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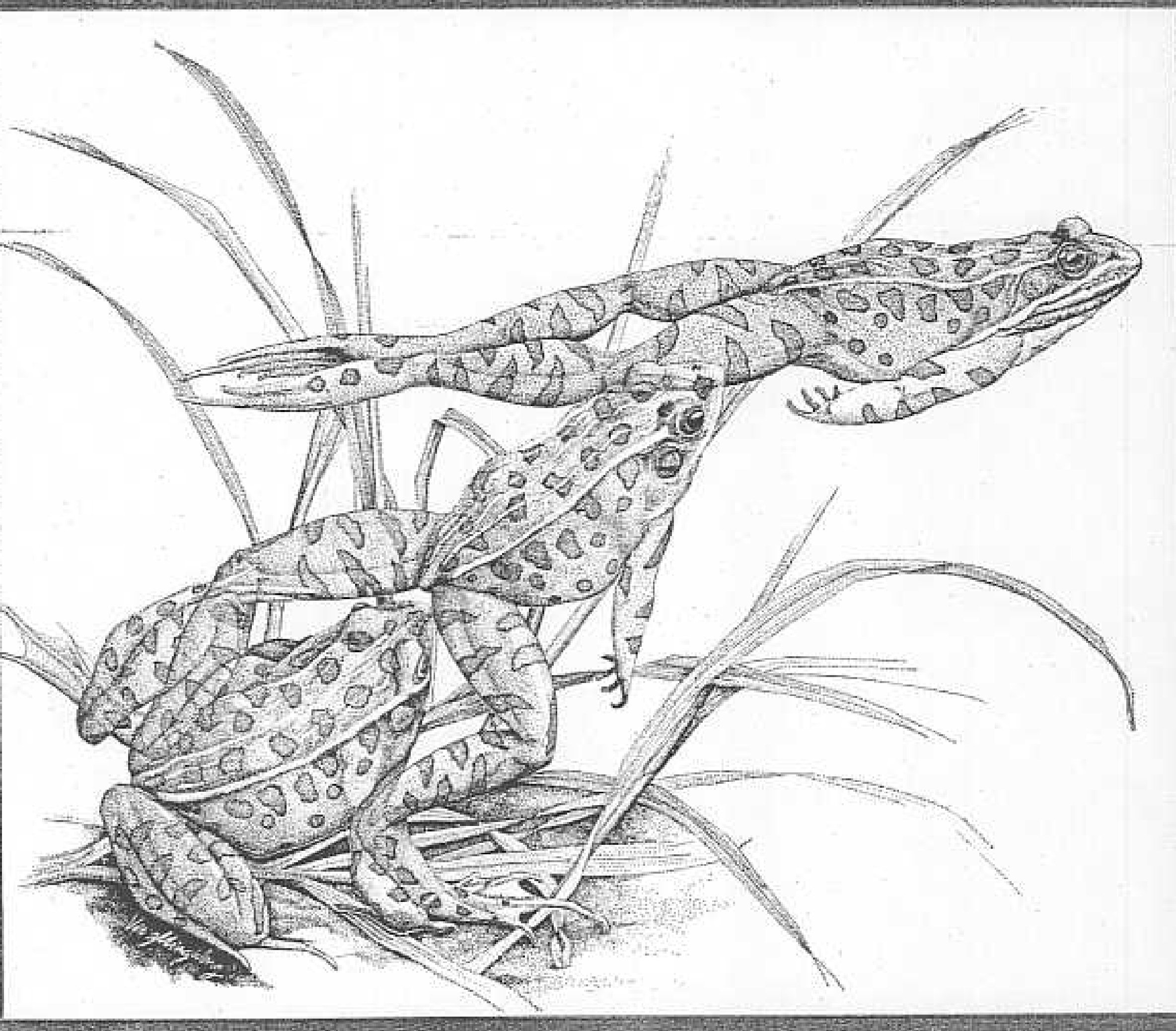

Field Herpetology

Methods for the Study of Amphibians and Reptiles in Minnesota

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Division of Comparative Biology

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Methods for the Study of Amphibians and Reptiles in Minnesota

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The mission of the Bell Museum is to encourage better understanding of the natural world through research, teaching, and public education in the natural sciences.

The Museum has three major goals: to encourage and conduct research in the natural sciences; to provide, with collections, a permanent record of biotic diversity in time and space; and to present and interpret the facts, principles, and controversies of the natural sciences to the public and the University community.

