13	In areas where any combinations of Northern Red-legged, California Red-legged, Cascades, and Oregon Spotted Frogs geographically overlap, photographs are often insufficient to distinguish species
14	In areas where Northern Pacific and Sierra Chorus Frogs geographically overlap, photographs are often insufficient to distinguish species
15	In areas where California Slender and Black-bellied Slender Salamanders overlap, photographs are not sufficient to distinguish species
16	In areas where Relictual Slender and Kern Canyon Slender Salamanders overlap, photographs are not sufficient to distinguish species
17	In areas where Fowler's and Woodhouse's Toads geographically overlap, photographs are not sufficient to distinguish species
18	In areas where Boreal and Western Chorus Frogs geographically overlap, photographs are not sufficient to distinguish species
19	In areas where any combinations of Eastern Red-backed, Southern Red-backed, Webster's, Ozark Zigzag, Southern Zigzag and Northern Zigzag Salamanders geographically overlap, photographs are often insufficient to distinguish species
20	In areas where any combinations of Western Slimy, Northern Slimy, Ravine, and Wehrle's Salamanders geographically overlap, photographs are often insufficient to distinguish species

APPENDIX VIII. SAMPLE DATASHEETS

Gabrielle J. Graeter

Here we present several example datasheets for inventory and monitoring projects. The information included on a datasheet will vary according to your objectives, so this list is only a sampling of the types of datasheets that could be created. Datasheets should be created so that the data collected are organized and standardized. See the section on "Standards and data management" in Chapter 4 for more information about creating datasheets.

Most datasheets will include the following basic information, in addition to other categories for more complex studies:

- Date
- Time of day
- Collector(s)
- Weather (e.g., air temperature, precipitation)
- Habitat sampled
- Habitat description
- Techniques used
- Species

INVENTORY DATASHEETS

Assessing the suitability of a habitat for certain species

This is the first step of an inventory and involves recording basic data that can be used to evaluate the quality of the habitat for a particular species. Data should include a clear identification of the location, a description of the habitat and natural community, a list of any disturbances and threats, the condition (e.g., size, health, reproduction) of the target species, and basic management and protection recommendations. This information will help in deciding whether to inventory or monitor in a particular area. (See page 278 for sample datasheet.)

More detailed assessment of the habitat and population status of a site

In addition to more thoroughly assessing the habitat of the site and the population status of the target species, the datasheet on page 279 could also be used as a draft "monitoring" datasheet, because successive visits to the site would collect the same data, and status of the site and species could be evaluated over time.

Site evaluation and species data

The datasheets on pages 280-282 are another example of datasheets used for assessing the habitat and inventorying the species located within that area.

Inventory at established sites, in which the habitat has already been assessed

The datasheets on pages 283 and 284 are from a herpetological survey of the National Parks in the southeastern United States. They are excellent examples of the types of datasheets you may want to design for surveying at sites where the areas to be inventoried have been established through habitat assessments.

Datasheet from an auditory survey program

The first datasheet on page 285 is an example of the type of information you would want to include on a datasheet for auditory surveys. The environmental conditions, such as temperature, wind, and the number of days since last rainfall, are key components of this type of survey. The most common format used for calling surveys is the one created by NAAMP (North American Amphibian Monitoring Program); the NAAMP auditory survey protocol is illustrated in this sample datasheet.

For recording data from a froglogger, a simple datasheet such as the second example on page 285 will suffice.

SAMPLE DATASHEET *for* BASIC INVENTORY

ELEMENT OCCURRENCE RECORD – *a species or a community type*

SCIENTIFIC NAME: _____
SITE NAME: _____
COUNTY: _____
QUADNAME: *the topo map name* QUADCODE: *GPS / location*
(LAT): _____ (N): _____ (S): _____
(LONG): _____ (W): _____ (E): _____
DIRECTIONS: *X.X miles from jct of Rd X and Rt Y or use GPS location*

PHYSPROV: *physiographic region* WATERSHED: *drainage basin*

SURVEY DATE: _____
LASTOBS: *last observation* FIRSTOBS: *first observation*

EO RANK: *ranking system to assess quality, condition, viability, & defensibility of a pop'n*
EO RANKCOM: *comments on rank*

EO DATA: *data on the species*
(pop'n size and structure, extent, reproduction, disease, predation; community condition, size, dominance, special fesatures)

GENDESC: *general description*
(associated species, habitat, landform, land use, threats, etc.)

MANAME: *if there is a management area, what is the name?*
MGMTCOM: *management comments*
PROTCOM: *protection comments*
OWNER: _____
OWNERCOM: *owner comments*
COMMENTS: *general comments*

BOUNDARIES: *is area defined?*
PHOTOS: *yes or no*
BESTSOURCE: *other existing data about the site*

SAMPLE DATASHEET *for* DETAILED INVENTORY

SUPPLEMENTAL FIELD DATA: RARE SPECIES OCCURRENCE

WEATHER: _____
SLOPE: _____ ASPECT: _____ ELEV.: _____ TOPO. POS.: _____
LIGHT: _____ MOISTURE: _____ GEOLOGY: _____
SOIL: _____

HABITAT DESCRIPTION: _____
describe the habitat (e.g., pine forest, hardwood forest, old field, etc)
_____
COMMUNITY COMMENTS: _____
applicable if site is a distinct entity, like an isolated wetland
_____
_____
DISTURBANCE: *for example: clearcut and site prepared, ditched wetland*
_____
THREATS: *for example: pollution observed, development nearby*
POPULATION DOCUMENTED VIA: Sight__ Tracks/sign__ Songs/calls__ Roadkill__ Photo__ Verbal__
ID PROBLEMS? (yes/no): _____ ID COMMENTS: _____
SPECIMEN NUMBER: *only if collected or vouchered* REPOSITORY: *name of museum*
NUMBERS OBSERVED: _____ POPULATION SIZE ESTIMATE: _____
ESTIMATED AMOUNT OF POTENTIAL HABITAT (acres): _____
PERCENT OF POTENTIAL HABITAT OCCUPIED: _____
POPULATION SIZE AND HABITAT AREA COMMENTS: _____
_____
REPRODUCTION: *any signs of reproduction? (e.g., nests or mating)*
DISEASE OR PREDATION: *any signs of disease or predation?*
PHENOLOGICAL CONDITION: *did the animals observed appear healthy?*
BEHAVIORAL NOTES: _____
_____
_____

EO RANKING CONSIDERATIONS: *assesses the condition of this population and habitat*

	RANK	COMMENT
QUALITY: (pop'n. size, productivity, vigor of individuals, etc.)	A B C D	*quality of the population*
CONDITION: (habitat pristine, recoverable, degraded, etc.)	A B C D	*health of the habitat*
VIABILITY (likelihood of long-term survival, based on intrinsic biological factors)	A B C D	*given the condition, threats, disturbance, will it persist?*
DEFENSIBILITY: (likelihood of long-term survival, based on intrinsic and extrinsic site factors)	A B C D	*can it be protected?*

NATIONAL WILDLIFE REFUGES
HABITAT MANAGEMENT RECOMMENDATIONS
SITE EVALUATION FORM

REFUGE_____ DATE_____

TIME _____ TO _____

PERSONNEL IN FIELD _____

REPORT FILED BY _____

SITE NAME_____

MANAGEMENT COMPARTMENT _____

GPS LOCATION: N: _____ W: _____

ELEVATION _____ (FEET)

MAP ATTACHED: _____ AREA BOUNDARIES IDENTIFIED _____

SITE LOCATION/DIRECTIONS_____

_____

TARGET SPECIES_____ **FOUND?** _____

HABITAT TYPE_____

SITE DESCRIPTION_____

HABITAT AREA/SIZE_____

DOMINANT VEGETATION_____

PLANT SPECIES LIST **ANIMAL SPECIES LIST**

Kurt Buhlmann

SITE EVALUATION FORM (PAGE 2)

HABITAT CONDITION_____

SITE QUALITY_____

THREATS TO HABITAT_____
(DIRECT IMPACTS, ADJACENT/LANDSCAPE MATRIX IMPACTS, DEGREE OF ISOLATION, EXOTIC SPECIES)

MANAGEMENT URGENCY _____

ADJACENT LANDOWNERS (OPPORTUNITIES FOR COOPERATIVE MGMT?)

_____

SUGGESTED MANAGEMENT

MANAGEMENT CATEGORIES
PROTECT, ENHANCE, RESTORE, CREATE, INTEGRATING MANAGEMENT

MANAGEMENT TO MINIMIZE IMPACTS TO HABITAT
_____

_____

MANAGEMENT TO BENEFIT HABITAT
_____
_____
_____

PHOTOGRAPHS TAKEN (List descriptions/digital picture number)

_____ _____

_____ _____
_____ _____
_____ _____

_____ _____

Kurt Buhlmann

Species and Species Specific Data

TARGET SPECIES_____

FOUND_____ SUSPECTED_____ ABSENT _____

POPULATION SIZE_____

POPULATION VIABILITY_____

Are all Habitat Components Present that are Known to be Used by the Target Species?

Number of Individuals of the Target Species Found

Marked, Measured, Sexed?

ID codes used

What techniques were used to assess population size—trapping, time-contrained, basking surveys, road-cruising.

Suitable for a monitoring project.

What data will need to be collected to start a monitoring evaluation of the management initiated??

Turtles Collected for Health Assessment?

Kurt Buhlmann

Note: All fields are required to be filled in (using pencil or <u>permanent</u> ink) Page_____ of _____

Herpetological Inventory

Date (day-month-yr; e.g, 5-May-07)_____ Start time_____ End time_____

Observers: TDT JWG MED JDW PAM _____

Park _____ State_____ County_____ Zone (from GPS unit) _____

Locality_____

Temp (C)_____ Cloud cover (%)_____ Wind: 0 1 2 3 Rain: 0 1 2 3 Rain last 24hr: Yes No

Habitat _____

Comments (habitat quality, threats, etc) _____

_____

Rec #	Sp. Code	No.	Tech	Photo?	Specimen?	UTM-N	UTM-E	Comments (incl. general locality)

Tracey Tuberville

Instruction for NPS Southeast Coastal Network Herpetological Inventory Datasheet

Date **–** formatted as day-mo-year (e.g., 5-May-02)

Time - 24 hr time

UTM - make sure GPS is set to WGS 84

Zone - recorded from GPS unit (e.g., 16S)

Locality - Be specific, use local names, direction and distance from intersections on roads or trails, avoid use of "from", "near", "close to", etc.

Park Abbreviations

CAHA - Cape Hatteras National Seashore
CALO - Cape Lookout Natipnal Seashore
CASA - Castillo de San Marcos
FOMA - Fort Matanzas National Monument
CHAT - Chattahoochee River National Recreation Area
COSW - Congaree Swamp National Monument
CUIS - Cumberland Island National Seashore
FOFR - Fort Frederica National Monument
FOPU - Fort Pulaski National Monument
FOSU - Fort Sumter National Monument
CHPI - Charles Pinckney National Historic Site
MOCR - Moore's Creek National Battlefield
HOBE - Horseshoe Bend National Military Park
KEMO - Kennesaw Mountain National Battlefield Park
OCMU - Ocmulgee National Monument
TIMU - Timucuan Ecological And Historical Preserve

Cloud Cover - percent of sky covered by clouds

Wind - 0 = none; 1 = breeze; 2 = gusts; 3 = steady wind

Rain - 0 = none; 1 = sprinkle; 2 = steady; 3 = downpour

Rain last 24 hrs - if amount known, record in comments

Habitat - examples - Hardwood, mixed, pine, shrub, meadow, landscaped, temp wetland, permanent wetland, stream, saltmarsh.

Record Number - DO NOT USE IN FIELD

Species Code - First three letters of genus and first three letters of species.

Number of Individuals - estimate for large numbers (e.g., frog choruses).

Technique Codes

OBS - Observation
H - Hand Capture
DN - Dipnet
MT - Minnow Trap
TT - Turtle Trap
SN - Seine
CB - Artificial Cover Object
CL - Call
DOR - Dead On Road
AOR - Live On Road

Photo? - Y or N – When turning in digital or 35mm photographs to data manager, make sure that photos for different parks on different dates are kept separately. Each photo should be labeled with species name, park name, date.

Specimen? – Y or N – Indicate whether live or dead in comment field. Make sure specimens clearly and securely labeled.

Tracey Tuberville

Sample Datasheet *for* an Auditory Survey (using NAAMP protocol)

Route Number: 630102 **Route Name:** Blowing Rock **State/Region:** North Carolina	**Run Number:** 3 **Date of Visit:** 5/19/2006
Start Time: 21:25 (05/19/2006)	**End Time:** 23:47 (05/19/2006)
Start Wind: 2	**End Wind:** 1
Start Sky: 2	**End Sky:** 2
Days Since Rain: 0	

Stop →	**1**	**2**	**3**	**4**	**5**	**6**	**7**	**8**	**9**	**10**
Start Time:	21:25	21:42	22:10	22:31	22:42	22:50	23:12	23:23	23:33	23:42
Air Temperature (°C):	17.1	16.4	15.9	16.1	15.9	16.0	15.8	15.6	15.6	16.1
Noise Disturbance:	2	0	0	0	0	0	0	1	0	0
Time Out:										
Skipped Stop:										
Species ↓ Stop →	**1**	**2**	**3**	**4**	**5**	**6**	**7**	**8**	**9**	**10**
American toad (*Bufo americanus*)					1					1
Cope's gray treefrog (*Hyla chrysoscelis*)				1		1	1			
Spring peeper (*Pseudacris crucifer*)	1		3	2	2	3	3	2		
American bullfrog (*Rana catesbeiana*)					2	1	2			
Green frog (*Rana clamitans*)				1			1			
Stop →	**1**	**2**	**3**	**4**	**5**	**6**	**7**	**8**	**9**	**10**
Moon:	N	N	N	N	N	N	N	N	N	N
Snow Cover:	N	N	N	N	N	N	N	N	N	N
No. Cars:	3	0	0	0	0	0	0	1	0	0

USGS Patuxent, NAAMP

SAMPLE DATASHEET *for* AUTOMATED RECORDING DEVICES (i.e., FROGLOGGERS)

Date	Time	Species	Comment

Mike Dorcas

MONITORING DATASHEETS

Monitoring a turtle population

The datasheet below is used for monitoring aquatic turtle populations at multiple sites. This datasheet requires the species, location, name of the person that captured and processed the turtle, date, identification code (i.e., mark), sex, various measurements [carapace length (CL), plastron length (PL), shell height, and mass (weight in grams), etc], number of annuli, whether the turtle is gravid, and provides an area for comments. Other data to consider including on this type of datasheet are the recapture status and the method of capture.

Monitoring a drift fence with pitfall traps encircling one wetland

A drift fence design with only one site makes for a simple datasheet. In designing a datasheet for monitoring a drift fence, the basic data should be recorded, such as environmental conditions, the date, and data collectors, as well as the trap number, species, sex, and identification code for captured animals. (See page 287 for sample datasheet.)

Monitoring a drift fence system with a complex design

Designing a datasheet for a drift fence system at multiple locations and habitat types (e.g., wetland margins, terrestrial areas) and multiple target species takes careful consideration. The datasheet on page 288 provides an excellent example of the type of data you may want to record for this type of program. Beyond the typical environmental data, the recapture status, fate, sex, age, and snout-vent length may need to be recorded. Each of these columns should have pre-established response options (see bottom of datasheet) so that the data entered into the database are clear and consistent.

SPECIES _____ LOCATION _____ PROCESSED BY: KAB TDT CH JLG ___ DATE (ex: 01/JAN/2010) _____

TURTLE RESEARCH - SAVANNAH RIVER ECOLOGY LAB																
ID	SEX	CL MAX	CL MID	PL MAX	PL MID	PL ANT MID	PL POST MID	SHELL HEIGHT	HEAD WIDTH	WEIGHT (g)	PRE-ANAL DIST	HEALTH ISSUE	PLASTRAL PATTERN	# ANNULI	GRAVID ?	COMMENTS

ALL INDIVIDUALS IN GROUP ACCOUNTED FOR? YES / NO DATA ENTERED IN COMPUTER?

Kurt Buhlmann

Datasheet used for long-term monitoring study with turtles on Savannah River Site. Includes species and ID, sex, size, mass, location, collector, and notes.

Ellenton Bay Data - 2003

put date on each page PAGE ____ of ____

(Circle one)

NEW Collector(s): _____

Date: _____ Snake Trap: Set Removed Amb: _____

Time begin (24hr.): _____ Time end _____ Temp: Min: _____

Rainfall (mm): _____ Max: _____

CIRCLE: BJW BDT BSM CAY CTW DAC JLG JLN JPN

Water Level (cm): _____ Weather _____

JWG KAB KMA LAF LRW RNT TDT TMM TBA XAG

#D = # dead / RTL = returned to lab # = # individuals

Trap Type	Trap #	Species	#	Meta	#D'	RTL	COMMENTS (TURTLES: ID, SEX,	Trap Type	Trap #	Species	#	Meta	#D'	RTL	COMMENTS (TURTLES: ID, SEX, REC):

Comments:

Recorded by: _____

Judy Greene

Datasheet from a drift fence study encircling a large Carolina bay wetland in South Carolina. Includes basic environmental data, collector, trap type and number, and the species and status of species captured.

LEAP Bays 2004-05 Year 2 Page ____ of ____

Bay: _____

Date: _____ Time begin: _____ Time end: _____ Precip (mm): _____ Temp (C) Min: _____

Observer(s): _____ Water depth (cm): _____ Max: _____

Species	ID	Recap	Sex/ Age	SVL (mm)	Trap #	Fate	Notes	Species	ID	Recap	Sex/ Age	SVL (mm)	Trap #	Fate	Notes

Species (4-letter code); ID (cohort #); Recap (write year if recap, leave blank if new); Fate (D = dead, RTL = returned to lab)
Sex/Age (F = female, M = male, UA = adult, sex unknown; UI = immature, sex unknown; J = juvenile/metamorph from this bay)
Notes: H = hand capture, P = predated, D = drowned, C = desiccated, ID? = for recaps if ID is questionable

Betsie Rothermel

Datasheet from a drift fence study in terrestrial habitat surrounding four isolated seasonal wetlands. Environmental data was recorded, as well as the species, the cohort mark used, their sex and age, SVL, and their fate.

DIGITAL DATASHEETS

Many researchers are beginning to use a Personal Digital Assistant (PDA; e.g., palm pilot) for their data entry, especially for more complex monitoring programs. There are advantages and disadvantages to eliminating the paper version of a datasheet. A major advantage of using a PDA is that data can be downloaded directly to a database (e.g., Microsoft Access), thereby eliminating the data entry process. However, data can be lost more easily if data are not properly backed up and managed in their electronic form. Although the software programs available for creating an electronic datasheet (e.g., pendragon) seem to work well, some people find writing the computer code challenging and an inefficient use of time. See the section on "Standards and data management" in Chapter 4 for more information about PDAs and data management.

REFERENCES FOR ADDITIONAL SAMPLE DATASHEETS

Dodd, C. K., Jr. 2003. Monitoring amphibians in Great Smoky Mountains National Park. Circular 1258, US Geological Survey, Tallahassee, Florida.

Heyer, W. R., M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, M. S. Foster, editors. 1994. Measuring and monitoring biological diversity: standard methods for amphibians. Smithsonian Institution Press, Washington, DC, USA.

Mitchell, J. C. 2000. Amphibian Monitoring Methods & Field Guide. Smithsonian National Zoological Park, Conservation Research Center, Front Royal, VA. 56pp.

Somers, A. B., and C. E. Matthews. 2006. The Box Turtle Connection: A Passageway into the Natural World. 127pp. http://www.uncg.edu/~absomers/BoxTurtleBook.pdf

APPENDIX IX. EQUIPMENT LISTS

Gabrielle J. Graeter

Here we provide several lists of the equipment needed for sampling amphibians and reptiles, including basic equipment lists and an example of what a detailed and comprehensive equipment list should look like. The basic equipment lists include items needed in many basic inventory or monitoring programs, but very few studies will require the use of all items in these lists. The sample comprehensive equipment list is shown to demonstrate the amount of thought and planning that must go into a study to ensure success. Please see the technique-specific equipment lists in Chapter 5.

BASIC EQUIPMENT LIST

Paper and data related materials

1. Collecting permits
2. Clipboard and datasheets (consider waterproof Rite-in-the-Rain paper) or a PDA (personal data assistant)
3. Pencil/pen and permanent marker
4. Field notebook
5. Field guide
6. Study protocol notes (e.g., toe-clipping scheme for frogs)

Navigating tools

1. GPS unit
2. Compass
3. Maps of study area

Measuring equipment

1. Metric rulers and tape measures
2. Pesola scales
3. Thermometers (e.g., air, substrate)
4. Psychrometer
5. Rain gauge
6. Barometer, clinometer, soil moisture meter, anemometer, wind recorder, etc
7. Water depth gauge (for water bodies)
8. Dissolved oxygen, pH, conductivity meters (for aquatic sampling)
9. Hand lens
10. Frogloggers or cassette recorders (for auditory surveys)

Collecting equipment

1. Plastic bags and/or containers
2. Dip nets (size depends on study)
3. Snake stick (i.e., stump ripper)

Safety equipment (to protect yourself and to prevent the spread of pathogens between sites)
1. Plastic gloves
2. Hand sanitizer
3. Bleach solution and tub to rinse boots and sampling equipment between ponds
4. First Aid kit

General field gear
1. Camera
2. Pocket knife with tools
3. Flashlight/headlamp for night or cave work
4. Binoculars
5. Appropriate field attire (e.g., boots, waders, hat, raingear, sunglasses)
6. Water and food for long-term field work
7. Sunblock and bug repellent/netting
8. Watch

Miscellaneous items
1. Extra batteries for electronic equipment
2. Flagging or stake flags
3. Tools for installing a sampling method (e.g., shovel, hammer, zip-ties, etc. for installing drift fences with pitfall traps)

SAMPLE EQUIPMENT LISTS

The purpose of including these sample equipment lists is to demonstrate the amount of thought and detail that goes into planning an inventory or monitoring project.

Equipment needed for surveys at National Wildlife Refuges (contributed by Kurt Buhlmann)

General equipment
1. Field guides
2. Aluminum clipboards
3. Waterproof notebooks and paper
4. Permanent ink pens for filling out datasheets and waterproof notebooks
5. Blank data sheets (including turtle datasheet)
6. Maps (for refuge, state maps, gazetteers)
7. Collecting permits (state) and refuge use permits
8. Refuge key
9. Contact info for each appropriate refuge
10. GPS units (2)
11. Batteries for GPS units (4 “AA”)
12. Calipers (2 plastic calipers, dial calipers, large tree calipers)
13. Scales (50g[x2], 100g, 2kg, 5kg, flat scale)
14. Max-Min thermometer
15. Miscellaneous tools (wrenches, vise grips, screwdrivers)
16. Leatherman pocket knife
17. Duct tape
18. Plastic bags
19. Digital camera (with batteries, charger, and memory cards)
20. Binoculars
21. Snake sticks
22. Spotting scope with Tripod (and camera adaptor)
23. Weather radio

Aquatic equipment
1. Nets (wide-mouth and fyke nets)
2. Mesh turtle bags
3. Turtle bins
4. Buckets
5. Hoop traps and poles
6. Sardines
7. Crawfish bait
8. Plastic containers for crawfish bait
9. Collapsible Australia traps
10. Flagging to locate traps
11. Marking file
12. Marking diagram
13. Waders
14. Head Lamps and flashlights

Tissue recovery
1. Vials
2. Buffer
3. Ethanol
4. Swab containers

Boating and snorkeling
1. Canoe and paddles
2. Life jackets (for total number people on trip)
3. Chain for canoe
4. Tie-down straps for canoe
5. John boat on Trailer
6. Gas and oil for boat
7. Rope
8. Qbeam lights and Car battery (for rivers at night)
9. Mask and Snorkels
10. Flippers

Turtle (Macrochelys) trapping materials (contributed by Jim Godwin)
1. 14 to 17’ jon boat with 25 hp motor, trailer, paddles, life vests (boat packages are available which include boat, motor, trailer and perhaps other items)
2. Hoop nets + rope, weights
3. Bait bottles: ½ to 1 liter bottles
4. Fish: heads, guts, etc. available from processing houses, or catch your own
5. Knives: to cut bait
6. Twine: for net repair and securing bait bottles in traps
7. Pesola scales (kg): 5 or 10 kg and 50 kg to cover the range of weights

8. Calipers (cm): Haglof 80 cm aluminum, or perhaps 100 cm calipers if you expect really big turtles
9. Clipboard, pencils, and waterproof paper datasheets
10. Webbing + carabiners: to construct a sling while weighing turtles. A bandana works for small ones. Sheep harness is available commercially.
11. GPS unit: for storing waypoints and tracking back to waypoints (i.e., trap sites).
12. Genetic sampling equipment
13. Dremel with drill bit: portable battery operated model. It should come with a 1/8" drill bit which is used to drill a hole in the marginal for insertion of a stainless steel screw.
14. Container of alcohol: drill bit is immersed immediately before and after the drilling of a hole in a shell
15. Stainless steel screws: purchase locally at a hardware or nut and bolt store. If you can locate a nut and bolt store the price on the screws will be substantially lower. Stainless steel is necessary for corrosion resistance. Screw size needed if using the 1/8" drill bit is #10 x ½ Phillips pan head (a rounded head).
16. Screwdriver: find one that firmly fits the screw head.
17. Digital camera
18. Cooler for food, drink, and bait
19. Metric tape measure (mm): 5 m tape

PUBLICATIONS WITH ADDITIONAL EQUIPMENT AND/OR VENDOR LISTS

Dodd, C. K., Jr. 2003. Monitoring amphibians in Great Smoky Mountains National Park. Circular 1258, US Geological Survey, Tallahassee, Florida.

Heyer, W. R., M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, M. S. Foster, editors. 1994. Measuring and monitoring biological diversity: Standard methods for amphibians. Smithsonian Institution Press, Washington, DC, USA.

Mitchell, J. C. 2000. Amphibian Monitoring Methods & Field Guide. Smithsonian National Zoological Park, Conservation Research Center, Front Royal, VA. 56pp.

APPENDIX X. CONSERVATION STATUS OF AMPHIBIANS AND REPTILES

Gabrielle J. Graeter and W. Jeffrey Humphries

The current status of the amphibian and reptile species that are federally listed under the Endangered Species Act are shown in Table 15-1. As the biological and legal status of amphibians and reptiles in the United States is constantly shifting and changing, we suggest taking the following steps to determine the current status of a particular species:

1. Consult recent publications in trusted sources (e.g., peer-reviewed journals)

2. Consult reputable well-respected internet sources that are updated regularly with this sort of information (see below)

3. Confirm your understanding of the status of a species with a local herpetological expert

ONLINE SOURCES FOR CURRENT BIOLOGICAL AND LEGAL STATUS

- The US Fish & Wildlife Service's information about threatened and endangered animals and plants (some lists are updated daily): www.fws.gov/Endangered

- State agencies (for e.g., Georgia Department of Natural Resources: www.gadnr.org; Oregon Department of Fish and Wildlife: www.dfw.state.or.us)

- The United States Geological Survey (USGS) Amphibian Research and Monitoring Initiative (ARMI): armi.usgs.gov

- The USGS Southeast ARMI: cars.er.usgs.gov/herps

- Amphibia Web: amphibiaweb.org

TABLE 15-1: FEDERALLY THREATENED AND ENDANGERED SPECIES AS OF JULY 2010.

Common Name	Scientific Name	Status	Where Listed
REPTILES			
American alligator	*Alligator mississippiensis*	T(SA)	-
Culebra Island giant anole	*Anolis roosevelti*	E	-
St. Croix ground lizard	*Ameiva polops*	E	-
Loggerhead sea turtle	*Caretta caretta*	T	-
Green sea turtle	*Chelonia mydas*	E	Breeding colony populations in FL and on Pacific coast of Mexico
Green sea turtle	*Chelonia mydas*	T	Wherever found except where listed as endangered
American crocodile	*Crocodylus acutus*	T	FL population
New Mexican ridge-nosed rattlesnake	*Crotalus willardi obscurus*	T	-
Mona ground iguana	*Cyclura cornuta stejnegeri*	T	-
Leatherback sea turtle	*Dermochelys coriacea*	E	-
Eastern indigo snake	*Drymarchon corais couperi*	T	-
Mona boa	*Epicrates monensis monensis*	T	-
Puerto Rican boa	*Epicrates inornatus*	E	-
Virgin Islands tree boa	*Epicrates monensis granti*	E	-
Hawksbill sea turtle	*Eretmochyls imbricata*	E	-
Bluetail mole skink	*Eumeces egregious lividus*	T	-
Blunt-nosed leopard lizard	*Gambelia silus*	E	-
Bog turtle	*Glyptemys muhlenbergii*	T	Entire, except GA, NC, SC, TN, VA
Bog turtle	*Glyptemys muhlenbergii*	T(SA)	GA, NC, SC, TN, VA
Desert tortoise	*Gopherus agassizii*	T(SA)	AZ south and east of Colorado River, and Mexico, when found outside of Mexico or said range in AZ
Desert tortoise	*Gopherus agassizii*	T	Entire, except AZ south and east of Colorado River, and Mexico
Gopher tortoise	*Gopherus polyphemus*	T	Wherever found west of Mobile and Tombigbee Rivers in AL, MS, and LA
Yellow-blotched map turtle	*Graptemys flavimaculata*	T	-
Ringed map turtle	*Graptemys oculifera*	T	-
Kemp's ridley sea turtle	*Lepidochelys kempii*	E	-
Olive ridley sea turtle	*Lepidochelys olivacea*	T	Wherever found except where listed as endangered
Alameda whipsnake (=striped racer)	*Masticophis lateralis euryxanthus*	T	-
Sand skink	*Neoseps reynoldsi*	T	-
Atlantic salt marsh snake	*Nerodia clarkia taeniata*	T	-
Copperbelly water snake	*Nerodia erythrogaster neglecta*	T	IN north of 400 N. Latitude; MI; OH
Concho water snake	*Nerodia paucimaculata*	T	-
Lake Erie water snake	*Nerodia sipedon insularum*	T	Lake Erie offshore islands and their adjacent waters (located more than 1 mile from mainland)
Alabama red-belly turtle	*Pseudemys alabamensis*	E	-

Plymouth red-bellied turtle	*Pseudemys rubriventris bangsi*	E	-
Monito gecko	*Sphaerodactylus micropithecus*	E	-
Flattened musk turtle	*Sternotherus depressus*	T	Black Warrior River system upstream from Bankhead Dam
Giant garter snake	*Thamnophis gigas*	T	-
San Francisco garter snake	*Thamnophis sirtalis tetrataenia*	E	-
Coachella Valley fringe-toed lizard	*Uma inornata*	T	-
Island night lizard	*Xantusia riversiana*	T	-
AMPHIBIANS			
Reticulated flatwoods salamander	*Ambystoma bishopi*	E	-
California tiger salamander	*Ambystoma californiense*	E	Santa Barbara and Sonoma Counties, CA
California tiger salamander	*Ambystoma californiense*	T	Central CA DPS, not including Santa Barbara and Sonoma DPS
Frosted flatwoods salamander	*Ambystoma cingulatum*	T	-
Santa Cruz long-toed salamander	*Ambystoma macrodactylum croceum*	E	-
Sonora tiger salamander	*Ambystoma tigrinum stebbinsi*	E	-
Desert slender salamander	*Batrachoseps aridus*	E	-
Wyoming toad	*Bufo baxteri (=hemiophrys)*	E	-
Arroyo toad (=arroyo southwestern)	*Bufo californicus (=microscaphus)*	E	-
Houston toad	*Bufo houstonensis*	E	-
Golden coqui	*Eleutherodactylus jasperi*	T	-
Guajon	*Eleutherodactylus cooki*	T	-
San Marcos salamander	*Eurycea nana*	T	-
Barton Springs salamander	*Eurycea sosorum*	E	-
Puerto Rican crested toad	*Peltophryne lemur*	T	-
Red Hills salamander	*Phaeognathus hubrichti*	T	-
Cheat Mountain salamander	*Plethodon nettingi*	T	-
Shenandoah salamander	*Plethodon shenandoah*	E	-
California red-legged frog	*Rana draytonii*	T	Entire
Mississippi gopher frog	*Rana sevosa*	E	Wherever found west of Mobile and Tombigbee Rivers in AL, MS, and LA
Chiricahua leopard frog	*Rana chiricahuensis*	T	-
Mountain yellow-legged frog	*Rana muscosa*	E	Southern California DPS
Texas blind salamander	*Typhlomolge rathbuni*	E	-

T = Threatened; E = Endangered; T(SA) = Threatened because of similarity of appearance; DPS = Distinct population segment; In the "where listed" category, "-" indicates that the entire U.S. range is listed.

APPENDIX XI. SOURCES FOR FURTHER INFORMATION

Gabrielle J. Graeter and W. Jeffrey Humphries

The purpose of this appendix is to provide the user with contact information for Partners in Amphibian and Reptile Conservation (PARC), federal and state agencies, museums and collections, and university programs (SREL, etc.) to help in obtaining resources and information on permitting, the legal status of species, information about a sampling technique or study species, as well as a potential contact for sharing information and future collaboration.

Partners in Amphibian and Reptile Conservation (PARC).....www.parcplace.org

UNITED STATES FEDERAL AGENCIES:

US Forest Service .....www.fs.fed.us

US Fish and Wildlife Service..... www.fws.gov

US Bureau of Land Management.....www.blm.gov

US Geological Survey ..... www.usgs.gov

U.S. Department of Defense .....www.defense.gov

STATE AGENCIES:

Alabama

Dept. of Conservation and Natural Resources .....www.outdooralabama.com

Forestry Commission .....www.forestry.state.al.us

Alaska

Dept. of Fish and Game..... www.adfg.alaska.gov

Dept. of Environmental Conservation ..... www.dec.state.ak.us

Dept. of Natural Resources..... www.dnr.alaska.gov

Arizona

Game and Fish..... www.azgfd.gov

Dept. of Environmental Quality .....www.azdeq.gov

State Land Dept ..... www.land.state.az.us

Arkansas

Game and Fish Commission.....www.agfc.com

Dept. of Environmental Quality .....www.adeq.state.ar.us

Forestry Commission ..... www.forestry.arkansas.gov

California

Dept. of Fish and Game.....www.dfg.ca.gov

Natural Resources Agency.....resources.ca.gov

Dept. of Forestry and Fire Prevention.....www.fire.ca.gov

Colorado

Dept. of Natural Resources.....dnr.state.co.us

Division of Wildlife ..... wildlife.state.co.us

Connecticut

Dept. of Environmental Protection ..... www.ct.gov/dep

Delaware

Dept. of Natural Resources and Environmental Control ..... www.dnrec.delaware.gov

Division of Fish and Wildlife ..... www.dnrec.delaware.gov/fw

Florida

Dept. of Environmental Protection ..... www.dep.state.fl.us

Fish and Wildlife Conservation Commission ..... myfwc.com

Forestry Service ..... www.fl-dof.com

Georgia

Department of Natural Resources ..... www.gadnr.org

Wildlife Resources Division ..... georgiawildlife.com

Forestry Commission ..... www.gfc.state.ga.us

Hawaii

Dept. of Land and Natural Resources ..... www.hawaii.gov/dlnr

Division of Forestry and Wildlife ..... www.hawaii.gov/dlnr/dofaw

Division of Aquatic Resources ..... www.hawaii.gov/dlnr/dar

Idaho

Dept. of Fish and Game ..... http://fishandgame.idaho.gov

Dept. of Environmental Quality ..... www.deq.idaho.gov

Dept. of Lands ..... www.idl.idaho.gov

Illinois

Dept. of Natural Resources ..... www.dnr.illinois.gov

Environmental Protection Agency ..... www.epa.state.il.us

Indiana

Dept. of Natural Resources ..... www.in.gov/dnr

Division of Fish and Wildlife ..... www.in.gov/dnr/fishwild

Natural Resources Commission ..... www.in.gov/nrc

Iowa

Dept. of Natural Resources ..... www.iowadnr.gov

Kansas

Dept. of Wildlife, Parks, and Tourism ..... www.kdwp.state.ks.us

Dept. of Health and Environment ..... www.kdheks.gov

State Conservation Commission ..... scc.ks.gov

Kentucky

Dept. of Fish and Wildlife Resources ..... http://fw.ky.gov

Dept. for Environmental Protection ..... www.dep.ky.gov

Division of Forestry ..... www.forestry.ky.gov

Louisiana

Dept. of Wildlife and Fisheries ..... www.wlf.louisiana.gov
Dept. of Natural Resources ..... http://dnr.louisiana.gov
Dept. of Environmental Quality ..... www.deq.louisiana.gov
Dept. of Agriculture and Forestry ..... www.ldaf.state.la.us

Maine

Dept. of Inland Fisheries and Wildlife ..... www.state.me.us/ifw
Dept. of Environmental Protection ..... www.maine.gov/dep
Dept. of Conservation ..... www.maine.gov/doc

Maryland

Dept. of Natural Resources ..... www.dnr.state.md.us
Dept. of the Environment ..... www.mde.state.md.us

Massachusetts

Dept. of Fish and Game ..... www.mass.gov/dfwele
Dept. of Environmental Protection ..... www.mass.gov/dep
Dept. of Conservation and Recreation ..... www.mass.gov/dcr

Michigan

Dept. of Environmental Quality ..... www.michigan.gov/deq
Dept. of Natural Resources ..... www.michigan.gov/dnr

Minnesota

Dept. of Natural Resources ..... www.dnr.state.mn.us
Division of Fish and Wildlife ..... www.dnr.state.mn.us/fishwildlife

Mississippi

Dept. of Wildlife, Fisheries, and Parks ..... www.mdwfp.com
Dept. of Environmental Quality ..... www.deq.state.ms.us
Forestry Commission ..... www.mfc.ms.gov

Missouri

Dept. of Natural Resources ..... www.dnr.mo.gov
Dept. of Conservation ..... www.mdc.mo.gov

Montana

Fish, Wildlife, and Parks ..... fwp.mt.gov
Dept. of Environmental Quality ..... www.deq.mt.gov.us
Dept. of Natural Resources and Conservation ..... http://dnrc.mt.gov

Nebraska

Dept. of Natural Resources ..... www.dnr.ne.gov
Dept. of Environmental Quality ..... www.deq.state.ne.us
Game and Parks Commission ..... www.outdoornebraska.ne.gov

Nevada

Dept. of Conservation and Natural Resources ..... http://dcnr.nv.gov
Division of Environmental Protection ..... http://ndep.nv.gov
Dept. of Wildlife ..... www.ndow.org

New Hampshire

Fish and Game Dept. ..... www.wildlife.state.nh.us
Dept. of Environmental Services ..... www.des.state.nh.us
Dept. of Resources and Economic Development ..... www.dred.state.nh.us

New Jersey

Dept. of Environmental Protection ..... www.state.nj.us/dep
Division of Fish and Wildlife ..... www.state.nj.us/dep/fgw

New Mexico

Energy, Minerals, and Natural Resources Dept. ..... www.emnrd.state.nm.us
Environment Dept. ..... www.nmenv.state.nm.us
Dept. of Game and Fish ..... www.wildlife.state.nm.us

New York

Dept. of Environmental Conservation ..... www.dec.ny.gov

North Carolina

Dept. of Environment and Natural Resources ..... www.ncdenr.gov
Wildlife Resources Commission ..... www.ncwildlife.org
Forest Service ..... www.ncforestservice.gov

North Dakota

Game and Fish Dept. ..... http://gf.nd.gov

Ohio

Dept. of Natural Resources ..... www.dnr.state.oh.us
Division of Natural Areas and Preserves ..... www.ohiodnr.com/dnap
Division of Wildlife ..... www.dnr.state.oh.us/wildlife

Oklahoma

Dept. of Environmental Quality ..... www.deq.state.ok.us
Dept. of Wildlife Conservation ..... www.wildlifedepartment.com

Oregon

Dept. of Environmental Quality ..... www.oregon.gov/deq
Dept. of Fish and Wildlife ..... www.dfw.state.or.us

Pennsylvania

Dept. of Conservation and Natural Resources ..... www.dcnr.state.pa.us
Dept. of Environmental Protection ..... www.depweb.state.pa.us
Game Commission ..... www.pgc.state.pa.us

Rhode Island

Dept. of Environmental Management.....www.dem.ri.gov

Division of Fish and Wildlife .....www.dem.ri.gov/programs/bnatres/fishwild

South Carolina

Dept. of Natural Resources.....www.dnr.sc.gov

Forestry Commission .....www.state.sc.us/forest

Dept. of Health and Environmental Control .....www.scdhec.gov

South Dakota

Dept. of Environment and Natural Resources ..... www.denr.sd.gov

Game, Fish, and Parks .....www.gfp.sd.gov

Tennessee

Wildlife Resources Agency.....www.state.tn.us/twra

Division of Forestry.....www.tn.gov/agriculture/forestry

Dept. of Environment and Conservation ..... www.state.tn.us/environment

Texas

Commission on Environmental Quality .....www.tceq.state.tx.us

Parks and Wildlife Dept. .....www.tpwd.state.tx.us

Utah

Dept. of Environmental Quality ..... www.deq.utah.gov

Dept. of Natural Resources.....www.naturalresources.utah.gov

Division of Wildlife Resources.....http://wildlife.utah.gov

Vermont

Agency of Natural Resources .....www.anr.state.vt.us

Dept. of Environmental Conservation ..... www.anr.state.vt.us/dec

Fish and Wildlife Dept. .....www.vtfishandwildlife.com

Dept. of Forests, Parks, and Recreation.....www.vtfpr.org

Virginia

Dept. of Conservation and Recreation.....www.dcr.virginia.gov

Dept. of Environmental Quality ..... www.deq.state.va.us

Dept. of Game and Inland Fisheries .....www.dgif.virginia.gov

Marine Resources Commission ..... www.mrc.state.va.us

Washington

Conservation Commission ..... www.scc.wa.gov

Dept. of Fish and Wildlife ..... http://wdfw.wa.gov

West Virginia

Dept. of Environmental Protection .....www.dep.wv.gov

Division of Natural Resources.....www.wvdnr.gov

Wisconsin

Dept. of Natural Resources ..... www.dnr.state.wi.us

Wyoming

Dept. of Environmental Quality ..... http://deq.state.wy.us

Game and Fish Dept. ..... http://gf.state.wy.us

EXAMPLES* OF ADDITIONAL SOURCES OF INFORMATION:

*Note: the following lists serve as examples only and are not comprehensive in scope

University-based Herpetology Programs

Harvard University ..... www.mcz.harvard.edu/Departments/Herpetology

Savannah River Ecology Laboratory (SREL) ..... www.uga.edu/srelherp

Davidson College ..... www.bio.davidson.edu/dorcas/midorcas/dorcas_home.htm

Marshall University ..... www.marshall.edu/herp

Natural History Museums and Collections

Florida Museum of Natural History ..... www.flmnh.ufl.edu/herpetology

NC Museum of Natural Sciences ..... www.naturalsciences.org/research-collections

American Museum of Natural History ..... http://research.amnh.org/vz

Research Stations

Long Term Ecological Research (LTER) Network. ..... www.lternet.edu

Organization of Biological Field Stations ..... www.obfs.org

APPENDIX XII. MANUFACTURERS AND SUPPLIERS

(Note: This is not intended to be an exhaustive list.)

AVID MicroChip I.D.
78294 Oak Ridge Road
Folsom, LA 70437
http://www.avidid.com
[passive integrated transponders]

AVM Instrument Company, Ltd.
2356 Research Drive
Livermore, CA 94550 USA
www.avminstrument.com
[radiotransmitters]

Bedford Technical
2908 Hickory Hill
Colleyville, TX 76034 USA
www.frogloggers.com
[automated recording systems and microphones]

Belfort Instrument Co.
727 S. Wolfe St.
Baltimore, MD 21231 USA
www.belfortinstrument.com
[dataloggers]

Ben Meadows Company
PO Box 5277
Janesville, WI 53547
www.benmeadows.com
[basic field gear, lab and weather equipment, tools, and more]

Campbell Scientific, Inc.
P.O. Box 551
Logan, UT 84321 USA
www.campbellsci.com
[dataloggers, sensors]

Carolina Biological Supply Company
2700 York Road
Burlington, NC 27215-3398
www.carolina.com
[lab equipment, environmental testing equipment]

Climatronics
140 Wilbur Place
Bohemia, NY 11716 USA
www.climatronics.com
[dataloggers]

Dallas Semiconductor – Maxim
120 San Gabriel Drive
Sunnyvale, CA 94086
www.maxim-ic.com
[ibuttons]

DayGlo Color Corporation
4515 St. Clair Ave
Cleveland, OH 44103
www.dayglo.com
[fluorescent powdered pigments]

Delta-T Devices
128 Low Rd, Burwell,
Cambridge, Cb5 OEJ, U.K.
www.delta-t.co.uk
[dataloggers, thermocouple wire]

ECD
4287-B SE International Way
Milwaukie, OR 97222-8825 USA
www.ecd.com
[dataloggers]

Eppley Laboratories
P.O. Box 419
Newport, RI 02840
www.eppleylab.com
[pyranometers]
Fisher Scientific
www.fishersci.com
[lab equipment and chemicals, UV lamps]

Forestry Suppliers
205 W. Rankin St
PO Box 8397
Jackson, MS 39284
www.forestry-suppliers.com
[basic field gear, lab and weather equipment, measuring devices, tools, and more]
Fuhrman Diversified, Inc.
2912 Bayport Blvd
Seabrook, TX 77586-1501
www.fieldcam.com
[field camera systems]

Hardware stores
(e.g., Lowe's, Home Depot, local hardware stores)
[drift fence materials, coverboard materials, mesh for mesh funnel traps, tools]

Holohil Systems Ltd.
112 John Cavanaugh Drive
Carp, Ontario, Canada K0A 1L0
www.holohil.com
[radiotransmitters]

LI-COR Instruments
Box 4425
Lincoln, NE 68504 USA
www.licor.com
[dataloggers, silicon cell pyranometers]

Memphis Net and Twine
P.O. Box 80331
Memphis, TN 38108-0331
www.memphisnet.net
[turtle traps, dipnets, seines]

Midwest Tongs
14505 S Harris Road
Greenwood, MO 64034
www.tongs.com
[reptile handling tools, scales]

MiniMitter
P.O. Box 3386
Sunriver, OR 97702 USA
www.minimitter.com
[data loggers, radiotransmitters, elvax]

Omega Engineering, Inc.
Box 4047
Stamford, CT 06907 USA
www.omega.com
[data loggers, thermocouple wire, thermistors, pH and conductivity probes]

Onset Computer Corporation
PO Box 3450
Pocasset, MA 02559-3450 USA
www.onsetcomp.com
[data loggers, weather stations, sensors]

Pelican Wire Co.,
Naples, FL USA
www.pelicanwire.com
[thermocouple wire]

REI
Sumner, WA 98352-0001
www.rei.com
[field attire, flashlights, GPS units, and more]

Skye Instruments Ltd.
21, Ddole Enterprise Park,
Llandrindod Wells, Powys LD1 6DF UK
www.skyeinstruments.com
[pyranometers, hygrometers]

Solar Light Company
100 East Glenside Avenue
Glenside, PA 19038 USA
www.solar.com
[UV radiation sensors]

Thermo Electric Co., Inc.
109 North Fifth Street
Saddlebrook, NJ USA
http://www.te-direct.com
[thermocouple wire, thermistors]

LITERATURE CITED

Adams, M. J., K. O. Richter, and W. P. Leonard. 1997. Surveying and monitoring amphibians using aquatic funnel traps. In D. H. Olson, W. P. Leonard, and R. B. Bury (eds.). Sampling Amphibians in Lentic Habitats (Northwest Fauna 4), pp. 47–54. Society for Northwestern Vertebrate Biology, Olympia, Washington.

Adams, S. B., and C. A. Frissell. 2001. Thermal habitat use and evidence of seasonal migration by Rocky Mountain tailed frogs, *Ascaphus montanus*, in Montana. Canadian Field-Naturalist 115:251-256.

Aho, M. J. 1990. Helminth communities of amphibians and reptiles: comparative approaches to understanding patterns and processes. In G. W. Esch, A. O. Bush, and J. Aho (eds.). Parasite Communities: Patterns and Processes, pp. 157-195. Chapman and Hall, London.

Akin, J. A. 1998. Fourier series estimation of ground skink population density. Copeia 2:519-522.

Aldridge, R. D. 1979. Seasonal spermatogenesis in sympatric *Crotalus viridis* and *Arizona elegans* in New Mexico. Journal of Herpetology 13:187-192.

Aldridge, R. D., and W. S. Brown. 1995. Male reproductive cycle, age at maturity, and cost of reproduction in the timber rattlesnake (*Crotalus horridus*). Journal of Herpetology 29:399-407.

Alford, R. A. 1986. Habitat use and positional behavior of anuran larvae in a northern Florida temporary pond. Copeia. 1986:408-23.

Alford, R. A., and S. J. Richards. 1999. Global amphibian declines: a problem in applied ecology. Annual Review of Ecology and Systematics 30:133-165.

Ali, J. H., J. Riley, and J. T. Self. 1984. A revision of the taxonomy of pentastomid parasites (genus *Raillietiella* Sambon, 1910) from American snakes and amphisbaenians. Systematic Parasitology 6:87-97.

Allen, C. R., K. G. Rice, C. P. Wojcik, and H. F. Percival. 1997. Effect of red imported fire ant envenomization on neonatal American alligators. Journal of Herpetology 31: 318-321.

Alley, R. B., P. A. Mayewski, T. Sowers, M. Stuiver, K. C. Taylor, and P. U. Clark. 1997. Holocene climatic instability: a prominent, widespread event 8200 yr ago. Geology 25: 483-486.

Altig, R. 1980. A convenient killing agent for amphibians. Herpetological Review 11:35.

Amstrup, S. C., T. L. McDonald, and B. F. J. Manly. 2005. Handbook of capture-recapture analysis. Princeton University Press, 296 pp.

Anderson, D. R., J. L. Laake, B. R. Crain, and K. P. Burnham. 1979. Guidelines for line transect sampling of biological populations. Journal of Wildlife Management 43(1):70-78.

Andrews, A. 1990. Fragmentation of habitat by roads and utility corridors; a review. The Australian Zoologist 26: 130-141.

Andrews, K. M., and J. W. Gibbons. 2005. How do highways influence snake movement? Behavioral responses to roads and vehicles. Copeia 2005:771-781.

Andrews, K. M., and J. W. Gibbons. 2006. Roads as catalysts of urbanization: snakes on roads face differential impacts due to inter- and intra-specific ecological attributes In J. C. Mitchell, R. E. Jung, and S. C. Walls (eds.). Urban Herpetology. Society for the Study of Amphibians and Reptiles, Herpetological Conservation Vol. 3, Salt Lake City, UT.