Staff

Cover illustration by Vera Wong

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PREFACE

This booklet is a practical guide to the field study of amphibians and reptiles in Minnesota. The emphasis is on materials and methods, using a "how-to" approach. I wrote it with several audiences in mind: weekend naturalists interested in expanding their natural history horizons, teachers looking for field-oriented exercises, and individuals, agencies, or institutions wishing to assess the herpetofauna of an area but having little background in herpetology. In general, the booklet is intended for anyone wanting to know more about amphibians and reptiles in their natural environment. It is *not* a field guide to the amphibians and reptiles of Minnesota, and contains no keys or species descriptions. It should be used as a companion to existing field guides (see Appendix III).

Furthermore, the booklet is not a comprehensive treatment of field techniques. I describe survey techniques for assessing the status of local amphibian and reptile populations, not more advanced topics such as methods of marking animals, diet analysis, or care of captive animals. This booklet will help you if you want to know the species present, their relative abundance, and their use of the available habitat. Appendix III lists books to consult on more sophisticated studies.

Chapter One is an introduction to herpetology, containing basic information on the biology of amphibians and reptiles with emphasis on the Minnesota herpetofauna. Chapter Two gives advice on how, when, and where to find and observe amphibians and reptiles. This chapter is intended as a practical guide for someone with little or no experience searching for amphibians and reptiles. Chapter Three is a survey of methods for the assessment of amphibian and reptile communities. I describe techniques for species inventory and discuss how to choose an appropriate technique based on cost/time effectiveness. This chapter is intended for someone faced with the problem of formally assessing the herpetofauna of a particular area and preparing a report on the findings. Chapter Four describes the methods for the preparation of herpetological study specimens for scientific study and teaching purposes. I also discuss the value of scientific collections and the appropriateness of collecting and preserving specimens.

The appendices provide specific information on herpetological resource people and institutions in Minnesota, a section on the legal status of amphibians and reptiles in Minnesota, and an annotated bibliography that provides a guide to the herpetological literature. A sample data sheet is also included.

Why did I write this booklet? Is there a need for it? There is a growing interest and concern about nongame species in Minnesota. Among vertebrates, the amphibians and reptiles are probably the most poorly studied and least appreciated group. For example, the Endangered Species Technical Advisory Committee (herpetology group) for the State of Minnesota faced an overwhelming problem in the preparation of its status report on the amphibians and reptiles of Minnesota: a lack of information on species distribution and abundance. One of the report's major recommendations was that special effort be made to collect more information on the state's herpetofauna to facilitate intelligent management.

Complementing the information problem, amphibians and reptiles face a serious "public relations" problem. Because they remain little known, they are often unfairly maligned. People are divided into two camps on amphibians and reptiles: herpetophiles (who find them fascinating) and herpetophobes (who find them disgusting). Unfortunately the majority are herpetophobes who consider the herpetophiles suspect and possibly deviant. I hope this booklet can improve this state of affairs.

A network of people who are in a position to increase our knowledge concerning Minnesota's amphibians and reptiles already exists. This network includes Department of Natural Resource personnel, county and municipal nature preserve employees, teachers interested in field biology, the Minnesota Herpetological Society, and amateur naturalists. Other interested individuals may be discouraged from attempting to collect data on amphibians and reptiles because they lack formal training. The scientific literature on herpetological field methods can be difficult for amateur naturalists to obtain or use, so I felt that a practical, informative, and readable guide was necessary. I hope this booklet helps bring these two fascinating groups of animals "out of the closet."

ACKNOWLEDGEMENTS

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I am especially indebted to the following people at the Bell Museum: Don Gilbertson (Director) for his support and encouragement; Kevin Williams (Curator of Exhibits) for overseeing the publication process; and Billy Goodman (Editor) for his editorial services. I thank John Slivon, Gerda Nordquist, Pamela Middleton, Mike Middleton, Ken Knouf, Dick Davis, and the Dunham family for assistance with photography. Barney Oldfield graciously supplied a number of herp photos. Bob Dorenfeld and Carlyn Iverson did the illustrations. I thank Jeff Lang, Dick Vogt, and Tom Anderson for reviewing the manuscript and for their helpful suggestions.

ONE
Introduction to Herpetology

Amphibians and Reptiles

The vertebrates (animals with backbones) are divided into five major taxonomic groups: fish, amphibians, reptiles, birds, and mammals. Note that three of these groups have their own designated fields of scientific study (fish = ichthyology; birds = ornithology; mammals = mammalogy), but that two are lumped together to form the subject matter of herpetology, the study of amphibians and reptiles. The term herpetology is derived from the Greek *herpeton* meaning creeping, and roughly translated as the study of crawling things. Though vague, this term is suitable enough—many amphibians and reptiles do indeed crawl along the ground.