Andrews, K. M., J. W. Gibbons, and D. M. Jochimsen. 2007. Ecological effects of roads on amphibians and reptiles: a literature review. In J. C. Mitchell, R. E. Jung, and S. C. Walls (eds.). Urban Herpetology. Society for the Study of Amphibians and Reptiles, Herpetological Conservation Vol. 3, Salt Lake City, UT.

Andrews, K. M., J. W. Gibbons, and D. M. Jochimsen. 2006. Literature synthesis of the effects of roads and vehicles on amphibians and reptiles. Federal Highway Administration (FHWA), U.S. Department of Transportation. Washington, D.C. 151 pp. Available online: www.parcplace.org.

Angilletta, M. J., and A. R. Krochmal. 2003. The thermochron: a truly miniature and inexpensive temperature-logger. Herpetological Review 34:31-32.

Anholt, B. R., S. Negovetic, and C. Som. 1998. Methods for anaesthetizing and marking larval anurans. Herpetological Review 29 (3):153-154.

Antworth, R. L., D. A. Pike, and E. E. Stevens. 2005. Hit and run: effects of scavenging on estimates of roadkilled vertebrates. Southeastern Naturalist 4: 647–656.

Aresco, M. J. 2005a. Mitigation measures to reduce highway mortality of turtles and other herpetofauna at a north Florida lake. Journal of Wildlife Management 69:540-551.

Aresco, M. J. 2005b. The effect of sex-specific terrestrial movements and roads on the sex ratio of freshwater turtles. Biological Conservation 123:37-44.

Arnason, A. N., and C. J. Schwartz. 1987. Popan-3. Extended analysis and testing features for POPAN-2. Charles Babbage Research Center, St. Norbert, Manitoba, Canada.

Ashley, P. E., and J. T. Robinson. 1996. Road mortality of amphibians, reptiles and other wildlife on the Long Point Causeway, Lake Erie, Ontario. Canadian Field Naturalist 110: 403-412.

ASIH (American Society of Ichthyologists and Herpetologists). 2004. Guidelines for use of live amphibians and reptiles in field and laboratory research, 2nd ed.

Associated Press. 2005. Savannah woman dies in hospital after U. S. 17 wreck with alligator. Island Packet, Beaufort, S.C. June 11, 2005.

August, P. V. 1993. GIS in mammalogy: building a database. In S. B. McLaren and J. K. Braun (eds.). GIS applications in mammalogy, pp. 11-26. Special Publication Oklahoma Museum of Natural History, Norman.

August, P. V., C. Baker, C. LaBash, and C. Smith. 1996. Geographic information systems: the storage and analysis of biodiversity data. In D. Wilson, F. R. Cole, J. D. Nichols, R. Rudran, and M. Foster (eds.). Measuring and monitoring biological diversity: standard methods for mammals, pp. 235-246. Smithsonian Institution Press, Washington, D.C.

Bailey, L. L. 2004. Evaluating elastomer marking and photo identification methods for terrestrial salamanders: marking effects and observer bias. Herpetological Review 35(1):38-41.

Bailey, L. L., T. R. Simons, and K. H. Pollock. 2004a. Estimating site occupancy and species detection probability parameters for terrestrial salamanders. Ecological Applications 14:692-702.

Bailey, L. L., T. R. Simons, and K. H. Pollock. 2004b. Comparing population size estimators for plethodontid salamanders. Journal of Herpetology 38:370-380.

Bailey, L. L., T. R. Simons, and K. H. Pollock. 2004c. Estimating detection probability parameters for Plethodon salamanders using the robust capture-recapture design. Journal of Wildlife Management 68:1-13.

Bailey, M. A., J. N. Holmes, K. A. Buhlmann, and J. C. Mitchell. 2006. Habitat Management Guidelines for Amphibians and Reptiles of the Southeastern United States. Partners in Amphibian and Reptile Conservation Technical Publication HMG-2, Montgomery, Alabama. 88pp.

Bailey, N. T. J. 1951. On estimating the size of mobile populations from recapture data. Biometrika 38:293-306.

Bakken, G. S. 1992. Measurement and application of operative and standard operative temperatures in ecology. American Zoologist 32:194-216.

Bakken, G. S., and D. M. Gates. 1975. Heat-transfer analysis of animals: some implications for field ecology, physiology, and evolution. In D. M. Gates and R. B. Schmerl (eds.). Perspectives of biophysical ecology, pp. 255-290. Springer-Verlag, New York.

Balazs, G. H., R. K. Miya, and S. C. Beavers. 1996. Procedures to attach a satellite transmitter to the carapace of an adult green sea turtle, *Chelonia mydas*. Proceedings of the 15th Annual Symposium on Sea Turtle Biology and Conservation. NOAA Technical Memorandum. NMFS-SEFSC-37:21-26.

Barichivich, W. J. 2003. Guidelines for building and operating remote field recorders (automated frog call data loggers). In K. C. Dodd, Jr. (ed.). Monitoring Amphibians in the Great Smoky Mountains National Park, pp. 87-96. United States Geological Service.

Bartareau, T. M. 2004. PVC pipe diameter influences the species and sizes of treefrogs captured in a Florida coastal oak scrub community. Herpetological Review 35:150-152.

Bartlett, R. D., and P. P. Bartlett. 1999. A Field Guide to Florida Reptiles and Amphibians. Gulf Publishing Co. Houston, TX. USA. 280 pp.

Baud, D. R., and M. L. Beck. 2005. Interactive effects of UV-B and copper on spring peeper tadpoles (*Pseudacris crucifer*). Southeastern Naturalist 4(1): 15–22.

Beaupre, S. J., and R. W. Beaupre. 1994. An inexpensive data collection method for temperature telemetry. Herpetologica 50:509-516.

Beebee, T. J. C. 1995. Amphibian breeding and climate. Nature 374: 219-220.

Begon, M. 1979. Investigating animal abundance: capture recapture for biologists. Edward Arnold, London, England.

Benfield, E. F. 1996. Leaf breakdown in stream ecosystems. p.579-589. In F. R. Hauer and G. A. Lamberti (eds.). Methods in stream ecology. Academic Press, San Diego, CA.

Bennett, C., and R. J. Taylor. 1968. Notes on the lesser siren, *Siren intermedia* (Urodela), Southwestern Naturalist 13:455–457.

Bennett, D. H., J. W. Gibbons, and J. C. Franson. 1970. Terrestrial activity of aquatic turtles. Ecology 51(4):738-740.

Bennett, R. A. 1991. A review of anesthesia and chemical restraint in reptiles. Journal of Zoo and Wildlife Medicine 22(3): 282-303.

Bennett, R. A. 1996. Anesthesia. In D. R. Mader (ed.). Reptile Medicine and Surgery, pp. 241-247. W. B. Saunders Company, Toronto, ON.

Berger, L., R. Speare, P. Daszak, D. E. Green, A. A. Cunningham, C. L. Goggin, R. Slocombe, M. Ragan, A. D. Hyatt, K. R. McDonald, H. B. Hines, K. R. Lips, G. Marantelli, and H. Parkes. 1998. Chytridiomycosis causes amphibian mortality associated with population declines in the rain forests of Australia and Central America. Proceedings of the National Academy of Sciences (USA) 95: 9031-9036.

Berish, J. E. D. 1998. Characterization of rattlesnake harvest in Florida. Journal of Herpetology 32(4): 551-557.

Bernardino, Jr., F. S., and G. H. Dalrymple. 1992. Seasonal activity and road mortality of the snakes of the Pa-hay-okee wetlands of Everglades National Park, USA. Biological Conservation 61: 71-75.

Binckley, C. A., B. Plesky, K. Werner, and S. Droege. 1998. Using the visible implant elastomer (VIE) tagging system to mark salamanders. USGS, Patuxent Wildlife Research Center. http://www.pwrc.usgs.gov/resshow/droege2rs/salmark.htm.

Binckley, C. A., B. Plesky, K. Werner, and S. Droege. 1998. Using the visible implant fluorescent elastomer to mark *Plethodon cinereus*. USGS Patuxent Wildlife Research Center. http://www.pwrc.usgs.gov/marking/pwrc.html.

Bingham, M. J., and S. P. Long. 1988. Equipment for crop and environment physiology: specifications, sources and costs. Department of Biology, University of Essex, Colchester, U.K.

Birks, H. H., and B. Ammann. 2000. Two terrestrial records of rapid climatic change during the glacial–Holocene transition (14 000–9000 calendar years BP) from Europe. Proceedings of the National Academy of Sciences 97: 1390–1394.

Bishop, C. A., P. Ng, K. E. Pettit, S. W. Kennedy, J. J. Stegeman, R. J. Norstrom, and R. J. Brooks. 1998. Environmental contamination and developmental abnormalities in eggs and hatchlings of the common snapping turtle (*Chelydra serpentina serpentina*) from the Great Lakes-St Lawrence River basin (1989-91). Environmental Pollution 101:143-156.

Bishop, J. E. 1959. A histological and histochemical study of the kidney tubule of the common garter snake, *Thamnophis sirtalis*, with special reference to the sexual segment in the male. Journal of Morphology 104:307-358.

Blanchard, F. N., and E. B. Finster. 1933. A method of marking living snakes for future recognition, with a discussion of some problems and results. Ecology 14:334-347.

Blankenship, E. L., T. W. Bryan, and S. P. Jacobsen. 1990. A method for tracking tortoises using fluorescent powder. Herpetological Review 21:88-89.

Blaustein, A. R. 1994. Chicken little or Nero's fiddle? A perspective on declining amphibian populations. Herpetologica 50: 85-97.

Blaustein, A. R., P. D. Hoffman, D. G. Hokit, J. M. Kiesecker, S. C. Walls, and J. B. Hays. 1994. UV repair and resistance to solar UV-B in amphibian eggs: a link to population declines? Proceedings of the National Academy of Sciences of the United States of America 91(5): 1791-1795.

Blouin-Demers, G., K. A. Prior, and P. J. Weatherhead. 2002. Comparative demography of black rat snakes (*Elaphe obsoleta*) in Ontario and Maryland. Journal of Zoology, London 256:1-10.

Boarman, W. I., and M. Sazaki. 1996. Highway Mortality in Desert Tortoises and Small Vertebrates: Success of Barrier Fences and Culverts, p. 169-173. In G.L. Evink, P. Garrett, D. Zeigler, and J. Berry (eds.), Trends in Addressing Transportation Related Wildlife Mortality, Proceedings of the Transportation Related Wildlife Mortality Seminar. State of Florida Department of Transportation, Tallahassee, Florida. FL - ER – 58-96.

Boarman, W. I., M. L. Beigel, G. C. Goodlett, and M. Sazaki. 1998. A passive integrated transponder system for tracking animal movements. Wildlife Society Bulletin 26:886-891.

Boone, M. D., and R. D. Semlitsch. 2001. Interactions of an insecticide with larval density and predation in experimental amphibian communities. Conservation Biology 15: 228-238.

Bonnet, X., G. Naulleau, and R. Shine. 1999. The dangers of leaving home: Dispersal and mortality in snakes. Biological Conservation 89: 39-50.

Bothner, R. C. 1963. A hibernaculum of the short-headed garter snake, *Thamnophis brachystoma* Cope. Copeia 3:572-573.

Boughton, R. G., and J. Staiger. 2000. Use of PVC pipe refugia as a sampling technique for hylid treefrogs. American Midland Naturalist 144:168-177.

Boundy, J. 2004. Amphibian and reptile distribution records for Louisiana. Herpetological Review 35:194.

Bradford, D. 1991. Mass mortality and extinction in a high-elevation population of *Rana muscosa*. Journal of Herpetology 25(2):174-177.

Brazaitis, P. 1989. The trade in crocodilians. In C. A. Ross (ed.). Crocodiles and Alligators, pp. 196-201. Facts on File, New York, NY.

Bridges, A. S., and M. E. Dorcas. 2000. Temporal variation in anuran calling behavior: implications for surveys and monitoring programs. Copeia 2000:587-592.

Brokes, B. J. 2000. Habitat segregation of two ambystomatids in mountain ponds, Mount Rainier National Park. MS Thesis, Oregon State University, Corvallis, OR. 55 pp.

Brooks, R. T. 2001. Effects of the removal of overstory hemlock from hemlock-dominated forests on eastern redback salamanders. Forest Ecology and Management 149:197-204.

Brown, G. W. 2001. The influence of habitat disturbance on reptiles in a Box-Ironbark eucalypt forest of south-eastern Australia. Biodiversity and Conservation 10(2):161-176.

Brown, W. S. 1976. A ventral scale clipping system for permanently marking snakes (Reptilia, Serpentes). Journal of Herpetology 10(3):247-249.

Brown, W. S. 1993. Biology, status and management of the Timber Rattlesnake (*Crotalus horridus*): a guide for conservation. Society for the Study of Amphibians and Reptiles (SSAR). Herpetological Circulars No. 22. 78 pp.

Brown, W. S., and W. S. Parker. 1976. Movement ecology of *Coluber constrictor* near communal hibernacula. Copeia (2):225-243.

Brown, W. S., V. P. J. Gannon, and D. M. Secoy. 1984. Paint-marking the rattle of rattlesnakes. Herpetological Review 15(3):75-76.

Brown, W. S., and W. S. Parker. 1982. Niche dimensions and resource partitioning in a Great Basin Desert snake community. Herpetological Communities. U.S. Dept. of the Interior, Fish and Wildlife Service, Wildlife Research Report 13. Washington DC.

Brown, W. S., W. S. Parker, and J.A. Elder. 1974. Thermal and spatial relationships of two species of colubrid snakes during hibernation. Herpetologica 30(1):32-38.

Buchan, A., L. Sun, and R. S. Wagner. 2005. Using alpha numeric fluorescent tags for individual identification of amphibians. Herpetological Review 36:43-44

Bufalino, A. P. 2004. *Hemidactylus turcicus turcicus* (Mediterannean Gecko). Herpetological Review 35:188.

Bugbee, R. E. 1945. A note on the mortality of snakes on highways in western Kansas. Transactions of the Kansas Academy of Science 47: 373-374.

Buhlmann, K. A. 1998. Ecology, terrestrial habitat use, and conservation of a freshwater turtle assemblage inhabiting a seasonally fluctuating wetland with emphasis of the natural history of *Deirochelys reticularia*. Ph. D. Thesis, University of Georgia, Athens.

Buhlmann. K. A., and G. Coffman. 2001. Fire ant predation of turtle nests and implications for the strategy of delayed emergence. The Journal of the Elisha Mitchell Scientific Society 117(2): 94-100.

Buhlmann. K. A., and J. W. Gibbons. 2001. Terrestrial habitat use by aquatic turtles from a seasonally fluctuating wetland: implications for wetland conservation boundaries. Chelonian Conservation and Biology 4(1): 115-127.

Buhlmann, K. A., and M. R. Vaughan. 1991. Ecology of the turtle *Pseudemys concinna* in the New River, West Virginia. Journal of Herpetology 25(1): 72-78.

Buhlmann, K. A., and T. D. Tuberville. 1998. Use of passive integrated transponder (PIT) tags for marking small freshwater turtles. Chelonian Conservation and Biology 3(1): 102-104.

Bull, J. J. 1980. Sex determination in reptiles. Quarterly Review of Biology 55:3-21.

Burbidge, A. A. 1967. The biology of south-western Australian tortoises. PhD. University of Western Australia, Perth.

Burgdorf, S. J., D. C. Rudolph, R. N. Conner, D. Saenz, and R. R. Schaefer. 2005. A successful trap design for capturing large terrestrial snakes. Herpetological Review 36:421-424.

Burger, J., R. T. Zappalorti, M. Gochfeld, W. Boarman, M. Caffrey, V. Doig, S. Garber, B. Lauro, M. Mikovsky, C. Safina, and J. Saliva. 1988. Hibernacula and summer den sites of pine snakes (*Pituophis melanoleucus*) in the New Jersey Pine Barrens. Journal of Herpetology 22(4):425-433.

Burke, R. L., A. A. Hussain, J. M. Storey, and K. B. Storey. 2002. Freeze tolerance and supercooling ability in the Italian Wall Lizards, *Podarcis sicula*, introduced to Long Island, New York. Copeia 2002:836-842.

Burke, R. L., and R. J. Mercurio. 2002. Food habits of a New York population of Italian Wall Lizards, Podarcis sicula (*Reptilia, Lacertidae*). American Midland Naturalist 147:368-375.

Burke, V. J, S. L. Rathbun, J. R. Bodie, and J. W. Gibbons. 1998. Effect of density on predation rate for turtle nests in a complex landscape. Oikos 83:3-11.

Burke, V. J., and J. W. Gibbons. 1995. Terrestrial buffer zones and wetland conservation: A case study of freshwater turtles in a Carolina bay. Conservation Biology 9: 1365-1369.

Burnham K. P., D. R. Anderson, and J. L. Laake. 1980. Estimation of density from line-transect sampling of biological populations. Wildlife Monographs 72:7-202.

Burnham, K. P., and W. S. Overton. 1978. Estimation of the size of a closed population when capture probabilities vary among individuals. Biometrika 65:625.

Burridge, M. J., and L. A. Simmons. 2003. Exotic ticks introduced into the United States on imported reptiles from 1962 to 2001 and their potential roles in international dissemination of diseases. Vet Parasitology 113:289-320.

Burrowes, P. A., R. L. Joglar, and D. E. Green. 2004. Potential causes for amphibian declines in Puerto Rico. Herpetologica 60: 141-154.

Bury, R. B., and P. S. Corn. 1991. Sampling methods for amphibians in streams in the Pacific Northwest. USFS Pacific Northwest Research Station General Technical Report PNW-GTR-275, Portland, Oregon.

Bury, R. B., and P. S. Corn. 1987. Evaluation of pitfall trapping in Northwest forests: trap arrays with drift fences. Journal of Wildlife Management 51(1):112-119.

Butler, J. A., and T. W. Hull. 1996. Reproduction of the gopher tortoise, *Gopherus polyphemus*, in northeastern Florida. Journal of Herpetology 30: 455-458.

Butts, S. R., and W. C. McComb. 2000. Associations of forest-floor vertebrates with coarse woody debris in managed forests of western Oregon. Journal of Wildlife Management 64:95-104.

Campbell, G. S. 1990. Biophysical Measurements and Instrumentation: A Laboratory Manual for Environmental Physics. Department of Agronomy and Soils, Washington State University, Pullman, WA.

Campbell, H. W. and S. P. Christman. 1982. Field techniques for herpetofaunal community analysis. In N. J. Scott, Jr. (ed.). Herpetological Communities: A Symposium for the Society for the Study of Amphibians and Reptiles and the Herpetologists' League, August 1977, pp. 192-200. Wildlife Research Report 13. United States Department of the Interior, Fish and Wildlife Service, Washington, D.C.

Campbell, T. S. 1996. Northern range expansion of the brown anole (*Anolis sagrei*) in Florida and Georgia. Herpetological Review 27:155-157.

Card, W., and A. G. Kluge. 1995. Hemipeneal skeleton and varanid lizard systematics. Journal of Herpetology 29:275-280.

Card, W., and D. Mehaffey. 1994. A radiographic sexing technique for *Heloderma suspectum*. Herpetological Review 25:17-19.

Cardona L., M. Revelles, C. Carreras, M. San Felix, M. Gazo, and A. Aguilar. 2005. Western Mediterranean immature loggerhead turtles: habitat use in spring and summer assessed through satellite tracking and aerial surveys. Marine Biology 147:583-591.

Carey, C. L., and M. A. Alexander. 2003. Climate change and amphibian declines: is there a link? Diversity and Distributions 2003: 111-121.

Carey, C., A. P. Pessier, and A. D. Peace. 2003. Pathogens, Infectious Disease, and Immune Defenses, p. 127-136. In R. D. Semlitsch (ed.). Amphibian Conservation, Smithsonian Books, Washington, D. C.

Carey, C., N. Cohen, and L. Rollins-Smith. 1999. Amphibian declines: an immunological perspective. Developmental and Comparative Immunology 23: 459-472.

Carey, S. D. 1982. Geographic distribution: *Eleutherodactylus planirostris*. Herpetological Review 13:130.

Carpenter, C. C. 1953. A study of hibernacula and hibernating associations of snakes and amphibians in Michigan. Ecology 34(1):74-80.

Carpenter, C. C. 1957. Hibernation, hibernacula and associated behavior of the three-toed box turtle (*Terrapene carolina triunguis*). Copeia 1957(4):278-282.

Carr, A. 1952. Handbook of Turtles: the Turtles of the United States, Canada, and Baja California. Cornell University Press, Ithaca, NY.

Carr, L., and L. Fahrig. 2001. Effect of road traffic on two amphibian species of differing vagility. Conservation Biology 15: 1071-1078.

Carter, S. L., C. A. Haas, and J. C. Mitchell. 1999. Home range and habitat selection of bog turtles in southwestern Virginia. Journal of Wildlife Management 63:853-860.

Casazza, M. L., G. D. Wyle, and C. J. Gregory. 2000. A funnel trap modification for surface collection of aquatic amphibians and reptiles. Herpetological Review 31(2):91-92.

Case, R. M. 1978. Interstate highway road-killed animals: a data source for biologists. Wildlife Society Bulletin 6:8-13.

Cassey, P., and G. T. Ussher. 1999. Estimating abundance of tuatara. Biological Conservation 88:361-366.

Castanet, J., H. Francillon-Vieillot, F. J. Meunier, and A. de Ricqlès. 1993. Bone and individual aging. In B. K. Hall, (ed.). Bone, pp. 245-283. CRC Press, London.

Caughley, G. 1974. Bias in aerial survey. Journal of Wildlife Management, 38(4):921-933.

Caughley, G. 1977. Sampling in aerial survey. Journal of Wildlife Management, 41(4):605-615.

Caughley, G., and J. Goddard. 1972. Improving the estimates from inaccurate censuses. Journal of Wildlife Management 36:135-140.

Caughley, G., Sinclair, R., and D. Scott-Kemmis. 1976. Experiments in aerial survey. Journal of Wildlife Management 40(2):290-300.

Chabreck, R. H. 1963. Methods of capturing, marking and sexing alligators. Proceedings of the Annual Conference of the Southeastern Association of Game and Fish Commission 17:47-50.

Chalmers, R. J., and S. Droege. 2002. Leaf litter bags as an index to populations of northern two-lined salamanders (*Eurycea bislineata*). Wildlife Society Bulletin 30:71-74.

Chapman, D. G. 1951. Some properties of the hypergeometric distribution with applications to zoological censuses. University of California Publications in Statistics. 1: 131-160.

Chapple, D. G., McCoull, C. J., and R. Swain. 2002. Changes in reproductive investment following caudal autotomy in viviparous skinks (*Niveoscincus metallicus*): Lipid depletion or energetic diversion? Journal of Herpetology 36(3): 480-486.

Church, D. R. 2003. Cost of reproduction in a variable environment of the eastern tiger salamander (*Ambystoma tigrinum tigrinum*). Dissertation. University of Virginia, Charlottesville, Virginia, USA.

Clark, D. R. 1971. Branding as a marking technique for amphibians and reptiles. Copeia 1971(1):148-151.

Clarke, J. A., J. T. Chopko, S. P. Mackessy. 1996. The effect of moonlight on activity patterns of adult and juvenile prairie rattlesnakes (*Crotalus viridis viridis*). Journal of Herpetology 20:192-197.

Clarke, R. D. 1972. The effect of toe clipping on survival in Fowler's toad (*Bufo woodhousei fowleri*). Copeia 1972:182-185.

Clevenger, A. P., M. McIvor, D. McIvor, B. Chruszcz, and K. Gunson. 2001. Tiger salamander, *Ambystoma tigrinum*, movements and mortality on the Trans-Canada Highway in southwestern Alberta. The Canadian Field-Naturalist 115: 199-204.

Cobb, V. A. 1994. The ecology of pregnancy in free-ranging great basin rattlesnakes (*Crotalus viridis lutosus*). Ph.D. Dissertation, Idaho State University, Pocatello, ID.

Cohen, E. 1948. Emergence of *Coluber c. constrictor* from hibernation. Copeia 1948:137-138.

Colburn, T., D. Dumanoski, and J. P. Myers. 1997. Our Stolen Future: Are We Threatening Our Fertility, Intelligence, and Survival? A Scientific Detective Story. Penguin Books USA, Inc. New York, NY.

Coles, W. C., and J. A. Musick. 2000. Satellite sea surface temperature analysis and correlation with sea turtle distribution off North Carolina. Copeia 2000: 551-554.

Conant, R. 1948. Regeneration of clipped subcaudals scales in a pilot black snake. Chicago Academy of Science, Nat. Hist. Misc. No. 13, 2 pp.

Conant, R., and J. T. Collins. 1998. A Field Guide to Reptiles and Amphibians of Eastern and Central North America. Third Edition, Expanded. Houghton Mifflin Company, Boston. 616 pp.

Congdon, J. D., A. E. Dunham, and R. C. van Loben Sels. 1993. Delayed sexual maturity and demographics of Blanding's turtles (*Emydoidea blandingii*): implications for conservation and management of long-lived organisms. Conservation Biology 7:826-833.

Congdon, J. D., R. D. Nagle, O. M. Kinney, M. Ostentoski, H. W. Avery, R. C. van Loben Sels, and D. W. Tinkle. 2000. Nesting ecology and embryo mortality: implications for hatchling success and demography of Blandings's turtles (*Emydoidea blandingii*). Chelonian Conservation and Biology 3:569-579.

Congdon, J. D., R. E. Ballinger, and K. A. Nagy. 1979. Energetics, temperature and water relations in winter aggregated *Sceloporus jarrovi* (Sauria: Iguanidae). Ecology 60(1):30-35.

Conroy, M. J., and J. D. Nichols. 1996. Designing a study to assess mammalian diversity. In D. E. Wilson, F. R. Cole, J. D. Nichols, R. Rudran, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for mammals, pp.41-49. Smithsonian Institution Press.

Cooper Jr., W. E., and N. Greenberg. 1992. Reptilian coloration and behavior. In C. Gans, and D. Crews (eds.). Hormones, Brain and Behavior. Biology of the Reptilia, pp 298-422. Chicago, University of Chicago Press.

Cooper, J. E., R. Ewbank, C. Platt, and C. Warwick (eds.). 1989. Euthanasia of Amphibians and Reptiles. University Federation for Animal Welfare, Potters Bar.

Cooper-Preston, H. 1992. Geographic variation in the population dynamics of *Crocodylus johnstoni* (Krefft) in three rivers in the Northern Territory, Australia. PhD dissertation, University of New England, Armidale.

Copeyon, C. K. 1997. Endangered and threatened wildlife and plants: Final rule to list the northern population of the bog turtle as threatened and the southern population as threatened due to similarity of appearance. Federal Register 62: 59605-59623.

Corn, P. S. 1994. Straight-line drift fences and pitfall traps. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp.109-117. Smithsonian Institution Press, Washington, DC. 364 pp.

Corn, P. S., and R. B. Bury. 1989. Logging in Oregon: responses of headwater habitats and stream amphibians. Forest Ecology and Management 29:23-57.

Corn, P. S., and R. B. Bury. 1990. Sampling methods for terrestrial amphibians and reptiles. U.S. Department of Agriculture, Forest Service, General Technical Report PNW-GTR-256.

Corn, P. S., E. Muths, and W. M. Iko. 2000. A comparison in Colorado of three methods to monitor breeding amphibians. Northwestern Naturalist 81:22–30.

Cowx, I. G., and P. Lamarque. 1990. Fishing with Electricity, application in freshwater fisheries management. Fishing News Books: Oxford, United Kingdom.

Crawford, K. M., J. R. Spotila, and E. A. Standora. 1983. Operative environmental temperature and basking behavior of the turtle *Pseudemys scripta*. Ecology 64:989-999.

Crayon, J. J. 2005. *Xenopus laevis*. In M. Lannoo editor, Amphibian Declines: The Conservation Status of United States Species, pp. 522-526. University of California Press, Berkeley, CA. USA. 1094 pp.

Cree, A., J. F. Cockrem, M. A. Brown, P. R. Watson, L. J. Guillette Jr., D. G. Newman, and G. K. Chambers. 1991. Laparoscopy, radiography, and blood analyses as techniques for identifying the reproductive condition of female tuatara. Herpetologica 47:238-249.

Criddle, S. 1937. Snakes from an ant hill. Copeia 1937:142.

Cristoffer, C. 1991. Road mortalities of northern Florida vertebrates. Florida Scientist 54: 65-68.

Crouch, W. B. and P. W. C. Paton. 2000. Using egg-mass counts to monitor wood frog populations. Wildlife Society Bulletin 28: 895-901.

Crump, M. L. 1994. Climate and environment. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp. 42-46. Smithsonian Institution Press, Washington, DC. 364 pp.

Crump, M. L., and N. J. Scott, Jr. 1994. Visual encounter surveys. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp. 84-92. Smithsonian Institution Press, Washington, DC. 364 pp.

Dahl, T. E. 2000. Status and trends of wetlands in the conterminous United States 1986 to 1997. U. S. Fish and Wildlife Service, Washington, D. C.

Daszak, P., and A. Cunningham. 2000. Emerging infectious diseases of wildlife – threats to biodiversity and human health. Science 287:443-449.

Daszak, P., D. E. Scott, A. M. Kilpatrick, C. Faggioni, J. W. Gibbons, and D. Porter. 2005. Amphibian population declines at Savannah River Site are linked to climate, not chytridiomycosis. Ecology 86: 3232-3237.

Daszak, P., L. Berger, A. A. Cunningham, A. D. Hyatt, D. E. Green, and R. Speare. 1999. Emerging infectious diseases and amphibian population declines. Emerging Infectious Diseases 5:735-748.

David, D, D. Brunell, D. Carbonneau, H. Dutton, L. Hord, N. Wiley, and A. Woodward. 1996. Florida's alligator management program, an update 1987-1995. In Proceedings of the 13th Working Meeting of the Crocodile Specialist Group, pp. 410-428. IUCN-The World Conservation Union. 11-17 May 1996. Gland, Switzerland.

de Buffrénil, V. 1982. Morphogenesis of bone ornamentation in extant and extinct crocodilians. Zoomorphology 99:155-166.

DeNardo, D. 1996. Reproductive biology. In D. R. Mader (ed.). Reptile Medicine and Surgery, pp. 212-224. Philadelphia, W. B. Saunders.

Dessauer, H. C. 1970. Blood chemistry of reptiles: physiological and evolutionary aspects. In C. Gans (ed.). Biology of the Reptilia, Vol. 3. Academic Press, New York.

Dessauer, H. C., C. J. Cole, and M. S. Hafner. 1996. Collection and storage of tissues. In Hillis et al. (eds.). Molecular Systematics. Sinauer, Sunderland, MA.

DeVault, T. L., and O. E. Rhodes, Jr. 2002. Identification of vertebrate scavengers of small mammal carcasses in a forested landscape. Acta Theriologica 47:185–192.

Dinsmore, A. 2004. Geographic distribution: *Eleutherodacytlus planirostris* (Greenhouse frog). Herpetological Review 35:403.

Dixon, J. R. 2000. Amphibians and Reptiles of Texas. Texas A & M University Press. College station, TX. USA. 421 pp.

Dixon, J. R., T. J. Hibbitts, and M. R. J. Forstner. 2007. Texas herpetological literature and county records 2000 to 2006-07. Texas Herpetological Society Publications 2007 1:1-58.

Docherty, D. E., C. U. Meteyer, J. Wang, J. Mao, S. T. Case, and V. G. Chinchar. 2003. Diagnostic and molecular evaluation of three iridovirus-associated salamander events. Journal of Wildlife Diseases 39(3):556-566.

Dodd, C. K., Jr. 1988. Disease and population declines in the flattened musk turtle, *Sternotherus depressus*. American Midlands Naturalist 119:394-401.

Dodd C. K., Jr., K. M. Enge, and J. N. Stuart. 1989. Reptiles on highways in north-central Alabama, USA. Journal of Herpetology 23:197-200.

Dodd, C. K., Jr. 1990. Line transect estimation of Red Hills salamander burrow density using a Fourier series. Copeia (2):555-557.

Dodd, C. K., Jr. 1992. Fluorescent powder is only partially successful in tracking movements of the six-lined racerunner (*Cnemidophorus sexlineatus*). Florida Field Naturalist 20:8-14.

Dodd, C. K., Jr. 1997. Imperiled amphibians: a historical perspective. In G. W. Benz and D. E. Collins (eds.). Aquatic Fauna in Peril: The Southeastern Perspective, pp. 165-200. Southeast Aquatic Research Institute, Lenz Design and Communications, Decatur, GA. Special Publication 1.

Dodd, C. K., Jr. 2003. Monitoring amphibians in Great Smoky Mountains National Park. U.S. Geological Survey Circular 1258, Tallahassee, FL. 117 pp.

Dodd, C. K., Jr., and D. E. Scott. 1994. Drift fences encircling breeding sites. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp.125-130. Smithsonian Institution Press, Washington, DC. 364 pp.

Dodd, C. K., Jr., and L. L. Smith. 2003. Habitat destruction and alteration: historical trends and future prospects for amphibians. In R. D. Semlitsch (ed.). Amphibian Conservation, pp. 94-112. Smithsonian Books, Washington, D. C.

Donnelly, M. A. 1989. Demographic effects of reproductive resource supplementation in a territorial frog, *Dendrobates pumilio*. Ecological Monographs 59:207-221.

Donnelly, M. A., C. Guyer, J. E. Juterbock, and R. A. Alford. 1994. Techniques for marking amphibians. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster. Measuring and monitoring biological diversity: standard methods for amphibians, pp. 277-284. Smithsonian Institution Press, Washington, DC.

Dorcas, M. E., and C. R. Peterson. 1998. Daily body temperature variation in free-ranging rubber boas. Herpetologica 54:88-103.

Dorcas, M. E. and K. D. Foltz. 1991. Environmental effects on anuran advertisement calling. American Zoologist 31:111A.

Douglas, M. E. 1979. Migration and sexual selection in *Ambystoma jeffersonianum*. Canandian Journal of Zoology 57(12): 2303-2310.

Downes, S., and R. Shine. 2001. Why does tail loss increase a lizard's later vulnerability to snake predators? Ecology 82(5):1293-1303.

Drda, W. J. 1968. A study of snakes wintering in a small cave. Journal of Herpetology 1:64-70.

Duellman, W. E. 1954. Observations on autumn movements of *Ambystoma tigrinum* in southeastern Michigan. Copeia 1954: 156-157.

Duellman, W. E., and L. Trueb. 1986. Biology of Amphibians. Baltimore, John Hopkins University Press.

Duever, M. J. 1967. Distributions in space and time of reptiles on the Savannah River Plant in South Carolina. Unpubl. M.S. Thesis. 70 pp. University of Georgia. Athens, GA.

Dundee, H. A., and D. A. Rossman. 1989. The Amphibians and Reptiles of Louisiana. Louisiana State University Press. Baton Rouge, LA. USA. 300 pp.

Dupuis, L. and P. A. Friele. 2006. The distribution of the Rocky Mountain tailed frog (*Ascaphus montanus*) in relation to the fluvial system: implications for management and conservation. Ecological Research 21: 489-502.

Durham, L., and G. W. Bennett. 1963. Age, growth and homing in the bullfrog. Journal of Wildlife Management 27: 107-123.

Dwyer, W. P., and R. G. White. 1995. Influence of electroshock on short term growth of adult rainbow trout and juvenile Arctic grayling and cutthroat Trout. North American Journal of Fisheries Management 15:148-151.

Eason, G. W., Jr., and D. R. McMillan. 2000. Geographic distribution: *Hemidactylus turcicus* (Mediterranean gecko). Herpetological Review 31:53.

Easterling, D. R., T. R. Karl, K. P. Gallo, D. A. Robinson, K. E. Trenberth, and A. Dai. 2000. Observed climate variability and change of relevance to the Biosphere. Journal of Geophysical Research 105: 20101-20114.

Eggert, C. 2002. Use of fluorescent pigments and implantable transmitters to track a fossorial toad (*Pelobates fuscus*). Herpetological Journal 12:69-74.

Eggert, C., P. H. Peyret, and R. Guyetant. 1999. Two complementary methods for studying amphibian terrestrial movements. In C. Miaud, and R. Guyetant (eds.). Current Studies in Herpetology, pp. 95-97. SEH, Le Bourget du Lac, France.

Emery, L., and R. S. Wydoski. 1987. Marking and tagging of aquatic animals: an indexed bibliography. U.S. Fish and Wildlife Service, Research Publication 165. 57pp.

Enge, K. M. 1993. Herptile use and trade in Florida. Final Performance Report, Bureau of Nongame Wildlife, Florida Game and Fresh Water Fish Commission, Quincy, FL.

Enge, K. M. 1997. A standardized protocol for drift-fence surveys. Florida Game and Fresh Water Fish Commission: 68. Quincy, Florida.

Enge, K. M. 2001. The pitfalls of pitfall traps. Journal of Herpetology 35(3):467-478.

Enge, K. M., and K. N. Wood. 2002. A pedestrian road survey of an upland snake community in Florida. Southeastern Naturalist 1:365-380.

Enge, K. M., and K. L. Krysko. 2004. A new exotic species in Florida, the bloodsucker lizard, *Calotes versicolor* (Daudin 1802) (Sauria: Agamidae). Florida Scientist 67:226-230.

Enge, K. M., K. L. Krysko, and B. L. Talley. 2004a. Distribution and ecology of the introduced African rainbow lizard, *Agama agama africana* (Sauria: Agamidae), in Florida. Florida Scientist 67:303-310.

Enge, K. M., K. L. Krysko, K. R. Hankins, T. S. Campbell, and F. W. King. 2004b. Status of the Nile monitor (*Varanus niloticus*) in southwestern Florida. Southeastern Naturalist 3:571-582.

Enge, K. M., B. W. Kaiser, and R. B. Dickerson. 2006. Another large exotic lizard in Florida, the Argentine black and white tegu. Abstract in Proceedings of the 28th Annual Gopher Tortoise Council Meeting, 26-29 October 2006. Valdosta, GA.

Ernst, C. H. 1965. Bait preferences of some freshwater turtles. Journal of the Ohio Herpetological Society 5(2):53.

Ernst, C. H., J. E. Lovich, and R. W. Barbour. 1994. Turtles of the United States and Canada. Smithsonian Institution Press. Washington, D.C. USA. 578 pp.

Fahrig, L. J., H. Pedlar, S. E. Pope, P. D. Taylor, and J. F. Wegner. 1995. Effect of road traffic on amphibian density. Biological Conservation 73:177-182.

Feder, M. E., and W. W. Burggren (Editors). 1992. Environmental Physiology of the Amphibians. University of Chicago Press.

Feinberg, J. A. 2004. Nest predation and ecology of terrapins, *Malaclemys terrapin terrapin*, at the Jamaica Bay Wildlife Refuge. In C. Swarth, W. M. Roosenburg, and E. Kiviat (eds.). Conservation and Ecology of Turtles of the Mid-Atlantic Region: A Symposium. Biblomania: Salt Lake City, UT.

Fellers, G. M., and C. A. Drost. 1989. Fluorescent powder: a method for tracking reptiles. Herpetological Review 20:91-92.

Fellers, G. M., and C. A. Drost. 1994. Sampling with artificial cover. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp.146-150. Smithsonian Institution Press, Washington, DC. 364 pp.

Fellers, G. M., and K. L. Freel. 1995. A standardized protocol for surveying aquatic amphibians. U.S. Department of the Interior, National Biological Service Technical Report NPS/WRUC/NRTR-59-001. 123 pp.

Ferner, J. W., and J. P. Ferner. 2002. Geographic distribution: *Podarcis muralis* (common wall lizard). Herpetological Review 33:226.

Findlay, C. S., and J. Bourdages. 2000. Response time of wetland biodiversity to road construction on adjacent lands. Conservation Biology 14: 86-94.

Finklestein, P. L., J. C. Kaimal, J. E. Gaynor, M. E. Graves, and T. J. Lockhart. 1986. Comparison of wind monitoring systems. Part I: In situ sensors. Journal of Atmospheric and Oceanic Technology 3:583-593.

Fisher, M., and A. Muth. 1989. A technique for permanently marking lizards. Herpetological Review 20(2):45-46.

Fitch, H. S. 1949. Road counts of snakes in western Louisiana. Herpetologica 5: 87-90.

Fitch, H. S. 1987. Collecting and life-history techniques. In R. A. Seigel, J. T. Collins, and S. S. Novak (eds.). Snakes: Ecology and Evolutionary Biology, pp. 143-164. Macmillan, New York.

Fitch, H. S. 1992. Methods of sampling snake populations and their relative success. Herpetological Review 23(1):17-19.

Fitzgerald, G. J., and J. R. Bider. 1974. Influence of moon phase and weather factors on locomotory activity in *Bufo americanus*. Oikos 25:338-340.

Fitzgerald, L. A., and C. W. Painter. 2000. Rattlesnake commercialization: long-term trends, issues, and implications for conservation. Wildlife Society Bulletin 28(1):235-253.

Flowers, E. C. 1978. Comparison of solar radiation sensors from various manufacturers. NOAA, ERL, Solar Radiation Facility, Boulder, CO.

Forman, R. T. 2000. Estimate of area affected ecologically by the road system in the United States. Conservation Biology 14(1):31-35.

Forman, R. T. T., D. Sperling, J. A. Bissonette, A. P. Clevenger, C. D. Cutshall, V. H. Dale, L. Fahrig, R. France, C. R. Goldman, K. Heanue, J. A. Jones, F. J. Swanson, T. Turrentine, and T. C. Winter. 2003. Road Ecology: Science and Solutions. Island Press. Washington, D.C.

Fox, H. 1977. The urinogenital system of reptiles. In C. Gans, and T. S. Parsons (eds.). Biology of the Reptilia, Vol. 6. Morphology, pp. 1-157. Academic Press, New York.

Frantz, A. C., L. C. Pope, P. J. Carpenter, T. J. Roper, G. J. Wilson, R. J. Delahay, and T. Burke. 2003. Reliable microsatellite genotyping of the Eurasian badger (*Meles meles*) using faecal DNA. Molecular Ecology 12:1649-1661.

Frick, M. G., C. K. Slay, C. A. Quinn, A. Windham-Reid, P. A. Duley, C. M. Ryder, and L. J. Morse. 2000. Aerial observations of courtship behavior in loggerhead sea turtles (*Caretta caretta*) from southeastern Georgia and northeastern Florida. Journal of Herpetology 34: 153-158.

Friend, G. R. 1984. Relative abundance of two pitfall-drift fence systems for sampling small vertebrates. Australian Zoologist 21(5): 423-434.

Fritschen, L. J., and L. W. Gay. 1979. Environmental Instrumentation. Springer-Verlog, New York.

Frost, C. C., J. Walker, and R. K. Peet. 1986. Fire-dependent savannas and prairies of the Southeast: original extent, preservation status and management problems. In D. L. Kulhavy and R. N. Connor (eds.). Wilderness and Natural Areas in the Eastern United States: A Management Challenge, pp. 348-357. Center for Allied Studies, School of Forestry, Stephen F. Austin University, Nacogdoches, Texas.

Frye, F. L. 1984. Euthanasia, necropsy techniques and comparative histology of reptiles. In G. L. Hoff, F. L. Frye, E. R. Jacobson (eds.). Diseases of Amphibians and Reptiles. Plenum Press, NY.

FSP (Floridians for a Sustainable Population). 2005. Retrieved March 2006 from the World Wide Web: www.flsuspop.org.

Fukatsu, T. 1999. Acetone preservation: a practical technique for molecular analysis. Molecular Ecology 8:1935-1945.

Fukumoto, J., and S. Herrero. 1998. Observations of the long-toed salamander, *Ambystoma macrodactylum*, in Waterton Lakes National Park, Alberta. Canadian Field-Naturalist 112: 579-585.

Funk, W. C., D. Almeida-Reinoso, F. Nogales-Sornosa, and M. R. Bustamante. 2003. Monitoring population trends of *Eleutherodactylus frogs*. Journal of Herpetology 37(2): 245-256.

Gaby, R. 1987. Utilization of human-altered habitats by American crocodiles in southern Florida. In R. R. Odum, K. A. Riddleberger, and J. C. Ozier (eds.). Proceedings of the 3rd Southeastern Nongame and Endangered Wildlife Symposium. University of Georgia. Athens, GA.

Galligan, J. H., and W. A. Dunson. 1979. Biology and status of timber rattlesnake (*Crotalus horridus*) populations in Pennsylvania. Biological Conservation 15:13-58.

Games, I. 1990. Growth curves for the Nile crocodile as estimated by skeletochronology. In Proceedings 10th Working Meeting, pp. 111-121. IUCN/SSC Crocodile Specialist Group.

Gates, J. E., C. H. Hocutt, J. R. Stauffer, Jr., and G. J. Taylor. 1985. The distribution and status of *Cryptobranchus alleganiensis* in Maryland. Herpetological Review 16:17-18.

Georges, A. 1985. Reproduction and reduced body size of reptiles in unproductive insular environments. In G. Grigg, R. Shine, and H. Ehmann (eds.). Biology of Australasian Frogs and Reptiles, pp. 311-318. Royal Zoological Society, New South Wales, Mosman.

Gibbons, J. W. 1970. Terrestrial activity and the population dynamics of aquatic turtles. American Midland Naturalist 83(2): 404-414.

Gibbons, J. W. 2003. Terrestrial habitat: a vital component for herpetofauna of isolated wetlands. Wetlands 23(3):630-635.

Gibbons, J. W., and J. L. Greene. 1979. X-ray photography: a technique to determine reproductive patterns of freshwater turtles. Herpetologica 35:86-89.

Gibbons, J. W., and K. M. Andrews. 2004. PIT tagging: Simple technology at its best. BioScience 54:447-454.

Gibbons, J. W., and R. D. Semlitsch. 1981. Terrestrial drift fences with pitfall traps: An effective technique for quantitative sampling of animal populations. Brimleyana 1981:1-16.

Gibbons, J. W., C. T. Winne, D. E. Scott, J. D. Willson, X. Glaudas, K. M. Andrews, B. D. Todd, L. A. Fedewa, L. Wilkinson, R. N. Tsaliagos, S. J. Harper, J. L. Greene, T. D. Tuberville, B. S. Metts, M. E. Dorcas, J. P. Nestor, C. A. Young, T. Akre, R. N. Reed, K. A. Buhlmann, J. Norman, D. A. Croshaw, C. Hagen, and B. B. Rothermel. 2006. Remarkable amphibian biomass and abundance in an isolated wetland, with implications for wetland conservation. Conservation Biology 20:1457-1465.

Gibbons, J. W., D. E. Scott, T. J. Ryan, K. A. Buhlmann, T. D. Tuberville, B. S. Metts, J. L. Greene, T. Mills, Y. Leiden, S. Poppy, and C. T. Winne. 2000. The global decline of reptiles, déjà vu amphibians. Bioscience 50(8):653-666.

Gibbons, J. W., J. L. Greene, and T. Mills. 2008. Lizards and Crocodilians of the Southeast. University of Georgia Press. Athens, GA.

Gibbons, J. W., J. W. Coker, and T. M. Murphy, Jr. 1977. Selected aspects of the life history of the rainbow snake (*Farancia erytrogramma*). Herpetologica 33(3): 2676-281.

Gibbons, J. W., V. J. Burke, J. E. Lovich, R. D. Semlitsch, T. D. Tuberville, J. R. Bodie, J. Greene, P. H. Niewiarowski, H. H. Whiteman, D. E. Scott, J. H. K. Pechmann, C. R. Harrison, S. H. Bennett, J. D. Krenz, M. S. Mills, K. A. Buhlmann, J. R. Lee, R. A. Seigel, A. D. Tucker, T. M. Mills, T. Lamb, M. E. Dorcas, N. B. Frazer, J. D. Congdon, M. H. Smith, D. H. Nelson, M. B. Dietsch, H. G. Hanlin, J. A. Ott, and D. J. Karapatakis. 1997. Perceptions of species abundance, distribution, and diversity: lessons from four decades of sampling on a government-managed reserve. Environmental Management 2:259-268.

Gibbs, E. L., G. W. Nace, and M. B. Emmons. 1971. The live frog is almost dead. Bioscience 21: 1027-1034.

Gibbs, J. P. 2000. Wetland loss and biodiversity conservation. Conservation Biology 14:314-317.

Gibbs, J. P., and A. R. Briesch. 2001. Climate warming and calling phenology of frogs near Ithaca, New York, 1900–1999. Conservation Biology 15:1175–1178.

Gibbs, J. P., and W. G. Shriver. 2002. Estimating the effects of road mortality on turtle populations. Conservation Biology 16: 1647-1652.

Gienger, C. M., D. D. Beck, N. C. Sabari, and D. L. Stumbaugh. 2002. Dry season habitat use by lizards in a tropical deciduous forest of western Mexico. Journal of Herpetology 36(3):487-490.

Gill, D. E. 1978. The metapopulation ecology of the red-spotted newt, *Notophthalmus viridescens* (Rafinesque). Ecological Monographs 48(2):145-166.

Glaudas, X. A., K. M. Andrews, J. D. Willson, and J. W. Gibbons. 2007. Migration patterns in a population of cottonmouths (*Agkistrodon piscivorus*) inhabiting an isolated wetland. Journal of Zoology 271:119-124.

Golay, N., and H. Durrer. 1994. Inflammation due to toe-clipping in natterjack toads (*Bufo calamita*). Amphibia-Reptilia 15(1):81-83.

Gomez de Segura, A., J. Tomas, S. N. Pedraza, E. A. Crespo, and J. A. Raga. 2003. Preliminary patterns of distribution and abundance of loggerhead sea turtles, *Caretta caretta*, around Columbretes Islands Marine Reserve, Spanish Mediterranean. Marine Biology 143:817-823.

Gorzula, S. J. 1978. An ecological study of *Caiman crocodilus crocodilus* inhabiting savanna lagoons in the Venezuelan Guayana. Oecologia 35:21-34.

Graeter, G. J. 2005. Habitat selection and movement patterns of amphibians in altered forest habitats. M.S. Thesis, University of Georgia, Athens.

Graeter, G. J., and B. B. Rothermel. 2007. The effectiveness of fluorescent powdered pigments as a tracking technique for amphibians. Herpetological Review 38:162-166.

Graham, T. E. 1979. Life History Techniques. In M. Harless and H. Morlock (eds.). Turtles: Perspectives and Research, pp 73-95. J. Wiley and Sons, New York.

Graham, T. E. 1981. New approaches to endangered turtle research. Bioscience 52:121-126.

Grant, B. W., A. D. Tucker, J. E. Lovich, A. M. Mills, P. M. Dixon, and J. W. Gibbons. 1992. The use of coverboards in estimating patterns of reptile and amphibian biodiversity. In D. R. McCullough and R. H. Barrett (eds.). Wildlife 2001, pp. 379-403. Elsevier Science Publications, London.

Grant, E. H. C., R. E. Jung, J. D. Nichols, and J. E. Hines. 2005. Double-observer approach to estimating egg mass abundance of pool-breeding amphibians. Wetlands Ecology and Management 13:305-320.