Placing amphibians and reptiles into one scientific discipline implies that these two groups are more similar than the other vertebrate classes that warrant their own disciplines. In fact, amphibians and reptiles are quite different.

Amphibians have a thin, moist glandular skin without scales or claws. The skin is permeable to gas and water; amphibians absorb and lose water through the skin and breathe through it. Amphibians have a two-phase life cycle unique among vertebrates: a larval form undergoes metamorphosis into an adult form. The larvae have gills and may be free swimming or develop within the egg. Compared to reptilian eggs, amphibian eggs are susceptible to dessication and must be laid in water or damp surroundings. Amphibians are thus dependent on the availability of water.

By comparison, reptiles are truly terrestrial organisms. Their thick, scaly skin is an effective water barrier. Reptiles can lay their eggs in relatively dry locations because their eggs have extraembryonic membranes, which amphibian eggs lack. Reptiles do not have a gilled larval stage; the young emerge from the eggs as miniature adults. The major differences between amphibians and reptiles are summarized in Tables 1 and 2.

The other major groups of vertebrates are easily distinguished from the amphibians and reptiles by a few key characteristics. Fish are scaled vertebrates that breathe with gills and live and reproduce in water. Mammals have hair and nurse their young with milk. Birds have feathers, are capable of flight (with some exceptions), and lay eggs. All vertebrates have an internal skeleton with a backbone and a brain enclosed and protected by a skull.

Given their differences, why have amphibians and reptiles been lumped into one scientific discipline? Both groups are commonly found under objects, crawl along the ground, and have historically been considered distasteful creatures. The great 18th century taxonomist Linneaus used phrases like "foul and loathesome animals," "abhorrent," "filthy skin," "squalid habitation," and "offensive smell" in his scientific descriptions of amphibians and reptiles. A more practical reason for combining the groups is that similar methods of collection and preservation are used for both amphibians and reptiles and they are conveniently stored together in museums.

As a result of this historical liason, amphibians and reptiles are often referred to as a unit. This linkage creates an editorial problem because the phrase "amphibians and reptiles" becomes increasingly cumbersome and annoying with repeated usage. Alternatives do exist. The term "herpetofauna" can be used to refer to the assemblage of amphibians and reptiles in a given area. The synthetic term "herptile" is used by some

TABLE 1
Characteristics of Amphibians (Class Amphibia)

Main Taxonomic Groups (worldwide)*

CAECILIANS	Order Gymnophiona (Apoda)	150 species
SIRENS	Order Meantes (Trachystomata)	3 species
SALAMANDERS	Order Caudata (Urodela)	310 species
FROGS AND TOADS	Order Salienta (Anura)	2510 species

General Characteristics

Skin: Soft, thin glandular skin without scales.

Limbs: Most adult forms have limbs with digits. No true nails or claws on digits.

Reproduction: Internal or external fertilization. Egg surrounded by several gelatinous envelopes rather than a tough shell; egg does not have the extra embryonic membranes typical of reptiles, birds, and mammals.

Life Cycle: Most amphibian life cycles have two distinct phases: a larval form which undergoes metamorphosis into an adult. All amphibian larvae have external gills at some stage of their development. Some species have no free swimming larvae; larvae develop within and hatch from the egg.

Respiration: A variety of gas exchange surfaces may be utilized: gills, lungs, mouth and throat membranes, and skin.

Heart: Three-chambered heart.

Water Balance: Compared to reptiles, skin is thin and water permeable; evaporative water loss is high in most species.

Metabolic Rate: Low compared to birds and mammals.

Body Temperature Control: Ectothermy (environment is the source of body heat). Amphibians are not capable of maintaining stable body temperature by internal heat production.

Feeding: Carnivores as adults (one known exception, *Bufo marinus*). Carnivorous and herbivorous larvae.

Evolution: Evolved from primitive fish 350 million years ago. Amphibians were the first vertebrates on land and gave rise to the other terrestrial vertebrates. 250 million years ago they were a dominant group; today they are the smallest vertebrate class.