Grant, B. W., and A. E. Dunham. 1988. Thermally imposed time constraints on the activity of the desert lizard *Sceloporus merriami*. Ecology 69:167-176.

Graves, B. M., and D. Duvall. 1987. An experimental study of aggregation and thermoregulation in prairie rattlesnakes, *Crotalus viridis viridis*. Herpetologica 43:253-258.

Graves, B. M., and D. Duvall. 1990. Spring emergence patterns of prairie rattlesnakes and wandering garter snakes in Wyoming. Journal of Herpetology 24:351-356.

Gray, J. S. 1997. Marine biodiversity: patterns, threats, and conservation needs. Biodiversity and Conservation 6:153-175.

Grayson, K. L., and M. E. Dorcas. 2004. Seasonal temperature variation in the painted turtle (*Chrysemys picta*). Herpetologica 60:325-336.

Green, D. E., K. A. Converse, and A. K. Schrader. 2002. Epizootiology of sixty-four amphibian morbidity and mortality events in the USA, 1996-2001. Annals of the New York Academy of Science 969:323-339.

Green, D. M. (ed.). 1997. Amphibians in Decline: Canadian Studies of a Global Problem. Herpetological Conservation, Volume 1. Society for the Study of Amphibians and Reptiles, St. Louis, MO.

Greenberg, C. H., and G. W. Tanner. 2005. Spatial and temporal ecology of eastern spadefoot toads on a Florida landscape. Herpetologica 61(1):20-28.

Greenberg, C. H., D. G. Neary, and L. D. Harris. 1994. A comparison of herpetofaunal sampling effectiveness of pitfall, single-ended, and double-ended funnel traps used with drift fences. Journal of Herpetology 28(3):319-324.

Gregory, P. T. 1974. Patterns of spring emergence of the red-sided garter snake (*Thamnophis sirtalis parietalis*) in the Interlake region of Manitoba. Canadian Journal of Zoology 52:1063-1069.

Gregory, P. T. 1984. Communal denning in snakes. In R. A. Seigel, L. E. Hunt, J. L. Knight, L. Malaret, and N. L. Zuschlag (eds.). Vertebrate ecology and systematics - A tribute to Henry S. Fitch, pp 57-75. Museum of Natural History, The University of Kansas, Lawrence, KS.

Griffis, M. R., and R. G. Jaeger. 1998. Competition leads to an extinction-prone species of salamander: interspecific territoriality in a metapopulation. Ecology 79(7):2494-2502.

Griffiths, R. A. 1985. A simple funnel trap for studying newt populations and an evaluation of trap behavior in smooth and palmate newts, *Triturus vulgaris* and *T. helveticus*. Herpetological Journal 1:5-10.

Grothe, S. 1992. Red-tailed hawk predation on snakes: the effects of weather and snake activity. M.S. Thesis. Idaho State University, Pocatello, ID.

Gruber, B. 2004. Measuring activity of geckos with an automatic movement monitoring system. Herpetological Review 35:245-247.

Guillette, L. J., and M. P. Gunderson. 2001. Alterations in development of reproductive and endocrine systems of wildlife populations exposed to endocrine-disrupting contaminants. Reproduction 122:857-864.

Guillette, L. J., D. A. Crain, M. P. Gunderson, S. A. E. Kools, M. R. Milnes, E. F. Orlando, A. A. Rooney, and A. R. Woodward. 2000. Alligators and endocrine disrupting contaminants: a current perspective. American Zoologist 40:438-452.

Gunzburger, M. S. 2007. Evaluation of seven aquatic sampling methods for amphibians and other aquatic fauna. Applied Herpetology 4:47–63.

Gustafson, E. J., N. L. Murphy, and T. R. Crow. 2001. Using a GIS model to assess terrestrial salamander response to alternative forest management plans. Journal of Environmental Management 63:281-292.

Guyer, C., C. T. Meadows, S. C. Townsend, and L. G. Wilson. 1997. A camera device for recording vertebrate activity. Herpetological Review 28:135-140.

Guynn, D. C., Jr., R. L. Downing, and G. R. Askew. 1985. Estimating the probability of non-detection of low density populations. Cryptozoology 4:55-60.

Haenel, G. J., L. C. Smith, and H. B. John-Alder. 2003. Home-range analysis in *Sceloporus undulatus*. II. A test of spatial relationships and reproductive success. Copeia 2003:113-123.

Hairston, N. G. 1980. Species packing in the salamander genus *Desmognathus*: what are the interspecific interactions involved? American Naturalist 115:354-366.

Halliday, T. 1994. Marking amphibians by toe-clipping. FROGLOG 10(August):2-3.

Halliday, T. 1995. More on toe-clipping. FROGLOG 12 (March):2-3.

Halliday, T. R., and P. A. Verrell. 1988. Body size and age in amphibians and reptiles. Journal of Herpetology 22: 253-265.

Halpin, P. N. 1997. Global climate change and natural area protection: management responses and research directions. Ecological Applications 7:828–843.

Hanlin, H. G., and R. H. Mount. 1978. Reproduction and activity of the greater siren, *Siren lacertina* (Amphibia: Sirenidae), in Alabama. Journal of the Alabama Academy of Science 49:31-39.

Hanlin, H. G., F. D. Martin, L. D. Wike, and S. H. Bennett. 2000. Terrestrial activity, abundance and species richness of amphibians in managed forests in South Carolina. American Midland Naturalist 143(1):70-83.

Hardy, L. M. 2004. Genus *Syrrhophus* (Anura: Leptodactylidae) in Louisiana. Southwestern Naturalist 49:263-266.

Hare, A. 2006. Exotic lizard discovered in Kansas. Journal of Kansas Herpetology 19:9.

Harper, S. J., and G. O. Batzli. 1996. Monitoring use of runways by voles with passive integrated transponders. Journal of Mammalogy 77:364-369.

Harpole, D. N., and C. A. Haas. 1999. Effects of seven silvicultural treatments on terrestrial salamanders. Forest Ecology and Management 114:349–356.

Harris, R. N., R. A. Alford, and H. M. Wilbur. 1988. Density and phenology of *Notophthalmus viridescens dorsalis* in a natural pond. Herpetologica. 44:234-42.

Hartman, B. J. 1992. Regulations and enforcement of "taking" of gopher tortoises by development activities under the rules of the Florida Game and Fresh Water Fish Commission. Florida Game and Fresh Water Fish Commission, Tallahassee, Florida, USA.

Hawkins, C. P., L. J. Gottschalk, and S. S. Brown. 1988. Densities and habitat of tailed frog tadpoles in small streams near Mt. St. Helens following the 1980 eruption. Journal of the North American Benthological Society 7(3): 246-252.

Hayek, L. C. 1994. Analysis of amphibian biodiversity data. In W. R. Heyer, M. A. Donnellly, R. W. Mc Diarmid, L. C. Hayek and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp. 207-269. Smithsonian Institution Press, Washington DC.

Hayes, G. C., A. C. Broderick, B. J. Godley, P. Luschi, and W. J. Nichols. 2003. Satellite tracking suggests high-levels of fishing-induced mortality in marine turtles. Marine Ecology Progress Series 292:305-309.

Hayes, T. B., A. Collins, M. Lee, M. Mendoza, N. Noriega, A. A. Stuart, and A. Vonk. 2002. Hermaphroditic, demasculinized frogs after exposure to the herbicide atrazine at low ecologically relevant doses. Proceedings of the National Academy of Sciences (USA) 99:5476-5480.

Hazen, T. C., C. B. Fliermans, R. P. Hirsch, and G. W. Esch. 1978. Prevalence and distribution of *Aeromonas hydrophila* in the United States. Application of Environmental Microbiology 36(5):731-738.

Heatwole, H. 1989. The concept of the econe, a fundamental ecological unit. Tropical Ecology 30:13-19.

Heatwole, H. 2007. Quadrat sampling. In R. McDiarmid, M. Foster, C. Guyer, N. Chernoff, and J. W. Gibbons (eds.). Reptile Biodiversity – Standard Methods for Inventory and Monitoring. University of California Press. Berkeley. In Press.

Helfman, G. S. 1992. Underwater methods. *In* L. A. Nielson, and D. L. Johnson (eds.). Fisheries Techniques, pp. 349-369. Southern Printing Company, Inc., Blacksburg, Virginia.

Hellman, R. E., and S. R. Telford, Jr. 1956. Notes on a large number of red-bellied mudsnakes, *Farancia a. abacura*, from northcentral Florida. Copeia 1956:257-258.

Hels, T., and E. Buchwald. 2001. The effect of road kills on amphibian populations. Biological Conservation 99:331-340.

Hendrix, P. F., and P. J. Bohlen. 2002. Exotic earthworm invasions in North America: ecological and policy implications. BioScience 52:801-811.

Herbeck, L. A., and D. R. Larsen. 1999. Plethodontid salamander response to silvicultural practices in Missouri Ozark forests. Conservation Biology 13:623-632.

Herbst, L. H. 1994. Fibropapillomatosis of marine turtles. Annual Review of Fish Disease 4:389-425.

Hero, J. M. 1989. A simple code for toe clipping anurans. Herpetological Review 20:66-67.

Hess, G. 1996. Disease in metapopulation models: implications for conservation. Ecology 77:1617-1632.

Heyer, W. R. 1994. Thread bobbins. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp.153-155. Smithsonian Institution Press, Washington, DC. 364 pp.

Heyer, W. R., M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). 1994. Measuring and monitoring biological diversity: standard methods for amphibians. Smithsonian Institution Press, Washington, DC. 364 pp.

Himes, J. G. 2000. Burrowing ecology of the rare and elusive Louisiana pine snake, *Pituophis ruthveni* (Serpentes: Colubridae). Amphibia-Reptilia 22:91-101.

Hinton, T. G., P. D. Fledderman, J. E. Lovich, J. D. Congdon, and J. W. Gibbons. 1997. Radiographic determination of fecundity: is the technique safe for developing turtle embryos? Chelonian Conservation and Biology 2:409-414.

Hirth, H. F. 1966. The ability of two species of snakes to return to a hibernaculum after displacement. Southwestern Naturalist 11(1):49-53.

Hoffman, R. L., T. J. Tyler, G. L. Larson, M. J. Adams, W. Wente, and S. Galvan. 2005. Sampling protocol for monitoring abiotic and biotic characteristics of mountain ponds and lakes. U.S. Geological Survey Techniques and Methods 2-A2, 90 p.

Holycross, A. T. 1995. *Crotalus viridis viridis*. Phenology. Herpetological Review 26:37-38.

Hooge, P. N., and B. Eichenlaub. 1997. Animal movement extension to ArcView. Version 1.1. Alaska Biological Science Center, U.S. Geological Survey, Anchorage, Alaska, USA.

Hopkins W. A., C. L. Rowe, and J. D. Congdon. 1999. Elevated trace element concentrations and standard metabolic rate in banded water snakes, *Nerodia fasciata*, exposed to coal combustion wastes. Environmental Toxicology and Chemistry 18(6):1258-1263.

Houze, Jr., C. M., and C. R. Chandler. 2002. Evaluation of coverboards for sampling terrestrial salamanders in South Georgia. Journal of Herpetology 36(1):75-81.

Humphries, W. H. and T. K. Pauley. 2000. Seasonal changes in nocturnal activity of the hellbender, *Cryptobranchus a. alleganiensis* in West Virginia. Journal of Herpetology, 34(4):604-607.

Humphries, W. J. 2007. Diurnal seasonal activity of *Cryptobranchus alleganiensis* (Hellbender) in North Carolina. Southeastern Naturalist 6(1):135-140.

Humphries, W. J., and T. K. Pauley. 2005. Life history of the hellbender, *Cryptobranchus alleganiensis*, in a West Virginia stream. American Midland Naturalist 154(1):135-142.

Hurlbert, S. H. 1984. Pseudoreplication and the design of ecological field experiments. Ecological Monographs 54:187-211.

Hutton, J. M. 1986. Age determination of living Nile crocodiles from the cortical stratification of bone. Copeia 1986:332-341.

Inger, R. F. 1994. Microhabitat description. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp.60-66. Smithsonian Institution Press, Washington, DC. 364 pp.

Iverson, J. B. 1977. Geographic variation in the musk turtle, *Sternotherus minor*. Copeia 1977(3):502-517.

Iverson, J. B. 1979. Another inexpensive turtle trap. Herpetological Review 10:55.

Jackson, S. D. 2003. Ecological Considerations in the Design of River and Stream Crossings. In C. L. Irwin, P. Garrett, and K. P. McDermott (eds.). 2003 Proceedings of the International Conference on Ecology and Transportation. Raleigh, NC: Center for Transportation and the Environment, North Carolina State University. 10 pp.

Jacob, J. S., and C. W. Painter. 1980. Overwinter thermal ecology of *Crotalus viridis* in the north-central plains of New Mexico. Copeia 1980:799-805.

Jacobs, J. F., and W. R. Heyer. 1994. Collecting tissue for biochemical analysis. In W. D. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp. 299-301. Smithsonian Institution Press, Washington D.C.

Jaeger, R. G. 1994a. Patch sampling. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp.107-109. Smithsonian Institution Press, Washington, DC. 364 pp.

Jaeger, R. G. 1994b. Standard techniques for inventory and monitoring: transect sampling. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp.103-107. Smithsonian Institution Press, Washington, DC. 364 pp.

Jaeger, R. G., and R. F. Inger. 1994. Quadrat sampling. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp. 97-102. Smithsonian Institution Press, Washington, DC. 364 pp.

Jancovich, J. A., E. W. Davidson, E. F. Morado, J. L. Bertram, and J. P. Collins. 1997. Isolation of a lethal virus from the endangered tiger salamander *Ambystoma tigrinum stebbinsi*. Diseases of Aquatic Organisms 31:161-167.

Janzen, F. J. 1994. Climate change and temperature dependent sex determination in reptiles. Proceedings of the National Academy of Sciences (USA) 91: 7487–7490.

Jehle, R., and W. Hodl. 1998. PITs versus patterns: effects of transponders on recapture rate and body condition of Danube crested newts (*Triturus dobrogicus*) and common spadefoot toads (*Pelobates fuscus*). Herpetological Journal 8:181-186.

Jenkins, C. L., K. McGarigal, and L. R. Gamble. 2003. Comparative effectiveness of two trapping techniques for surveying the abundance and diversity of reptiles and amphibians along drift fence arrays. Herpetological Review 34(1):39-42.

Jennings, M. L., D. N. David, and K. M. Portier. 1991. Effect of marking techniques on growth and survivorship of hatchling alligators. Wildlife Society Bulletin 19(2):204-207.

Jensen, J. B., and C. D. Camp. 2003. Human exploitation of amphibians: direct and indirect impacts. In R. D. Semlitsch (ed.). Amphibian Conservation, pp. 94-112. Smithsonian Books, Washington, D. C.

Jensen, J. B., and C. Waters. 1999. The “spring lizard” bait industry in the state of Georgia, USA. Herpetological Review 30:20-21.

Jochimsen, D. 2006. Seasonal and landscape variation of snake road mortality on the upper Snake River Plain. Unpubl. M.S. Thesis, Idaho State University, Pocatello, Idaho.

Johnson, M. L., L. Berger, L. Philips, and R. Speare. 2003. Fungicidal effects of chemical disinfectants, UV light, desiccation and heat on the amphibian chytrid *Batrachochytrium dendrobatidis*. Diseases of Aquatic Organisms 57:255-260.

Johnson, P. T. J., K. B. Lunde, E. G. Ritchie, and A. E. Launer. 1999. The effect of trematode infection on amphibian limb development and survivorship. Science 284:802-804.

Johnson, S. A. 2003. Orientation and migration distances of a pond-breeding salamander (*Notophthalmus perstriatus*, *Salamandridae*). Alytes 21: 3-22.

Johnston, B., and L. Frid. 2002. Clearcut logging restricts the movements of terrestrial Pacific giant salamanders (*Dicamptodon tenebrosus* Good). Canadian Journal of Zoology-Revue Canadienne De Zoologie 80:2170-2177.

Jolly, G. M. 1965. Explicit estimates from capture-recapture data with both death and immigration-stochastic model. Biometrika 52:225-247.

Jones, G. S., L. A. Thomas, and K. Wong. 1995. Geographic distribution: *Ramphotyphlops braminus* (Braminy blind snake). Herpetological Review 26:210-211.

Jones, K. B. 1986. Amphibians and reptiles. In A. Y. Cooperrider, R. J. Boyd, and H. R. Stuart (eds.). Inventory and Monitoring of Wildlife Habitat, pp. 267-290. U.S. Bureau of Land Management Service Center. Denver, Colorado.

Jones, S. M., and G. W. Ferguson. 1980. The effect of paint marking on mortality in a Texas population of *Sceloporus undulatus*. Copeia 1980(4):850-854.

Jung, R. E., S. Droege, J. R. Sauer, and R. B. Landy. 2000. Evaluation of terrestrial and streamside monitoring techniques at Shenandoah National Park. Environmental monitoring and assessment 63:65-79.

Karesh, W. B., R. A. Cook, E. L. Bennett, and J. Newcomb. 2005. Wildlife trade and global disease emergence. Emerging Infectious Diseases 11:1000-1002.

Kauffeld, C. 1957. Snakes and Snake Hunting. Garden City, NY, Hanover House.

Keck, M. B. 1994. A new technique for sampling semi-aquatic snake populations. Herpetological Natural History 2(2):101-103.

Kendall, W. L., and J. D. Nichols. 1995. On the use of secondary capture-recapture samples to estimate temporary emigration and breeding populations. Journal of Applied Statistics 22:751-762.

Kendall, W. L., J. D. Nichols, and J. E. Hines. 1997. Estimating temporary emigration using capture-recapture data with Pollock's robust design. Ecology 78:563-578.

Kennett, R. M., and A. Georges. 1990. Habitat utilization and its relationship to growth and reproduction of the eastern long-necked turtle, *Chelodina longicollis* (Testudinata: Chelidae), from Australia. Herpetologica 46:22-33.

Kennish, M. J. (ed.). 1996. Practical Handbook of Estuarine and Marine Pollution. CRC Press, Boca Raton, FL.

Kéry, M. 2002. Inferring the absence of a species – a case study of snakes. Journal of Wildlife Management 66:330-338.

King, F.W. 1989. Conservation and Management. In Ross, C.A. (ed.) Crocodiles and Alligators, pp. 216-229. Facts on File, New York, NY.

Kingsbury, B. A., and C. J. Coppola. 2000. Hibernacula of the Copperbelly Water Snake (*Nerodia erythrogaster neglecta*) in Southern Indiana and Kentucky. Journal of Herpetology 34(2):294-298.

Kjoss, V. A., and J. A. Litvaitis. 2001. Community structure of snakes in a human-dominated landscape. Biological Conservation 98:285-292.

Klauber, L. M. 1931. A statistical survey of the snakes of the southern border of California. Bulletin of the Zoological Society of San Diego 8:1-93.

Klauber, L. M. 1939. Studies of reptile life in the arid southwest, Part 1. Night collecting on the desert with ecological statistics. Bulletin of the Zoological Society of San Diego 14:2-64.

Klauber, L. M. 1956. Rattlesnakes: their habits, life histories, and influence on mankind. Berkeley, University of California Press.

Klauber, L. M. 1972. Rattlesnakes: Their Habits, Life Histories and Influence on Mankind. Volumes 1 and 2. University of California Press, 2nd edition, Los Angeles.

Klawinski, P. D., R. K. Vaughan, D. Saenz, and W. Godwin. 1994. Comparison of dietary overlap between allopatric and sympatric geckos. Journal of Herpetology 28:225-230.

Kleopfer, J. D., S. H. Watson, and J. C. Mitchell. 2006. Geographic distribution: *Hemidactylus turcicus* (Mediterranean Gecko). Herpetological Review 37:106-107.

Kline, N. C., and D. E. Swann. 1998. Quantifying wildlife road mortality in Saguaro National Park. In G. L. Evink, P. Garrett, D. Zeigler and J. Berry (eds.), Proceedings of the 1998 International Conference on Wildlife Ecology and Transportation, pp. 23-31. Florida Department of Transportation, Tallahassee, FL.

Klinger, R. C., and J. A. Musick. 1995. Age and growth of loggerhead turtles (*Caretta caretta*) from Chesapeake Bay. Copeia 1995:204-209.

Klinger, R. C., R. H. George, and J. A. Musick. 1997. A bone biopsy technique for determining age and growth in sea turtles. Herpetological Review 28:31-32.

Koenig, J., R. Shine, and G. Shea. 2001. The ecology of an Australian reptile icon: how do blue-tongued lizards (*Tiliqua scincoides*) survive in suburbia? Wildlife Research 28:215-227.

Kraus, F. 2005. *Rana rugosa*. In M. Lannoo editor, Amphibian Declines: The Conservation Status of United States Species, pp. 580-581. University of California Press, Berkeley, CA. USA. 1094 pp.

Kraus, F. and F. Duvall. 2004. New records of alien reptiles and amphibians in Hawai'i. Bishop Museum Occasional Papers 79:62-64.

Krysko, K. L., and C. M. Sheehy, III. 2005. Ecological status of the ocellated gecko, *Sphaerodactylus argus argus* Gosse 1850, in Florida, with additional herpetological notes from the Florida Keys. Caribbean Journal of Science 41:169-172.

Krysko, K. L., and K. M. Enge. 2005. A new non-native lizard in Florida, the butterfly lizard, *Leiolepis belliana* (Sauria: Agamidae). Florida Scientist 68:247-249.

Krysko, K. L., A. N. Hooper, and C. M. Sheehy, III. 2003. The Madagascar giant day gecko, *Phelsuma madagascariensis grandis* Gray 1870 (Sauria: Gekkonidae): a new established species in Florida. Florida Scientist 63:222-225.

Krysko, K. L., K. M. Enge, and F. W. King. 2004. The veiled chameleon, *Chamaeleo calyptratus* Duméril and Bibron 1851 (Sauria: Chamaeleonidae): a new exotic species in Florida. Florida Scientist 67:249-253.

Kuchling, G. 1999. The Reproductive Biology of the Chelonia. Springer-Verlag, Berlin.

Kunz, T. H. 2001. Seeing in the dark: recent technological advances for the study of free-ranging bats. Bat Research News 42:91.

Kupferberg, S. J. 1997. Bullfrog (*Rana catesbeiana*) invasion of a California river: the role of larval competition. Ecology 78:1736-1751.

Lagler, K. F. 1943. Methods of collecting freshwater turtles. Copeia 1943(1):21-25.

Lamb, T., J. W. Bickham, T. B. Lyne, and J. W. Gibbons. 1995. The slider turtle as an environmental sentinel: multiple tissue assays using flow cytometric analysis. Ecotoxicology 4:5-13.

Lamoureux, V. S., and D. M. Madison. 1999. Overwintering habitats of radio-implanted green frogs, *Rana clamitans*. Journal of Herpetology 33:430-435.

Lancia, R. A., J. D. Nichols, and K. H. Pollock. 1994. Estimating the number of animals in wildlife populations. In T. A. Bookhout (ed.). Research and management techniques for wildlife and habitats. The Wildlife Society, Bethesda, Maryland.

Lang, C., M. Leuenberger, J. Schwander, and S. Johnson. 1999. 16ºC rapid temperature variation in central Greenland 70 000 years ago. Science 286:934–937.

Lang, J. W. 1987. Crocodilian behavior: implications for management. In G. J. W. Webb, S. C. Manolis, and P. J. Whitehead (eds.). Wildlife Management: Crocodiles and Alligators, pp. 273-294. Surrey Beatty & Sons Pty Limited, The Conservation Commission of the Northern Territory, Chipping Norton, NSW.

Langley, W. M., H. W. Lipps, and J. F. Theis. 1989. Responses of Kansas motorists to snake models on a rural highway. Transactions of the Kansas Academy of Science 92:43-48.

Lannoo, M. J. 1998. Status and Conservation of Midwestern Amphibians. University of Iowa Press, Iowa City. 507p.

Lannoo, M. J., and P. Nanjappa. 2005. *Dendrobates auratus*. In M. Lannoo editor, Amphibian Declines: The Conservation Status of United States Species, pp. 440-441. University of California Press, Berkeley, CA. USA. 1094 pp.

Legler, J. M. 1960. A simple and inexpensive device for trapping aquatic turtles. Utah Academy of Science: Proceedings 37:63-66.

Lemckert, F., and T. Brassil. 2000. Movements and habitat use of the endangered giant barred river frog (*Mixophyes iteratus*) and the implications for its conservation in timber production forests. Biological Conservation 96:177-184.

Lever, C. 2003. Naturalized reptiles and amphibians of the world. Oxford University Press, New York, NY, USA. 318 pp.

Lewke, R. E. 1974. Freeze branding as a method of marking snakes. Copeia 1974(4):997-1000.

Lieberman, S. 1994. Can CITES save the box turtle? Endangered Species Technical Bulletin. U.S. Department of the Interior Fish and Wildlife Service. Federal Register 19(5):15-17.

Lillywhite, H. B. 1982. Tracking as an aid in ecological studies of snakes. In N. J. Scott (ed.). Herpetological Communities: a Symposium of the Society for the Study of Amphibians and Reptiles and the Herpetologists' League, August 1977, pp. 181-191. U.S. Fish and Wildlife, Wildlife Research Report 13.

Limpus, C. J. 1984. Identification of ovarian follicles and oviductal eggs by cloacal examination of live Australian freshwater crocodiles, *Crocodylus johnstoni*. Australian Wildlife Research 11:203-204.

Limpus, C. J., and P. C. Reed. 1985. The green turtle, Chelonia mydas, in Queensland: a preliminary description of the population structure in a coral reef feeding ground. In G. Grigg, R. Shine, and H. Ehmann (eds.). Biology of Australasian Frogs and Reptiles, pp. 46-52. Surrey Beatty and Sons, Chipping Norton.

Lincoln, F. C. 1930. Calculating waterfowl abundance on the basis of banding returns. U.S. Department of Agriculture Circular 118:1-4.

Lindeman, P. V. 1996. Distribution, relative abundance, and basking ecology of the razorback musk turtle, *Kinosternon carinatum*, in the Pearl and Pascagoula River drainages. Herpetological Natural History 4:23-34.

Lindeman, P. V. 1997. A comparative spotting scope study of the distribution and relative abundance of river cooters (*Pseudemys concinna*) in western Kentucky and southern Mississippi. Chelonian Conservation and Biology 2:378-383.

Lindeman, P. V. 1998. Of deadwood and map turtles (*Graptemys*): An analysis of species status for five species in three river drainages using replicated spotting-scope counts of basking turtles. Chelonian Conservation and Biology 3(1):137-141.

Lips, K. R. 1991. Vertebrates associated with tortoise (*Gopherus polyphemus*) burrows in four habitats in south-central Florida. Journal of Herpetology 25:477-481.

Lloyd, M. L. 1998. Crocodilian Anesthesia. In R. E. Miller, M. E. Fowler, and S. Donley (eds.). Zoo and Wild Animal Medicine: Current Therapy, pp. 205-216. W. B. Saunders Company, Philadelphia.

Loafman, P. 1991. Identifying individual spotted salamanders by spot pattern. Herpetological Review 22:91-92.

Lohman, K. 2002. Annual variation in the density of stream tadpoles in a northern Idaho (USA) watershed. Verhandlungen der Internationalen Vereingung für Theoretische und Angewandte Limnologie 28:802-806.

Longcore, J. R., J. E. Longcore, A. P. Pessier, and W. A. Halteman. 2007. Chytridiomycosis widespread in anurans of northeastern United States. Journal of Wildlife Management 71(2):435-444.

Lotz, A., and C. R. Allen. 2007. Observer bias in anuran call surveys. Journal of Wildlife Management 71(2):675-679.

Lovich, J. E., and J. W. Gibbons. 1997. Conservation of covert species: protecting species we don't even know. In J. V. Abbema (ed.). Proceedings: Conservation, Restoration, and Management of Tortoises and Turtles – An International Conference, pp. 426-429. New York Turtle and Tortoise Society, New York, NY.

Lovich, J. E., S. W. Gotte, C. H. Ernst, J. C. Harshburger, A. F. Laemmerzahl, and J. W. Gibbons. 1996. Prevalence and histopathology of shell disease in turtles from Lake Blackshear, Georgia. Journal of Wildlife Diseases 32:259-265.

Lowe, W. H. 2003. Linking dispersal to local population dynamics: A case study using a headwater salamander system. Ecology 84(8): 2145-2154.

Lowe, W. H., and D. T. Bolger. 2002. Local and landscape-scale predictors of salamander abundance in New Hampshire headwater streams. Conservation Biology 16(1):183-193.

Luhring, T. M., and C. A. Jennison. 2008. A new stratified aquatic sampling technique for aquatic vertebrates. Journal of Freshwater Ecology. Vol. 23 (3):445-450.

Lutterschmidt, W. I., J. J. Lutterschmidt, and H. K. Reinert. 1996. An improved timing device for monitoring pulse frequency of temperature-sensing transmitters in free-ranging animals. The American Midland Naturalist 136:172-180.

Lutz, C. G., and J. L. Avery. 1999. Bullfrog culture. Southern Region Aquaculture Center Publication No. 436, March 1999.

Mac, M. J., P. A. Opler, C. E. P. Haecker, and P. D. Doran (eds.). 1998. Status and Trends of the Nation's Biological Resources. Volumes 1 and 2. U. S. Geological Survey, Reston, VI.

MacCulloch, R. D., and D. M. Gordon. 1978. A simple trap for basking turtles. Herpetological Review 9:133.

MacKenzie, D. I., J. D. Nichols, G. B. Lachman, S. Droege, J. A. Royle, and C. A. Langtimm. 2002. Estimating site occupancy when detection probabilities are less than one. Ecology 83:2248-2255.

MacKenzie, D. I., J. D. Nichols, J. E. Hines, M. G. Knutson and A. B. Franklin. 2003. Estimating site occupancy, colonization, and local extinction probabilities when a species is detected imperfectly. Ecology 84:2200-2207.

MacKenzie, D. I., L. L. Bailey, and J. D. Nichols. 2004. Investigating species co-occurrence patterns when species are detected imperfectly. Journal of Animal Ecology 73:546-555.

MacKenzie, D. I. and J. A. Royle. 2005. Designing occupancy studies: general advice and allocating survey effort. Journal of Applied Ecology 42:1105-1114.

MacKenzie D. I., J. D. Nichols, J. A. Royle, K. H. Pollock, L. L. Bailey, and J. E. Hines. 2006. Occupancy Estimation and Modeling: Inferring Patterns and Dynamics of Species Occurrence. Academic Press, Burlington, MA.

MacLean, G. S., A. K. Lee, and K. J. Wilson. 1973. A simple method of obtaining blood from lizards. Copeia 1973:338-339.

Mader, D. R. 1996. Reptile Medicine and Surgery. W. B. Saunders Company, Toronto, ON. 512 pp.

Maerz, J. C., B. Blossey, and V. Nuzzo. 2005. Green frogs show reduced foraging success in habitats invaded by Japanese knotweed. Biodiversity and Conservation 14:2901-2911.

Maerz, J. C., J. M. Karuzas, D. M. Madison, and B. Blossey. 2005. Introduced invertebrates are important prey for a generalist predator. Diversity and Distributions 11:83-90.

Magnusson, W. E., G. C. Grigg, and J. A. Taylor. 1978. An aerial survey of potential nesting areas of the saltwater crocodile, *Crocodylus porosus* Scheider, on the north coast of Arnhem Land, Northern Australia. Australian Wildlife Research 5(3):401-415.

Marsh, D. M., and P. C. Trenham. 2001. Metapopulation dynamics and amphibian conservation. Conservation Biology 15:40-49.

Marsh, H., and J. A. Saalfeld. 1989. Aerial surveys of sea turtles in the northern Great Barrier Reef Marine Park. Australian Wildlife Research 16:239-249.

Marshall, B. and F. I. Woodward (eds). 1985. Instrumentation for environmental physiology. Cambridge University Press, Cambridge.

Martof, B. S. 1953. Territoriality in the green frog, *Rana clamitans*. Ecology 34:165-174.

Mason, C. F. 1996. Biology of Freshwater Pollution. 3rd ed. Addison, Wesley Longman, Harlow.

Mathis, A. 1990. Territoriality in a terrestrial salamander: the influence of resource quality and body size. Behaviour 112:162-175.

Mazerolle, M. J. 2004. Amphibian road mortality in response to nightly variations in traffic intensity. Herpetologica 60:45-53.

Mazerolle, M. J., M. Huot, and M. Gravel. 2005. Behavior of amphibians on the road in response to car traffic. Herpetologica 61(4):380-388.

Mazerolle, M. J., L. L. Bailey, W. L. Kendall, J. A. Royle, S. J. Converse, and J. D. Nichols. 2007. Making great leaps forward: accounting for detectability in herpetological field studies. Journal of Herpetology 41(4):672-689.

Mazzotti, F. J. 1999. The American crocodile in Florida Bay. Estuaries 22(2B):552-561.

McArdle, B. H. 1990. When are rare species not there? Oikos 57:276-277.

McCarthy, M. A., and K. M. Parris. 2004. Clarifying the effects of toe clipping on frogs with Bayesian statistics. Journal of Applied Ecology 41:780-786.

McDiarmid, R. W. 1994a. Data standards. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp.57-60. Smithsonian Institution Press, Washington, DC. 364 pp.

McDiarmid, R. W. 1994b. Preparing amphibians as scientific specimens. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster. Measuring and monitoring biological diversity: standard methods for amphibians, pp. 289-297. Smithsonian Institution Press, Washington, DC.

McDiarmid, R. W., and M. A. Donnelly. 1994. Group activities and field trips. In W. Heyer, R. W. McDiarmid, M. Donnelly, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, p. 175-182. Smithsonian Institute Press, Washington, DC. 364pp.

McDowell, W. T., A. K. Wilson, and D. B. Clark. 2006. Geographic distribution: *Hemidactylus turcicus* (Mediterranean house gecko). Herpetological Review 37:495.

McFarlane, R. W. 1963. Disorientation of loggerhead hatchlings by artificial road lighting. Copeia 1963:153.

McKeown, S. 1996. A Field Guide to Reptiles and Amphibians in the Hawaiian Islands. Diamond Head Publishing, Inc. Los Osos, CA. USA. 172 pp.

Meffe, G. K., and A. L. Shelton. 1987. Habitat use by dwarf waterdogs (*Necturus punctatus*) in South Carolina streams, with life history notes. Herpetologica 43(4):490-496.

Meik, J. M., R. M. Jeo, J. R. Mendelson III, and K. E. Jenks. 2002. Effects of bush encroachment on an assemblage of diurnal lizard species in central Namibia. Biological Conservation 106:29-36.

Mendelson, J. R., III and W. B. Jennings. 1992. Shifts in the relative abundance of snakes in a desert grassland. Journal of Herpetology 26:38-45.

Meshaka, W. E., Jr. 1999a. The herpetofauna of the Kampong. Florida Scientist 62:153-157.

Meshaka, W. E., Jr. 1999b. The herpetofauna of the Doc Thomas house in South Miami, Florida. Florida Field Naturalist 27:121-123.

Meshaka, W. E., Jr. 2000. Colonization dynamics of two exotic geckos (*Hemidactylus garnotii* and *H. mabouia*) in Everglades National Park. Journal of Herpetology 34:163-168.

Meshaka, W. E., Jr. 2001. The Cuban Treefrog in Florida: Life History of a Successful Colonizing Species. University Press of Florida, Gainesville, FL, USA. 191 pp.

Meshaka, W. E., Jr. 2005a. *Eleutherodactylus planirostris*. In M. Lannoo editor, Amphibian Declines: The Conservation Status of United States Species, pp. 499-500. University of California Press, Berkeley, CA. USA. 1094 pp.

Meshaka, W. E., Jr. 2005b. *Osteopilus septentrionalis*. In M. Lannoo editor, Amphibian Declines: The Conservation Status of United States Species, pp. 463-464. University of California Press, Berkeley, CA. USA. 1094 pp.

Meshaka, W. E., Jr. 2006. An update on the list of Florida's exotic amphibian and reptile species. Journal of Kansas Herpetology.

Meshaka, W. E., Jr., and J. N. Layne. 2005. Habitat relationships and seasonal activity of the greenhouse frog (*Eleutherodactylus planirostris*) in southern Florida. Florida Scientist 68:35-43.

Meshaka, W. E., Jr., and K. G. Rice. 2005. The knight anole: ecology of a successful colonizing species in extreme southern mainland Florida. In W. E. Meshaka, Jr., and K. J. Babbitt, editors. Amphibians and Reptiles: Status and Conservation in Florida, pp. 225-230. Krieger, Malabar, FL, USA. 318 pp.

Meshaka, W. E., Jr., B. P. Butterfield, and J. B. Hauge. 2004. The Exotic Amphibians and Reptiles of Florida. Krieger, Melbourne, FL, USA. 166 pp.

Meshaka, W. E., Jr., R. D. Bartlett, Jr., and H. T. Smith. 2004b. Colonization success by green iguanas in Florida. Iguana 11:154-161.

Meshaka, W. E., Jr., S. D. Marshall, J. Boundy, and A. A. Williams. 2006a. Status and geographic expansion of the Mediterranean Gecko, *Hemidactylus turcicus*, in Louisiana: Implications for the southeastern United States. Herpetological Conservation and Biology Vol 1. pp. 46-51.

Meshaka, W. E., Jr., H. L. Cress, K. L. Kingsland, H. T. Smith, S. A. Fitchett, J. A. Moore, and E. M. Cowan. 2006b. *Hemidactylus* (house gecko) dynamics on south Florida buildings. Journal of Kansas Herpetology 17:8-9.

Meshaka, W. E., Jr., H. T. Smith, R. Severson, and M. A. Severson. 2005b. Spatial picture of a gecko assemblage in flux. Florida Scientist 68:53-55.

Meshaka, W. E., Jr., R. M. Engeman, H. T. Smith, C. L. Dean, J. A. Moore, and W. E. O'Brien. 2005a. The geographically contiguous and expanding coastal range of the northern curlytail lizard (*Leiocephalus carinatus armouri*) in Florida. Southeastern Naturalist, 4:521-526.

Messerre, M., and P. K. Ducey. 1998. Forest floor distribution of northern redback salamanders, *Plethodon cinereus*, in relation to canopy gaps: first year following selective logging. Forest Ecology and Management 107:319-324.

Metts, B. S., W. A. Hopkins, and J. P. Nestor. 2005. Interaction of an insecticide with larval density and pond-breeding salamanders (*Ambystoma*). Freshwater Biology 50:685-695.

Meylan, P. A., C. Steven, E. Dohm, and M. E. Barnwell. 1992. Observations on the turtle community of Rainbow Run, Marion Co., Florida. Florida Scientist 55:219-228.

Mills, M. S., C. J. Hudson, and H. J. Berna. 1995. Spatial ecology and movements of the brown water snake (*Nerodia taxipilota*). Herpetologica 51:412-423.

Millspaugh, J. J., and J. M. Marzluff. 2001. Radio tracking and animal populations. Academic Press: San Diego. 474pp.

Mitchell, J. C. 2000. Amphibian Monitoring Methods and Field Guide. Smithsonian National Zoological Park, Conservation Research Center: Front Royal, VA. 56 pp.

Mitchell, J. C., A. R. Breisch, and K. A. Buhlmann. 2006. Habitat Management Guidelines for Amphibians and Reptiles of the Northeastern United States. Partners in Amphibian and Reptile Conservation. Technical Publication HMG-3, Montgomery, Alabama. 106 pp.

Mitchell, J. C., B. W. Steury, K. A. Buhlmann, and P. P. van Dyk. 2007. Chinese softshell turtle (*Pelodiscus sinensis*) in the Potomac River and notes on eastern spiny softshells (*Apalone spinifera*) in Northern Virginia. Banisteria 30:41-43

Mittermeier, R. A., J. L. Carr, I. R. Swingland, T. B. Werner, and R. B. Mast. 1992. Conservation of Amphibians and Reptiles. In K. Adler (ed.) Herpetology: Current Research on the Biology of Amphibians and Reptiles, pp. 59-80. SSAR Publication, St. Louis, MO.

Mittermeier, R. A., N. Myers, and C. G. Mittermeier. 1999. Hotspots—Earth's Biologically Richest and Most Endangered Terrestrial Ecosystems. Toppan Printing Co. Japan. 431 pp.

Moa, J., D. E. Green, G. Fellers, and V. G. Chinchar. 1999. Molecular characterization of iridioviruses isolated from sympatric amphibians and fish. Virus Research 63(1999):42-52.

Mohr, J. R. and M. E. Dorcas. 1999. A comparison of anuran calling patterns at two Carolina Bays in South Carolina. Journal of the Elisha Mitchell Scientific Society 115:63-70.

Moler, P. E. 1992. Eastern indigo snake, *Drymarchon corais couperi*. In P.E. Moler (ed.). Rare and Endangered Biota of Florida, Vol. III. Amphibians and Reptiles, pp. 181-186. University of Florida Presses, Gainesville, Florida.

Moll, E. O., and J. M. Legler. 1971. The life history of a neotropical slider turtle, *Pseudemys scripta* (Schoepff), in Panama. Bulletin of the Los Angeles County Museum of Natural History Science 11:1-102.

Mooney, H. A., and E. E. Cleland. 2001. The evolutionary impact of invasive species. Proceedings of the National Academy of Sciences 98:5446-5451.

Morgan, J. A. T., V. T. Vredenburg, L. J. Rachowicz, R. A. Knapp, M. J. Stice, T. Tunstall, R. E. Bingham, J. M. Parker, J. E. Longcore, C. Moritz, C. J. Briggs, and J. W. Taylor. 2007. Population genetics of the frog-killing fungus *Batrachochytrium dendrobatidis*. Proceedings of the National Academy of the Sciences 104(34): 13845-13850.

Morreale, S. J., J. W. Gibbons, and J. D. Congdon. 1984. Significance of activity and movement in the yellow-bellied slider turtle (*Pseudemys scripta*). Canadian Journal of Zoology 62:1038-1042.

Morris, P. J., and A. C. Alberts. 1996. Determination of sex in white-throated monitors (*Varanus albigularis*), Gila monsters (*Heloderma suspectum*), and beaded lizards (*H. horridum*) using two-dimensional ultrasound imaging. Journal of Zoo and Wildlife Medicine 27:371-377.

Morris, P. J., L. A. Jackintell, and A. C. Alberts. 1996. Predicting the gender of subadult Komodo dragons (*Varanus komodoensis*) using two-dimensional ultrasound imaging and plasma testosterone concentration. Zoo Biology 15:341-348.

Moulis, R. A. 1997. Predation of the imported fire ant (*Solenopsis invicta*) on loggerhead sea turtle (*Caretta caretta*) nests on Wassaw National Wildlife Refuge, Georgia. Chelonian Conservation and Biology 2:433-436.

Moulton, C. A., W. J. Fleming, and B. R. Nerney. 1996. The use of PVC pipes to capture hylid frogs. Herpetological Review 27:186-187.

Mourao, G., M. Coutinho, R. Mauro, Z. Campos, W. Tomas, and W. Magnusson. 2000. Aerial surveys of caiman, marsh deer, and pampas deer in the Pantanal wetland of Brazil. Biological Conservation 92:175-183.

Mullican, T. R. 1988. Radio telemetry and fluorescent pigments - a comparison of techniques. Journal of Wildlife Management 52:627-631.

Murie, O. J. 1954. A Field Guide to Animal Tracks: The Peterson field guide series.

Murphy, B. R., and D. W. Willis. 1996. Fisheries Techniques. Second Edition. American Fisheries Society: Bethesda, Maryland.

Murphy, J. B., and T. T. Fendley. 1975. A new technique for live trapping of nuisance alligators. Proceedings of the Annual Conference of the Southeast Association of Game and Fish Commission 27:308-311.

Murphy, T. M. 1977. Distribution, movement, and population dynamics of the American alligator in thermally altered reservoir. M.S. Thesis, University of Georgia, Athens.

Murray, J. D., J. J. Bahen, and R. A. Rulifson. 1992. Management implications for By-catch in the North Carolina and Southeast Shrimp Fishery. Fisheries 17:21-26.

NAAMP Protocol. 2006. U. S. Geological Survey, North American Amphibian Monitoring Program. Retrieved March 14 2006 from the World Wide Web: http://www.pwrc.usgs.gov/NAAMP/protocol/index.html.

Nelson, D. H. and J. W. Gibbons. 1972. Ecology, abundance, and seasonal activity of the scarlet snake, *Cemophora coccinea*. Copeia 1972(3):582-584.

Nelson, D. H., and S. D. Carey. 1993. Range extension of the Mediterranean gecko (*Hemidactylus turcicus*) along the northeastern Gulf Coast of the United States. Northeast Gulf Science 13:53-58.

Nelson, G. L., and B. M. Graves. 2004. Anuran population monitoring: comparison of the North American Amphibian Monitoring Program's calling index with mark-recapture estimates for *Rana clamitans*. Journal of Herpetology 38:355-359.

Nickerson, M. A., and K. L. Krysko. 2003. Surveying for hellbender salamanders, *Cryptobranchus alleganiensis* (Daudin): A review and critique. Applied Herpetology 1(1-2):37-44.

Nickerson, M. A., and C. E. Mays. 1973. The hellbenders: North American "giant salamanders". Milwaukee Public Museum Publications in Biology and Geology. Vol 1. 106 pp.

Norden, A. W., and B. B. Norden. 1989. The Mediterranean gecko (*Hemdiactylus turcicus*) in Baltimore, Maryland. The Maryland Naturalist 33:57-58.

Northwest Marine Technology. 2000. Visible implant fluorescent elastomer (VIE) product information. www.nmt-inc.com.

Northwest Marine Technol¬ogy. 2007. Tagging reptiles and amphibians. Application Note APG02. 4 p. www.nmt.us.

Novotny, V., and G. Chesters. 1981. Handbook of Nonpoint Pollution: Sources and Management. Van Nostrand Reinhold Environmental Engineering Series. Van Nostrand Reinhold Co. New York.

Nussebaum, R. A., E. D. Brodie, and R. M. Storm. 1983. Amphibians and reptiles of the Pacific Northwest. University of Idaho Press, Moscow, Idaho.

O'Connor, M. P., and C. R. Tracy. 1992. Thermoregulation by juvenile woodhouse toads (*Bufo woodhousei)* in the laboratory and the field. Copeia 1992: 865-876.

Oliveira, M. E. and Martins, M. 2001. When and where to find a pit viper: activity patterns and habitat use of the lancehead, *Bothrops atrox*, in central Amazonia, Brazil. Herpetological Natural History 8(2):101-110.

Olson, D. H., W. P. Leonard, and R. B. Bury (eds.). 1997. Sampling amphibians in lentic habitats: Northwest Fauna No. 4, Olympia, Washington, Society for Northwestern Vertebrate Biology. 134 pp.

Oreskes, N. 2004. The scientific consensus on climate change. Science 306:1686.

Otis, D. L., K. P. Burnham, G. C. White, and D. R. Anderson. 1978. Statistical inference from capture data on closed animal populations. Wildlife Monographs 62. 135 pp.

Ott, J. A., and D. E. Scott. 1999. Effects of toe-clipping and PIT-tagging on growth and survival in metamorphic *Ambystoma opacum*. Journal of Herpetology 33(2):344-348.

Overton, W. S., and D. E. Davis. 1969. Estimating the numbers of animals in wildlife populations. In R. H. Giles, Jr. (ed.). Wildlife Management Techniques, pp.403-455. 3rd ed. The Wildlife Society, Washington, D.C.

Owens, D. W., and G. W. Ruiz. 1980. New methods of obtaining blood and cerebrospinal fluid from marine turtles. Herpetologica 36(1):17-20.

Parham, J. F., and G. R. Zug. 1997. Age and growth of loggerhead sea turtles (*Caretta caretta*) of coastal Georgia: An assessment of skeletochronological age estimates. Bulletin of Marine Science 61:287-304.

Parham, J. F., C. K. Dodd, Jr., and G. R. Zug. 1996. Skeletochronological age estimates for the Red Hills Salamander, *Phaeognathus hubrichti*. Journal of Herpetology 30: 401-404.

Parker, W. S., and W. S. Brown. 1973. Species composition and population changes in two complexes of snake hibernacula in northern Utah. Herpetologica 29(4):319-326.

Parmalee, J. R., and H. S. Fitch. 1995. An experiment with artificial shelters for snakes: effect of material, age, and surface preparation. Herpetological Natural History 3(2):187-191.

Pauley, T. K., and M. Little 1998. A new technique to monitor larval and juvenile salamanders in stream habitats. Banisteria 12:32-36.

Paulissen, M. A. 1986. A technique for marking teiid lizards in the field. Herpetological Review 17(1):16-17.

Pearcy, R. W., J. Ehleringer, H. A. Mooney, and P. W. Rundel. 1989. Plant Physiological Ecology:Field Methods and Instrumentation. Chapman and Hall, New York.

Pechmann, J. H. K., D. E. Scott, R. D. Semlitsch, J. P. Caldwell, L. J. Vitt, and J. W. Gibbons. 1991. Declining amphibian populations: The problem of separating human impacts from natural fluctuations. Science 253:892-895.

Pérez-Ponce de León, G., A. Jiménex-Ruiz, B. Mendoza-Garfias, and L. García-Prieto. 2001. Helminth parasites of garter snakes and mud turtles from several localities of the Mesa Central of Mexico. Comparative Parasitology 68:9-20.

Pérez-Ponce de León, G., L. García-Prieto, and U. Razo-Mendivil. 2002. Species richness of helminth parasites in Mexican amphibians and reptiles. Diversity and Distributions 8:211-218.

Peterson, B. L., B. E. Kus, and D. H. Deutschman. 2004. Determining nest predators of the Least Bell's Vireo through point counts, tracking stations, and video photography. Journal of Field Ornithology 75:89-95.

Peterson, C. C. 1994. Different rates and causes of high mortality in two populations of the threatened desert tortoise (*Gopherus agassizii*). Biological Conservation 70: 101-108.

Peterson, C. G. J. 1896. The yearly immigration of young plaice into Limfjord from the German sea. Report of the Danish Biological Station 6. Copenhagen, Denmark.

Peterson, C. L. 1987. Movement and catchability of the hellbender, *Cryptobranchus alleganiensis*. Journal of Herpetology 21(3):197-204.

Peterson, C. R. 1987. Daily variation in the body temperatures of free-ranging garter snakes. Ecology 68:160-69.

Peterson, C. R. and M. E. Dorcas. 1992. The use of automated data acquisition techniques in monitoring amphibian and reptile populations. In D. R. McCullough and R. H. Barrett (eds.). Wildlife 2001: Populations, pp. 369-378. Elsevier Applied Science, London.

Peterson, C. R., A. R. Gibson, and M. E. Dorcas. 1993. Snake thermal ecology: the causes and consequences of body temperature variation. In: Snakes: Ecology and Behavior. R.A. Seigel and J.T. Collins, (eds.). McGraw-Hill Publishing Company, New York, NY.

Peterson, C. R., and M. E. Dorcas. 1994. Automated data acquisition. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp.47-57. Smithsonian Institution Press, Washington, DC. 364 pp.

Peterson, R. C., and K. W. Cummins. 1974. Leaf processing in a woodland stream. Freshwater Biology 4:343-368.

Petranka, J. W. 1998. Salamanders of the United States and Canada. Smithsonian Institution Press, Washington, DC. 587 pp.

Petranka, J. W., M. P. Brannon, M. E. Hopey, and C. K. Smith. 1994. Effects of timber harvesting on low elevation populations of southern Appalachian salamanders. Forest Ecology and Management 67:135-147.

Phillips, K. 1994. Tracking the Vanishing Frogs—An Ecological Mystery. St. Martin's Press, New York. 244p.

Pilliod, D. S., and C. R. Peterson. 2000. Evaluating effects of fish stocking on amphibian populations in wilderness lakes. USDA Forest Service Proceedings RMRS-P-15-Vol. 5.

Pilliod, D. S., and C. R. Peterson. 2001. Local and landscape effects of introduced trout on amphibians in historically fishless watersheds. Ecosystems 4:322-333.

Pilliod, D. S., and E. Wind. 2008. Habitat Management Guidelines for Amphibians and Reptiles of the Northwestern United States and Canada. Partners in Amphibian and Reptile Conservation, Technical Publication HMG-4, Birmingham, AL. 139 pp.

Pimentel, D., S. McNair, J. Janecka, J. Wightman, C. Simmonds, C. O'Connell, E. Wong, L. Russel, J. Zern, T. Aquino, and T. Tsomondo. 2001. Economic and environmental threats of alien plant, animal, and microbe invasions. Agriculture, Ecosystems, and Environment 84:1-20.

Pisani, G. R. 1973. A guide to preservation techniques for amphibians and reptiles. Society for the Study of Amphibians and Reptiles, Herpetological Circular 1.

Platt, D. R. 1969. Natural history of the hognose snakes *Heterodon platyrhinos* and *Heterodon nasicus*. University of Kansas Publications Museum of Natural History 18(4):253-420.

Platz, C. C. J., G. Mengden, H. Quin, F. Wood, and J. Wood. 1980. Semen collection, evaluation and freezing attempts in the green sea turtle, Galapagos tortoise, and red-eared pond turtle. Annual Proceedings American Association of Zoo Veterinarians 1980:47-54.

Plummer, M. V. 2002. Observations on hibernacula and overwintering ecology of eastern hognose snakes (*Heterodon platirhinos*). Herpetological Review 33:89-90.

Pollock, K. H.1982. A capture-recapture design robust to unequal probability of capture. Journal of Wildlife Management 46:757-760

Pollock, K. H., and M. C. Otto. 1983. Robust estimation of population size in closed animal populations from capture-recapture experiments. Biometrics 39:1035-1049.

Pollock, K. H., J. D. Nichols, C. Brownie, and J. E. Hines. 1990. Statistical inference for capture-recapture experiments. Wildlife Monographs #107.

Poschadel, J. R., and D. Moller. 2004. A versatile field method for tissue sampling on small reptiles and amphibians, applied to pond turtles, newts, frogs, and toads. Conservation Genetics 5:865-867.

Pounds, J. A., and M. L. Crump. 1994. Amphibian declines and climate disturbance: the case of the golden toad and the harlequin frog. Conservation Biology 8:72-85.

Pounds, J. A., M. P. L. Fogden, and J. H. Campbell. 1999. Biological response to climate change on a tropical mountain. Nature 398:611-615.

Pounds, J. A., M. R. Bustamante, L. A. Coloma, J. A. Consuegra, M. P. L. Fogden, P. N. Foster, E. La Marca, K. L. Masters, A. Merino-Viteri, R. Puschendorf, S. R. Ron, G. A. Sanchez-Azofeifa, C. J. Still, and B. E. Young. 2006. Widespread amphibian extinctions from epidemic disease driven by global warming. Nature 439:161-167.

Prentice, E. F., T. A. Flagg, C. S. McCutcheon, D. F. Brastow. 1990. PIT-tag monitoring systems for hydroelectric dams and fish hatcheries. American Fisheries Society Symposium 7:317-322.

Pretzlaw, T., M. Huynh, L. Takats, and L. Wilkinson. 2002. Protocol for monitoring long-toed salamander (*Ambystoma macrodactylum*) populations in Alberta. Alberta Species at Risk Report No. 35. Alberta Fish and Wildlife Division. 26 pp.

Prior, K. A., and P. J. Weatherhead. 1996. Habitat features of black rat snake hibernacula in Ontario. Journal of Herpetology 30:211-218.

Punzo, F. 2001a. Diet composition of the rainbow whiptail, *Cnemidophorus lemniscatus* (Sauria: Teiidae), from southern Florida. Herpetological Review 32:85-87.

Punzo, F. 2001b. The Mediterranean gecko, *Hemidactylus turcicus*: life in an urban landscape. Florida Scientist 64:56–66.

Quinn, H., and J. P. Jones. 1974. Squeeze box technique for measuring snakes. Herpetological Review 5:35.

Quinn, T., M. Hayes, D. J. Dugger, T. L. Hicks, and A. Hoffman. 2007. Comparison of two tailed frog sampling methods in headwater streams. Journal of Wildlife Management 71(1): 282–288.

Ramirez-Pinilla, M., V. H. Serrano, and J. C. Galeano. 2002. Annual reproductive activity of *Mabuya mabouya* (Squamata, Scincidae). Journal of Herpetology 36:667-677.

Reading, C. J. 1997. A proposed standard method for surveying reptiles on dry lowland heath. Journal of Applied Ecology 34:1057-1069.

Ream, C., and R. Ream. 1966. The influence of sampling methods on the estimation of population structure in painted turtles. American Midland Naturalist 75:325-338.

Reaser, J. 1995. Marking amphibians by toe-clipping. FROGLOG 12(March):1-2.

Reed, J. M. 1996. Using statistical probability to increase confidence of inferring species extinction. Conservation Biology 10:1283-1285.

Reed, R. A., J. Johnson-Barnard, and W. L. Baker. 1996. Contribution of roads to forest fragmentation in the Rocky Mountains. Conservation Biology 10(4):1098-1106.

Reed, R. N. 2005. An ecological risk assessment of nonnative boas and pythons as potentially invasive species in the United States. Risk Analysis 25:753-766.

Reed, R. N. and M. E. Douglas. 2002. Ecology of the Grand Canyon rattlesnake (*Crotalus viridis abyssus*) in the Little Colorado River Canyon, Arizona. Southwestern Naturalist 47:30-39.

Reed, R. N., B. S. Yang, J. W. Gibbons, R. N. Fisher, and C. Rochester. 2004. Impacts of domestic and international trade on the conservation status of snakes native to the United States. U. S. Department of the Interior, U. S. Fish and Wildlife Service, Division of Scientific Authority. Unpublished report.

Reed, R. N., L. J. Trenkamp, B. E. Cox, and S. M. Sorensen. 2006. Geographic distribution: *Hemidactylus turcicus* (Mediterranean Gecko). Herpetological Review 37:106.

Regaud, C., and A. Policard. 1903. Recherches sur la structure du rein de quelques ophidians. Archives d Anatomie Microscopique 6:191-282.

Reinert, H. K. 1992. Radio-telemetric field studies of pitvipers: data acquisition and analysis. In J. A. Campbell and E. D. Brodie (eds). The Biology of Pitvipers, pp. 185-197. Selva, Tyler, Texas.

Reinert, H. K., and L. M. Bushar. 1991. A safe and simple method of blood collection from rattlesnakes. Herpetological Review 22:51-52.

Reinert, H. K., and R. T. Zappalorti. 1988. Timber rattlesnakes (*Crotalus horridus*) of the Pine Barrens: their movement patterns and habitat preference. Copeia 1988(4):964-978.

Renfrew, R. B., and C. A. Ribic. 2003. Grassland passerine nest predators near pasture edges identified on videotape. Auk 120:371-383.

Rexstad, E., and K. P. Burnham. 1991. Users Guide for Interactive Program CAPTURE. Colorado Cooperative Fish & Wildlife Research Unit, Colorado State University, Fort Collins, Colorado.

Rice, K. G., H. F. Percival, A. R. Woodward, and M. L. Jennings. 1999. Effects of egg and hatchling harvest on American alligators in Florida. Journal of Wildlife Management 63:1193-1200.

Richards, S. J., U. Sinsch, and R. A. Alford. 1994. Radio Tracking. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp.155-158. Smithsonian Institution Press, Washington, DC. 364 pp.

Richardson, K. C., G. J. W. Webb, and S. C. Manolis. 2002. Crocodiles: Inside Out. A guide to the crocodilians and their functional morphology. Surrey Beatty and Sons, Chipping Norton, NSW.

Rittenhouse, T. A. G., T. A. Altnether, and R. D. Semlitsch. 2006. Florescent powder pigments as a harmless tracking method for ambystomatids and ranids. Herpetological Review 37(2):188-191.

Rivas, J. A., and T. M. Avila. 1996. Sex determination in juvenile green iguanas (*Iguana iguana*) by cloacal analysis. Copeia 1996:219-221.

Roberts, K. A., and M. B. Thompson. 2003. Reconstructing Thermochron iButton's to reduce size and weight as a new technique in the study of small animal thermal biology. Herpetological Review 34:130-132.

Rodda, G. H. 1993. Where's Waldo (and the snakes)? Herpetological Review 24:44-45.

Rodda G. H., E. W. Campbell, and T. H. Fritts. 2001. A high validity census technique for herpetofaunal assemblages. Herpetological Review 32(1):24-30.

Rodda, G. H., and K. Dean-Bradley. 2002. Excess density compensation of island herpetofaunal assemblages. Journal of Biogeography 29:623-632.

Rodda, G. H., E. W. Campbell III, T. H. Fritts, and C. S. Clark. 2005. The predictive power of visual searching. Herpetological Review 36(3):259-264.

Rogers, A. R. 2001. Recent telemetry technology. In J. J. Millspaugh and J. M. Marzluff (eds.). Radio Tracking and Animal Populations. Pp. 79-121. Academic Press, San Diego, CA.

Rogers, A. R., and A. P. Carr. 1998. HRE: the home range extension for ArcView user's manual. Centre for Northern Forest Ecosystem Research, Ontario Ministry of Natural Resources, Canada.

Rossman, D. A. 1960. Herpetological survey of the Pine Hills area of southern Illinois. Quarterly Journal of the Florida Academy of Science 22:207-225.

Rostal, D. C., T. R. Robeck, D. W. Owens, and D. C. Kraemer. 1990. Ultrasound imaging of ovaries and eggs in Kemp's ridley sea turtles (*Lepidochelys kempii*). Journal of Zoo and Wildlife Medicine 21:27-35.

Rostal, D. C., J. S. Grumbles, V. A. Lance, and J. R. Spotila. 1994. Non-lethal sexing techniques for hatchling and immature desert tortoises (*Gopherus agassizii*). Herpetological Monographs 8:83-87.

Rudolph, D. C., S. J. Burgdorf, R. N. Conner, and R. R. Schaefer. 1999. Preliminary evaluation of the impact of roads and associated vehicular traffic on snake populations in eastern Texas. In G. L. Evink, P. Garrett, D. Zeigler, and J. Berry (eds.). Proceedings of the International Conference on Wildlife Ecology and Transportation, pp 129-136. Florida Department of Transportation, Tallahassee, Florida. Fl-ER-73-99.

Russell, K. R., H. G. Hanlin, T. B. Wigley, D. C. Guynn. 2002. Responses of isolated wetland herpetofauna to upland forest management. Journal of Wildlife Management 66(3):603-617.

Ryan, T. J., T. Philippi, Y. A. Leiden, M. E. Dorcas, T. B. Wigley, and J. W. Gibbons. 2002. Monitoring herpetofauna in a managed forest landscape: effects of habitat types and census techniques. Forest Ecology and Management 167:83-90.

Sagor, E. S., M. Ouellet, E. Barten, and D. M. Green. 1998. Skeletochronology and geographic variation in age structure in the wood frog, *Rana sylvatica*. Journal of Herpetology 32:469-474.

Saint Girons, H. 1982. Reproductive cycles of male snakes and their relationships with climate and female reproductive cycles. Herpetologica 38:5-16.

Saint Girons, H. 1985. Comparative data on lepidosaurian reproduction and some time tables. In C. Gans, and F. Billett (eds.) Biology of the Reptilia, pp. 35-58. Vol. 15. New York, John Wiley and Sons.

Sambrook, J., and D. W. Russell. 2001. Molecular Cloning: A Laboratory Manual, 3rd ed. Cold Spring Harbor Laboratory Press, Cold Spring Harbor, NY.

Samson, F., and F. Knopf. 1994. Prairie Conservation in North America. BioScience 44:418-421.

Sanzo, D., and S. J. Hecnar. 2006. Effects of road de-icing salt (NaCl) on larval wood frogs (*Rana sylvatica*). Environmental Pollution 140(2):247-256.

Savitzky, B. A., A. H. Savitzky, R. T. Belcher, and S. Ewers. 2002. Geographic distribution: *Ramphotyphlops braminus* (Brahminy blindsnake). Herpetological Review 33:150-151.

Schaefer, W. H. 1934. Diagnosis of sex in snakes. Copeia 1934:181.

Schildger, B. J., and R. Wicker. 1989. Sex determination and clinical examination in reptiles using endoscopy. Herpetological Review 20:9-10.

Schlaepfer, M. A., C. Hoover, and C. K. Dodd, Jr. 2005. Challenges in evaluating the impact of the trade in amphibians and reptiles on wild populations. Bioscience 55:256-264.

Schneider S. H., and T. L. Root. 1998. Climate Change. In M. J. Mac, P. A. Opler, C. E. P. Haecker, and P. D. Doran (eds.). Status and Trends of the Nation's Biological Resources, pp. 89-116. Vol 1.1. US Department of Interior, US Geological Survey, Reston, VA.

Schwarz, C. J., and G. A. F. Seber. 1999. Estimating animal abundance: review III. Statistical Science 14:427-456.

Schwarzkopf, L., and R. A. Alford. 1996. Dessication and shelter-site use in a tropical amphibian: comparing toads with physical models. Functional Ecology 10:193-200.

Scott, J. M., and P. J. Heglund, J. B. Haufler, M. Morrison, M. G. Raphael, and W. B. Wall. 2002. Predicting Species Occurrences: Issues of Accuracy and Scale. Island Press, Covelo, CA.

Scott, N. J., and B. D. Woodward. 1994. Surveys at breeding sites. In W. Heyer, R. W. McDiarmid, M. Donnelly, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp. 118-125. Smithsonian Institute Press, Washington, DC. 364 pp.

Sealy, J. B. 2002. Ecology and behavior of the timber rattlesnake (*Crotalus horridus*) in the upper Piedmont of North Carolina: Identified threats and conservation recommendations. In G. W. Schuett, M. Hoggren, M. E. Douglas, and H. W. Greene (eds.) Biology of the Vipers, pp.561-580. Eagle Mountain Publishing. Eagle Mountain, Utah.

Seber, G. A. F. 1965. A note on the multiple-recapture census. Biometrika 52:249-259.

Seber, G. A. F. 1973. The estimation of animal abundance and related parameters. Griffin, London.

Seigel, R. A., and M. A. Pilgrim. 2002. Long-term changes in movement patterns of massasaugas (*Sistrurus catenatus*). In G. W. Schuett, M. Hoggren, M. E. Douglas, and H. W. Greene (eds.) Biology of the Vipers, pp. 405-412. Eagle Mountain Publishing. Eagle Mountain, Utah.

Seigel, R. A., R. B. Smith, and N. A. Seigel. 2003. Swine flu or 1918 pandemic? Upper respiratory tract disease and the sudden mortality of gopher tortoises (*Gopherus polyphemus*) on a protected habitat in Florida. Journal of Herpetology 37:137–144.

Semlitsch, R. D. (ed.). 2003. Amphibian Conservation. Smithsonian Books, Washington, D. C.

Semlitsch, R. D. 1998. Biological determination of terrestrial buffer zones for pond-breeding salamanders. Conservation Biology 12:1113-1119.

Semlitsch, R. D., and J. R. Bodie. 2003. Biological criteria for buffer zones around wetlands and riparian habitats for amphibians and reptiles. Conservation Biology 17:1219-1228.

Semlitsch, R. D., K. L. Brown, and J. P. Caldwell. 1981. Habitat utilization, seasonal activity, and population size structure of the southeastern crowned snake *Tantilla coronata*. Herpetologica 37(1):40-46.

Seutin, G., B. N. White, and P. T. Boag. 1991. Preservation of avian blood and tissue samples for DNA analyses. Canadian Journal of Zoology 69:82-90.

Sever, D. M., and W. A. Hopkins. 2005. Renal sexual segment of the ground skink, *Scincella laterale* (Reptilia, Squamata, Scincidae). Journal of Morphology 266:26-59.

Sever, D. M., R. A. Stevens, T. J. Ryan, and W. C. Hamlett. 2002. Ultrastructure of the reproductive system of the black swamp snake (*Seminatrix pygaea*). III. Sexual segment of the male kidney. Journal of Morphology 252:238-254.

Sexton, O. J. 1959. Spatial and temporal movements of a population of the painted turtle, (*Chrysemys picta marginata*). Ecological Monographs 29:113-140.

Sexton, O. J., and S. R. Hunt. 1980. Temperature relationships and movements of snakes (*Elaphe obsoleta*, *Coluber constrictor*) in a cave hibernaculum. Herpetologica 36(1):20-26.

Shaffer, B., R. A. Alford, B. D. Woodward, S. J. Richards, R. G. Altig, and C. Gascon. 1994. Quantitative sampling of amphibian larvae. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and monitoring biological diversity: standard methods for amphibians, pp. 130-141. Smithsonian Institution Press. 364 pp.

Shaffer, H. B., and J. E. Juterbock. 1994. Night driving. In W. R. Heyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster (eds.). Measuring and Monitoring Biological Diversity: standard methods for amphibians, pp. 163-166. Smithsonian Books, Washington, DC.

Shea, G. M., and L. A. Reddacliff. 1986. Ossifications in the hemipenes of varanids. Journal of Herpetology 20:568-570.

Sherbrooke, W. C. 2002. Seasonally skewed sex-ratio of road-collected Texas horned lizards (*Phrynosoma cornutum*). Herpetological Review 33:21-24.

Shields, M. A. 1985. Selective use of pitfall traps by southern leopard frogs. Herpetological Review 16:14.

Shine, R. 1977. Reproduction in Australian elapid snakes I. Testicular cycles and mating seasons. Australian Journal of Zoology 25(4):647-653.

Shine, R., and M. Kearney. 2001. Field studies of reptile thermoregulation: how well do physical models predict operative temperatures? Functional Ecology: 15:282-288.

Shine, R., M. Lemaster, M. Wall, T. Langkilde, and R. Mason. 2004. Why did the snake cross the road? Effects of roads on movement and location of mates by garter snakes (*Thamnophis sirtalis parietalis*). Online report. www.ecologyandsociety.org. Vol. 9 (1), article 9.

Shine, R., M. M. Olsson, P. Harlow, I. T. Moore, M. P. LeMaster, and R. T. Mason. 2000. The transvestite serpent: Why do male garter snakes court (some) other males? Animal Behaviour 59:349-359.

Shine, R., M. J. Elphick, P. S. Harlow, I. T. Moore, M. P. LeMaster, and R. T. Mason. 2001a. Movements, mating and dispersal of red-sided garter snakes from a communal den in Manitoba. Copeia 2001:82-91.

Shine, R., M. P. LeMaster, I. T. Moore, M. M. Olsson, and R. T. Mason. 2001b. Bumpus in the snake den: effects of sex, size and body condition on mortality in red-sided garter snakes. Evolution 55:598-604.

Shirose, L. J., C. A. Bishop, D. M. Green, C. J. MacDonald, R. J. Brooks and N. J. Helferty. 1997. Validation tests of an amphibian call count survey technique in Ontario, Canada. Herpetologica 53:312-320.

Shoop, C. R. 1965. Orientation of *Ambystoma maculatum*: movements to and from breeding ponds. Science 149(3683):558-559.

Sibley, C. G., and J. E. Ahlquist. 1981. Instructions for specimen preservation for DNA extraction: a valuable source of data for systematics. Association of Systematics Collections Newsletter 9:44-45.

Silverman, S., and D. L. Janssen. 1996. Diagnostic imaging. In D. R. Mader (ed.). Reptile Medicine and Surgery, pp. 258-264. W. B. Saunders Company, Philadelphia.

Simon, C. A., and B. E. Bissinger. 1983. Paint marking lizards: does the color affect survivorship? Journal of Herpetology 17(2):184-186.

Skaar, J., K. Hegg, T. Moe, and K. Smedsurd. 1989. WMO international hygrometer intercomparison (Norway, 1987-1989). World Meteorological Organization, Geneva, Switzerland.

Skelly, D. K. 1996. Pond drying, predators, and the distribution of *Pseudacris tadpoles*. Copeia. 1996:599-605.

Skelton, C. and D. Parmley. 2005. Geographic distribution: *Anolis sagrei* (Brown anole). Herpetological Review 36:467.

Smith, L. L., and C. K. Dodd, Jr. 2003. Wildlife mortality on U.S. highway 441 across Paynes Prairie, Alachua County, Florida. Florida Scientist 66:128-140.

Smith, L. L., W. J. Barichivich, J. S. Staiger, K. G. Smith, and C. K. Dodd, Jr. 2006. Detection probabilities and occupancy estimates for amphibians at Okefenokee National Wildlife Refuge. American Midland Naturalist 155:149-161.

Smith, R. B., R. A. Seigel, and K. R. Smith. 1998. Occurrence of upper respiratory tract disease in gopher tortoise populations in Florida and Mississippi. Journal of Herpetology 32:426-430.

Snow, R. W., K. L. Krysko, K. M. Enge, L. Oberhoffer, A. Warren-Bradley, and L. Wilkins. 2007. Introduced populations of *Boa constrictor* (Boidae) and *Python molurus bivittatus* (Pythonidae) in southern Florida. Pp. 416-438. In Biology of the Pythons and Boas. R. W. Henderson and R. Powell (Eds.). Eagle Mountain Press. Eagle Mountain, UT. USA. 438 pp.

Sooter, C. A. 1955. Technique for bleeding snakes by cardiac puncture. Copeia 1955:254-255.

Sorensen, K. 2003. Trapping success and population analysis of *Siren lacertina* and *Amphiuma means*. Unpublished M.S. thesis, University of Florida, Gainesville, FL.

Sparling, D. W., C. A. Bishop, and G. Linder. 2000. The current status of amphibian and reptile ecotoxicological research. In D. W. Sparling, G. Linder, and C. A. Bishop (eds.). Ecotoxicology of Amphibians and Reptiles. Society of Environmental Toxicology and Chemistry, Pensacola, FL.

Stall, C. 1989. Animal Tracks of the Rocky Mountains: Idaho, Montana, Wyoming, Utah, Colorado, Arizona and New Mexico. Mountaineers Publishers. Seattle.

Stark, R. C., and S. F. Fox. 2000. Use of fluorescent powder to track horned lizards. Herpetological Review 31:230-231.

Stebbins, R. C. 2003. Western Reptiles and Amphibians. 3rd edition. Houghton Mifflin Co. Boston, MA. USA. 533 pp.

Steen, D. A., M. J. Aresco, S. G. Beilke, B. W. Compton, E. P. Congdon, C. K. Dodd, Jr., H. Forrester, J. W. Gibbons, J. L. Greene, G. Johnson, T. A. Langen, M. J. Oldham, D. N. Oxier, R. A. Saumure, F. W. Schueler, J. Sleeman, L. L. Smith, J. K. Tucker, and J. P. Gibbs. 2006. Relative vulnerability of female turtles to road mortality. Animal Conservation 9:269-273.

Stewart, M. C., D. F. Austin, and G. R. Bourne. 1993. Habitat structure and the dispersion of gopher tortoises on a nature preserve. Florida Scientist 56:70-81.

Stewart, M. M., and M. J. Lannoo. 2005. *Eleutherodactylus coqui*. 2005. In M. Lannoo editor, Amphibian Declines: The Conservation Status of United States Species, pp. 492-494. University of California Press, Berkeley, CA. USA. 1094 pp.

Stiles, J. H., and R. H. Jones. 1998. Distribution of the red imported fire ant, *Solenopsis invicta*, in road and powerline habitats. Landscape Ecology 335:335-346

Stokes, D., and L. Stokes. 1986. A guide to animal tracking and behaviour. Stokes Nature Guides. Little, Brown & Co, Boston, MA.

Stoner, D. 1925. The toll of the automobile. Science 61:56-57.

Stuart, S. N., J. S. Chanson, N. A. Cox, B. E. Young, A. S. L. Rodrigues, D. L. Fischman, R. W. Waller. 2004. Status and Trends of Amphibian Declines and Extinctions Worldwide. Science 306(5702):1783-1786.

Sullivan, B. K. Road running. In R. McDiarmid, M. Foster, C. Guyer, N. Chernoff, and J. W. Gibbons (eds.). Reptile Biodiversity – Standard Methods for Inventory and Monitoring. University of California Press. Berkeley. In Press.

Sutton, P. E., H. R. Mushinsky, and E. D. McCoy. 1999. Comparing the use of pitfall drift fences and cover boards for sampling the threatened sand skink (*Neoseps reynoldsi*). Herpetological Review 30(3):149-151.

Tanner, B. D. 1990. Automated weather stations. Remote Sensing Reviews 5:73-98.

Taylor, B. W., A. R. McIntosh, and B. L. Peckarsky. 2001. Sampling stream invertebrates using electroshocking techniques: implications for basic and applied research. Canadian Journal of Fisheries and Aquatic Sciences 58(3):437-445.

Taylor, D., and L. Winder. 1997. The use of imitation sand lizards to assess the accuracy of visual surveying techniques. Herpetological Journal 7:119-121.

Taylor, K. 1999. Rapid climate change. American Scientist 87:320–327.

Thomas, R. A. 1994. Geographic distribution: *Ramphotyphlops braminus* (Braminy blindsnake). Herpetological Review 25:34.

Thomas, R. B., D. W. Beckman, K. Thompson, K. A. Buhlmann, J. W. Gibbons, and D. L. Moll. 1997. Estimation of age for *Trachemys scripta* and *Deirochelys reticularia* by counting annual growth layers in claws. Copiea 1997:842-845.

Tinkle, D. W. 1957. Ecology, maturation, and reproduction of *Thamnophis sauritus proximus*. Ecology 38:69-77.

Tinkle, D. W. 1958. The systematics and ecology of the *Sternothaerus carinatus* complex (Testudinata, Chelydridae). Tulane Studies in Zoology 6:1-56.

Todd, B. D. and B. B. Rothermel. 2006. Assessing quality of clearcut habitats for amphibians: effects on abundances versus vital rates in the southern toad (*Bufo terrestris*). Biological Conservation 133(2):178-185.

Todd, B. D., B. B. Rothermel, R. N. Reed, T. M. Luhring, K. Schlatter, L. Trenkamp, and J. W. Gibbons. 2008. Habitat alteration increases invasive fire ant abundance to the detriment of amphibians and reptiles. Biological Invasions 10: 539-546.

Todd, B. D., and C. T. Winne. 2006. Ontogenetic and interspecific variation in timing of movement and responses to climatic factors during migrations by pond-breeding amphibians. Canadian Journal of Zoology 84: 715-722.

Todd, B. D., C. T. Winne, J. D. Willson, and J. W. Gibbons. 2007. Getting the drift: effects of timing, trap type, and taxon on herpetofaunal drift fence surveys. American Midland Naturalist 158 (2): 292-305.

Todd, M. J., R. R. Cocklin, and M. E. Dorcas. 2003. Temporal and spatial variation in anuran calling activity in the western Piedmont of North Carolina. Journal of the North Carolina Academy of Science 119:103-110.

Tozetti, A. M., and L. F. Toledo. 2005. Short-term movement and retreat sites of *Leptodactylus labyrinthicus* (Anura: Leptodactylidae) during the breeding season: a spool-and-line tracking study. Journal of Herpetology 39:640-644.

Tracy, C. R., and C. R. Tracy. 1995. Estimating age of desert tortoises (*Gopherus agassizii*) from scute rings. Copeia 1995:964-966.

Trauth, S. E., H. W. Robison, and M. V. Plummer. 2004. The Amphibians and Reptiles of Arkansas. University of Arkansas Press. Fayetteville, AR, USA. 421 pp.

Tuberville, T. D., and V. J. Burke. 1994. Do flag markers attract turtle nest predators? Journal of Herpetology 28:514-516.

Tuberville, T. D., J. R. Bodie, J. B. Jensen, L. LaClaire, and J. W. Gibbons. 2000. Apparent decline of the southern hog-nosed snake, *Heterodon simus*. The Journal of the Elisha Mitchell Scientific Society 116(1):19-40.

Tucker, A. D. 1997. Validation of skeletochronology to determine age of freshwater crocodiles (*Crocodylus johnstoni*). Marine and Freshwater Research 48:343-351.

Tucker, A. D., and C. J. Limpus. 1997. Assessment of reproductive status in Australian freshwater crocodiles (*Crocodylus johnstoni*) by ultrasound imaging. Copeia 1997:851-857.

Tucker, A. D., H. I. McCallum, and C. J. Limpus. 1997. Habitat use by *Crocodylus johnstoni* in the Lynd River, Queensland. Journal of Herpetology 31:114-121.

Turnbough, N. W. 2006. Geographic distribution: *Norops sagrei* (Brown anole). Herpetological Review 37:361.

Twitty, V. C. 1966. Of Scientists and Salamanders. Freeman, San Francisco, California.

Tyler, T., W. J. Liss, L. M. Ganio, G. L. Larson, R. L. Hoffman, E. Deimling, and G. Lomnicky. 1998. Interaction between introduced trout and larval salamanders (*Ambystoma macrodactylum*) in high-elevation lakes. Conservation Biology, 12(1):94-105.

Udevitz, M. S., and K. H. Pollock. 1991. Change-in-ratio methods for estimating population size. In D. R. McCullogh and R. H. Barrett (eds.). Wildlife 2001: Populations. Elsevier Applied Science, New York, NY.

Unrine, J. M., and C. H. Jagoe. 2004. Dietary mercury exposure and bioaccumulation in southern leopard frog (*Rana sphenocephala*) larvae. Environmental Toxicology and Chemistry 23(12):2956-2963.

Unrine, J. M., C. H. Jagoe, A. C. Brinton, H. A. Brant, and N. T. Garvin. 2005. Dietary mercury exposure and bioaccumulation in amphibian larvae inhabiting Carolina bay wetlands. Environmental Pollution 135(2):245-253.

Unrine, J. M., C. H. Jagoe, W. A. Hopkins, and H. A. Brant. 2004. Adverse effects of ecologically relevant dietary mercury exposure in southern leopard frog (*Rana sphenocephala*) larvae. Environmental Toxicology and Chemistry 23(12):2964-2970.

USDA-NRCS. 2001. National Resources Inventory. Retrieved March 2006 from the World Wide Web: http://www.nrcs.usda.gov/.

Vaughan, R. K., J. R. Dixon, and J. L. Cooke. 1996. Behavioural interference for perch sites in two species of introduced house geckos. Journal of Herpetology 30:46-51.

Vetas, B. 1950. Temperatures of entrance and emergence. Herpetologica 7:15-20.

Viitanen, P. 1967. Hibernation and seasonal movements of the vipers, *Vipera berus berus* (L.), in southern Finland. Annales Zoologici Fennici 4:472-546.

Vitt, L. J., and S. S. Sartorius. 1999. HOBOs, Tidbits, and lizard models: the utility of electronic devices in field studies of ectotherm thermoregulation. Function Ecology 13:670-674.

Vogt, R. C. 1980. New methods for trapping aquatic turtles. Copeia 1980:368-371.

Vogt, R. C. and R. L. Hine. 1982. Evaluation of techniques for assessment of amphibian and reptile populations in Wisconsin. In N. J. Scott, Jr. (ed.), Herpetological Communities, pp. 201-217. U.S. Department of Interior, Fish and Wildlife Service, Wildlife Research Report 13.

Vos, C. C., and A. H. P. Stumpel. 1996. Comparison of habitat isolation parameters in relation to fragmented distribution patterns in the tree frog (*Hyla arborea*). Landscape Ecology 11:203-214.

Vos, C. C., and J. P. Chardon. 1998. Effects of habitat fragmentation and road density on the distribution pattern of the moor frog *Rana arvalis*. Journal of Applied Ecology 35:44-56.

Waichman, A. V. 1992. An alphanumeric code for toe clipping amphibians and reptiles. Herpetological Review 23:19-21.

Wake, D. B. 1991. Declining amphibian populations. Science 253:860.

Walczak, J. T. 1991. A technique for the safe restraint of venomous snakes. Herpetological Review 22(1):17-18.

Waldron, J. L., C. K. Dodd, and J. D. Corser. 2003. Leaf litterbags: Factors affecting capture of stream-dwelling salamanders. Applied Herpetology 1:23-36.

Walker, Z., and G. Deichsel. 2005. Geographic distribution: *Podarcis muralis* (Common wall lizard). Herpetological Review 36:202.

Wallace, J. E. 2005. *Eleutherodactylus* (= *Syrrhophus*) *cystignathoides*. In M. Lannoo (ed.), Amphibian Declines: The Conservation Status of United States Species, pp. 494-495. University of California Press, Berkeley, CA, USA. 1094 pp.

Weary, G. C. 1969. An improved method of marking snakes. Copeia 1969(4):854-855.

Weatherhead, P. J. 1989. Temporal and thermal aspects of hibernation of black rat snakes (*Elaphe obsoleta*) in Ontario. Canadian Journal of Zoology 67:2332-2335.

Webb, G. J. W., R. Buckworth, and S. C. Manolis. 1983. *Crocodylus johnstoni* in the McKinlay River Area, N. T., III. Growth, movement and the population age structure. Australian Wildlife Research 10(2):383-401.

Webb, G. J. W., S. C. Manolis, and G. C. Sack. 1984. Cloacal sexing of hatchling crocodiles. Australian Wildlife Research 11:201-202.

Webb, R., D. Mendez, L. Berger, and R. Speare. 2007. Additional disinfectants effective against the amphibian chytrid fungus *Batrachochytrium dendrobatidis*. Diseases of Aquatic Organisms 74:13-16.

Weir, L. A., and M. J. Mossman. 2005. North American Amphibian Monitoring Program (NAAMP), p. 307-313. In M. J. Lannoo (ed.), Amphibian Declines: Conservation Status of United States Species. Univ. of California Press, Berkeley, CA, USA. 1094 pp.

Weir, L. A., J. A. Royle, P. Nanjappa and R. E. Jung. 2005. Modeling anuran detection and site occupancy on North American Amphibian Monitoring Program (NAAMP) routes in Maryland. Journal of Herpetology 39:627-639.

Wells, D. E. 1986. Guide to GPS positioning. Canadian GPS Association, Fredericton, Canada.

Welsh, H. H., L. M. Ollivier, and D. G. Hankin. 1997. A habitat-based design for sampling and monitoring stream amphibians with an illustration from the Redwood National Park. Northwest Naturalist 78(1): 1-16.

Whiles, M. R., K. R. Lips, C. M. Pringle, S. S. Kilham, R. J. Bixby, R. Brenes, S. Connelly, J. C. Colon-Gaud, M. Hunte-Brown, A. D. Huryn, C. Montgomery, and S. Peterson. 2006. The effects of amphibian population declines on the structure and function of Neotropical stream ecosystems. Frontiers in Ecology 4:27-34.

White, G. C., and K. P. Burnham. 1999. Program MARK: Survival estimation from populations of marked animals. Bird Study 46 Supplement, pp. 120-138.

White, G. C., and R. A. Garrott. 1990. Analysis of Wildlife Radio-tracking Data. Academic Press, San Diego, California.

White, G. C., K. P. Burnham, D. L. Otis, and D. R. Anderson. 1978. Users Manual for Program CAPTURE, Utah State University Press, Logan, Utah.

Wilcove, D. S., D. Rothstein, J. Dubow, A. Phillips, and E. Losos. 1998. Quantifying threats to imperiled species in the United States. Bioscience 48(8):607-615.

Wilhoft, D. C. 1963. Gonadal histology and seasonal changes in the tropical Australian lizard, *Leiolopisma rhomboidalis*. Journal of Morphology 113:185-204.

Williams, B. K., J. D. Nichols, and M. J. Conroy. 2002. Analysis and management of animal populations. Academic Press, 1040 pp.

Williams, R. D., G. E. Gates, and C. H. Holcutt. 1981. An evaluation of known and potential sampling techniques for hellbender, *Cryptobranchus allegainiensis*. Journal of Herpetology 15(1):23-27.

Willson, J. D., and M. E. Dorcas. 2003a. Effects of habitat disturbance on stream salamanders: Implications for buffer zones and watershed management. Conservation Biology 17(3):763-771.

Willson, J. D., and M. E. Dorcas. 2003b. Quantitative sampling of stream salamanders: comparison of dipnetting and funnel trapping techniques. Herpetological Review 34:128-130.

Willson, J. D., and M. E. Dorcas. 2004. A comparison of aquatic drift fences with traditional funnel trapping as a quantitative method for sampling amphibians. Herpetological Review 35:148-150.

Willson, J. D., C. T. Winne, and L. A. Fedewa. 2005. Unveiling escape and capture rates in aquatic snakes and salamanders (*Siren* spp. and *Amphiuma means*) in commercial funnel traps. Journal of Freshwater Ecology 20:397-403.

Willson, J. D., C. T. Winne, M. E. Dorcas, and J. W. Gibbons. 2006. Post-drought responses of semi-aquatic snakes inhabiting an isolated wetland: Insights on different strategies for persistence in a dynamic habitat. Wetlands 26:1071-1078.

Wilson, D. S., C. R. Tracy, and C. R. Tracy. 2003. Estimating age of turtles from growth rings: a critical review of the techniques. Herpetologica 59: 178-194.

Winn, B., J. B. Jensen, and S. Johnson. 1999. Geographic distribution: *Eleutherodactylus planirostris* (Greenhouse frog). Herpetological Review 30:49.

Winne, C. T. 2005. Increases in capture rates of an aquatic snake (*Seminatrix pygaea*) using naturally baited minnow traps: evidence for aquatic funnel trapping as a measure of foraging activity. Herpetological Review 36:411-413.

Winne, C. T., J. D. Willson, K. M. Andrews and R. N. Reed. 2006. Efficacy of marking snakes with disposable medical cautery units. Herpetological Review 37:52-54.

Winne, C. T., Willson, J. D., Todd, B. D., Andrews, K. M. and J. W. Gibbons. 2007. Enigmatic decline of a protected population of Eastern Kingsnakes, *Lampropeltis getula*, in South Carolina. Copeia 2007 (3): 507-519.

WMO. 1983. Guide to meteorological instruments and methods of observation. World Meteorological Organization No. 8, 5th edition, Geneva, Switzerland.

Wood, J. R., F. E. Wood, K. H. Critchley, D. E. Wildt, and M. Bush. 1983. Laparoscopy of the green sea turtle, *Chelonia mydas*. British Journal of Herpetology 6:323-327.

Wood, R. C., and R. Herlands. 1997. Turtles and Tires: the impact of roadkills on northern diamondback terrapin, *Malaclemys terrapin terrapin*, populations on the Cape May Peninsula, Southern New Jersey, USA. In Jim Van Abbema (ed.). Proceedings of the Conservation, Restoration and Management of Tortoises and Turtles – An International Conference, pp. 46-53. New York Turtle and Tortoise Society, New York, NY.

Woodbury, A. M. 1956. Uses of marking animals in ecological studies: marking amphibians and reptiles. Ecology 37(4):670-674.

Woodward, A. R., and C. T. Moore. 1992. Alligator age determination. Game and Fresh Water Fish Commission, Statewide Wildlife Research Final Performance Report Study No. 7563, Tallahassee, Florida, USA.

Wright, A. H. 2002. Life-histories of the frogs of Okefinokee Swamp, Georgia – North American Salientia (Anura) No. 2. Ithaca and London, Comstock Publishing Associates, Cornell University Press.

Wright K. M. 2001. Restraint techniques and euthanasia. In K. M. Wright and B. R. Whitaker (eds.). Amphibian Medicine and Captive Husbandry, pp. 111-128. Krieger Publishing Company Malabar, FL.

Wright, K. M., and B. R. Whitaker. 2001. Amphibian medicine and captive husbandry. Krieger, Malabar, Florida.

Wright, K., C. Pugh, L. Walker. 1995. Ultrasonographic sexing of helodermatid lizards. Proceedings of the American Association of Zoo Veterinarians: 239-242.

WWFC [World Wildlife Fund Canada]. 1988. Prairie Conservation Action Plan. World Wildlife Fund Canada, Toronto, Ontario.

Wygoda, M. L. 1979. Terrestrial activity of striped mud turtles, *Kinosternon baurii* (Reptilia, Testudines, Kinosternidae) in west-central Florida. Journal of Herpetology 13(4):469-480.

Wyman, R. L. 2003. Conservation of terrestrial salamanders with direct development. In R. D. Semlitsch (ed.). Amphibian Conservation, pp. 37-52. Smithsonian Books, Washington, D. C.

Yasuda, T., and N. Arai. 2005. Fine-scale tracking of marine turtles using GPS-Argos PTTs. Zoological Science 22:547-553.

Young, B. E., K. R. Lips, J. K. Reaser, R. Ibáñez, A. W. Salas, J. R. Cedeño, L. A. Coloma, S. Ron, E. La Marca, J. R. Meyer, A. Muñoz, F. Bolaños, G. Chaves, and D. Romo. 2001. Population declines and priorities for amphibian conservation in Latin America. Conservation Biology 15:1213-1223.

Zacharow, M., W. J. Barichivich, and C. K. Dodd, Jr. 2003. Using ground-placed PVC pipes to monitor hylid treefrogs: capture biases. Southeastern Naturalist 2(4):575-590.

Zappalorti, R. T., and H. K. Reinert. 1994. Artificial refugia as a habitat-improvement strategy for snake conservation. J. B. Murphy, K. Adler, and J. T. Collins (eds.). In Captive Management and Conservation of Amphibians and Reptiles, pp 369-375. Society for the Study of Amphibians and Reptiles, Ithaca NY. Contributions to Herpetology, Volume 11.

Zappalorti, R. T., and M. E. Torocco. 2002. A standardized protocol for sampling rare snakes in the New Jersey pine barrens: critical habitat assessment, survey techniques, and trapping methods. Herpetological Associates, Inc., New Jersey.

Zappalorti, R. T., E. W. Johnson, and Z. Lezcyznaki. 1983. The ecology of the northern pine snake (*Pituophis m. melanoleucus*) in southern New Jersey with special notes on habitat and nesting behavior. Bulletin of the Chicago Herpetological Society 18:57-72.

Zug, G. R., and A. S. Rand. 1987. Estimation of age in nesting female *Iguana iguana*: testing skeletochronology in a tropical lizard. Amphibia-Reptilia 8:237-250.

Zug, G. R., and J. F. Parham. 1996. Age and growth in leatherback turtles, *Dermochelys coriacea* (Testudines: Dermochelyidae): a skeletochonological analysis. Chelonian Conservation and Biology 2:244-249.

Zug, G. R., and R. E. Glor. 1998. Estimates of age and growth in a population of green sea turtles (*Chelonia mydas*) from the Indian River lagoon system, Florida: a skeletochronological analysis. Canadian Journal of Zoology 76:1497-1506.

Zug, G. R., M. M. Barber, and J. C. Dudley. 1982. Gonadal histology and reproduction in *Carlia bicarinata* (Scincidae, Sauria, Reptilia) of the Port Moresby area, Papua New Guinea. Herpetologica 38:418-424.

Zug, G. R., L. J. Vitt, and J. P. Caldwell. 2001. Herpetology. An Introductory Biology of Amphibians and Reptiles.

Zwickel, F. C., and A. Allison. 1983. A back marker for individual identification of small lizards. Herpetological Review 14(3):82-83.

ACKNOWLEDGEMENTS

A special thanks is given to the US Forest Service's Regional Fish and Wildlife Directors for their financial support and encouragement. Without their strong support, this project would not have been possible. We would also like to thank the Department of Defense for their significant contributions to this project. And last, but not least, we need to thank the Bureau of Land Management for their support through the pre-purchase of copies of the book.

Additional thanks go to three people who were integral in getting this book project off the ground, Ernie Garcia (USFWS), Kevin Leftwich (USFS), and Priya Nanjappa (AFWA). Ernie, Kevin, and Priya were vital to the initial planning stages of this book, acted as a continual support during the process, and their patience and endless enthusiasm for the project are greatly appreciated.

We also want to extend our appreciation to the following people for helpful comments on and reviews of drafts of this book and for providing technical assistance: George Bakken, Richard D. Bartlett, Colden V. Baxter, Art Beaubian, Steve Bennett, Dan Berger, Saul Berger, Andrew Blaustein, Jeff Boundy, Dave Bradford, Alvin L. Braswell, Al Breisch, Jim Brock, Angelo P. Bufalino, R. Bruce Bury, Gaylon Campbell, Steve Carey, Joseph T. Collins, John De Luisi, James R. Dixon, Kevin M. Enge, Lori Erb, Mark Ferguson, Jeff Foster, Ernie Garcia, Travis C. Glenn, Jim Godwin, Joel Green, Andrew Grosse, Hank Gruner, Craig Guyer, Cris Hagen, Scott Jackson, Chris Jenkins, John Jensen, Joanne Jerolman, Glenn Johnson, Bernard Kaiser, Wade Kalinowsky, Mark Kallgren, Michael S. Kellett, Karen E. Kinkead, John Kleopfer, Jim Knight, Mary Beth Kolozsvary, Fred Kraus, Kevin Leftwich, Leslie Long, Aaron Lotz, Tom Luhring, Mike Marchand, Dave Meek, Peter Meylan, Priya Nanjappa, Holly Niederriter, Michael O' Connor, Gretchen Padgett-Flohr, Tom Pauley, Charles R. Peterson, Chuck Peterson, David S. Pilliod, William Pitt, Sean Poppy, Warren Porter, Steven Price, Robert N. Reed, Barbara A. Savitzky, David Scott, Duncan Simpson, David Skelly, Scott Smith, Bert Tanner, Ed Thompson, Valorie Titus, Tracey Tuberville, Nathan W. Turnbough, Linda Weir, Alison Whitlock, Meredith Whitney, Mindy M. Wilkinson, and Elke Wind. This acknowledgement list includes people recognized by individual authors.

A special thank you goes to those that coordinated efforts in each of the five PARC regions for the *Species x Techniques Table*: J.D. Kleopfer (NE), Dede Olson (NW), Polly Conrad (SW), Kurt Buhlmann (SE), and Gary Casper (MW). Brett DeGregorio deserves particular thanks for consolidating vast amounts of contributed techniques text into the 50 coded techniques. This table would never have been completed without the assistance of many experts, and we want to thank the following persons for their contributions to the table: Tom Anton, Brian Aucone, Kurt Buhlmann, Janalee Caldwell, Carlos Camp, Jeff Camper, Gary Casper, Polly Conrad, Char Corkran, Brett DeGregorio, Andrew Durso, Nathan Engbrecht, Kevin Enge, Michael Forstner, Melissa Foster, J.

Tom Giermakowski, Jim Godwin, Dave M. Golden, Tim Gowan, Gabrielle Graeter, Sean Graham, Evan Grant, Andrew Grosse, Leigh Anne Harden, Justin Henningsen, Dennis Herman, Toby Hibbitts, Nathaniel Hilzinger, Kerry Holcomb, Kelly Irwin, Tina Jackson, Chris Jenkins, John Jensen, Kris Kendell, Bruce Kingsbury, J.D. Kleopfer, Travis LaDuc, Bradley Lambert, Lee Ann Linam, Lauren J. Livo, Kim Lovich, Robert Lovich, Stephen P. Mackessy, John Maerz, David Mauger, Aimee Mcintyre, Ann M. Mcluckie, Brian Metts, Joseph Milanovich, Paul Moler, Erin Muths, Kerry Nelson, Matthew Niemiller, Jen O'Conner, Dede Olson, Charles W. Painter, Tom Pauley, Todd Pierson, Jerry Reynolds, Jennifer Sevin, Jon Sjoberg, Laurie Vitt, Zack Walker, Linda Weir, Lori Williams, Steve Williams, J.D. Willson, and Krissy Wilson. In addition, we want to thank those individuals who made efforts to contribute to the PARC on-line submission form for these techniques: Joe Bartoszek, Kurt Buhlmann, Joshua M. Kapfer, Max A. Nickerson, Ryan P. O'Donnell, Brian E. Smith, Chris Swarth, Brian Todd, and Michael Welker.

We also want to thank everyone that contributed to the *Photo Vouchering Table*. The biggest thank you goes to John Jensen for spearheading the creation of this table and for coordinating amongst all the regional contributors to create a top quality resource. Many thanks go to those people that coordinated and ensured completion of their region's share of the table: John White (NE), J.D. Kleopfer (NE), Dede Olson (NW), Chris Jenkins (NW), Polly Conrad (SW), John Jensen (SE), and Gary Casper (MW). We would also like to thank several others that made significant contributions to this table: Tom Anton, Brian Aucone, Bruce Bury, Janalee Caldwell, Char Corkran, Michael Forstner, Tom Giermakowski, Sean Graham, James Harding, Selina Heppell, Toby Hibbitts, Tina Jackson, Travis LaDuc, Lee Ann Linum, Lauren Livo, Robert Lovich, Ann McLuckie, Donna Mobbs, Charlie Painter, Christopher Phillips, Chip Ruthven, Jennifer Sevin, Jon Sjoberg, Laurie Vitt, and Krissy Wilson.

Finally, a special thanks is extended to the following people who helped proof the final copy: Kimberly Andrews, David Brothers, John Byrd, Andrew Cantrell, Brett DeGregorio, Kevin Enge, Jennifer Evans, Dante' Fenolio, Barbara Foster, Michael Fulbright, Angela Getz, Linda Green, Andrew Grosse, Jeff Hall, Robert Hastings, Justin Henningsen, Robert Herrington, Chris Jenkins, John Jensen, Tanner Jessel, Rachel King, Joyce Klaus, James L. Knight, Mike Lannoo, Jessica McGuire, Kevin Messenger, Kristin Moore, Kathy Shelton, Greg Skupien, Steve Spear, Thilina Surasinghe, Bill Sutton, Tracey Tuberville, Kent Vliet, and Margaret Wead.

This book, and in particular, the *Species x Techniques Table*, could not have been accomplished without the assistance of many dedicated people. Thank you to all that were involved in making this book a reality!

Thank you for your interest in PARC's Technical Publication Series. For updates and additional information about Inventory and Monitoring techniques, visit the PARC website: www.parcplace.org.

Also, check out PARC's other publications:

All of these are available onine at

www.parcplace.org