*(from Goin, Goin, and Zug, 1978)

TABLE 2
Characteristics of Reptiles (Class Reptilia)

Main Taxonomic Groups (worldwide)*

TURTLES	Order Testudines (Testudinata)	230 species
SQUAMATES	Order Squamata	
TUATARA	Suborder Sphenodontia	1 species
WORM LIZARDS	Suborder Amphisbaenia	140 species
SNAKES	Suborder Serpentes (Ophidia)	2700 species
LIZARDS	Suborder Sauria (Lacertilia)	3000 species
ALLIGATORS & CROCODILES	Order Crocodylia	21 species

General Characteristics

Skin: Tough, thick skin covered with scales.

Limbs: May or may not have limbs; if limbs are present, digits often have well developed true claws.

Reproduction: Internal fertilization. Egg surrounded by tough shell and extraembryonic membranes. Egg is dessication resistant and can be laid on land. Some species bear live young.

Life Cycle: Reptiles develop directly from the embryo into a miniature version of the adult form. There is no larval phase with external gills.

Respiration: Lungs are the most important gas exchange surface.

Heart: Three-chambered heart (four-chambered in crocodiles and alligators).

Water Balance: Compared to amphibians, the skin is tougher, thicker, and less water permeable. It is a very effective barrier against evaporative water loss.

Metabolic Rate: Low compared to birds and mammals.

Body Temperature Control: Ectothermy (environment is the source of body heat). Reptiles are not capable of maintaining high body temperatures by internal heat production.

Feeding: Most are carnivores; however, herbivory is not uncommon in turtles and some of the larger lizard species.

Evolution: Reptiles are the descendants of early amphibians and first appeared around 200 million years ago. They dominated the earth for some 200 million years and gave rise to the birds and mammals.

*(from Goin, Goin, and Zug, 1978)

herpetologists as a general term for both groups. In this booklet, I will often employ the term "herps" (singular = "herp") to refer to amphibians, reptiles, or the herpetofauna. The term herps has been widely accepted. The "-ing" form of this word describes the act of searching for herps. Thus, the herpetologist goes "herping," just as the ornithologist goes "birding."

On Being an Ectotherm ("Cold-Blooded")

Among the vertebrates, only fish, amphibians, and reptiles (with a few exceptions) are incapable of maintaining a high, constant body temperature by metabolic heat production. That is, they cannot use energy obtained from food to keep a high body temperature. The environment (the sun, a hot rock) is their only source of body heat. These animals are ectotherms (ecto = external; therm = heat), or, as they are sometimes misleadingly called, "cold-blooded." In contrast, birds and mammals are endotherms (endo = internal), or "warm-blooded," and maintain a high body temperature by metabolic heat production.

Two other useful terms describe control of body temperature. Homeothermy refers to the ability to maintain a constant body temperature (within a few degrees of some set point). Poikilothermy describes animals whose body temperature fluctuates by more than a few degrees from some set point. Note that these two terms refer to the *degree* of body temperature control, whereas endothermy and ectothermy refer to the *source* of heat used in body temperature control.

The terms "cold-blooded" and "warm-blooded" are commonly used to describe body temperature control. These terms, however, are inaccurate and should be avoided. A "cold-blooded" lizard can maintain a high body temperature by shuttling between sun and shade and by various physiological mechanisms. The temperature of an active desert lizard is often higher than a human's body temperature (37 °C). Calling this animal "cold-blooded" is misleading; many ectotherms maintain high, constant body temperatures, and are thus homeothermic, for much of their activity periods.

On the other hand, some "warm-blooded" animals (such as bats) allow their body temperature to fluctuate considerably from their normal active temperature when they rest during the day or during the winter. This relaxation of body temperature control appears to be an energy saving adaptation. There are many endotherms that, like bats, are poikilothermic (fluctuating body temperatures) on a daily or seasonal basis. Humans maintain a high, almost constant body temperature; we are homeothermic endotherms. Thus, we can scarcely imagine the world of an ectotherm, dependent on its surroundings for body heat.

There are advantages and disadvantages to both methods of temperature control. Endotherms can be active over a wide range of environmental conditions; ectotherm activity is restricted to seasons and habitats with favorable thermal conditions. Endothermy allows for biochemical efficiency; the endotherm's physiology can be "designed" to work at one specific temperature. Ectothermic physiology must deal with temperature fluctuations. Endothermy, coupled with an efficient cardiovascular system, allows for sustained high levels of activity. Amphibians and reptiles, in contrast, do not have much stamina.

Countering these advantages of endothermy is one major disadvantage: endothermy is energetically very expensive. About 85 percent of the energy budget of an endotherm goes to maintaining its high, constant body temperature; an ectotherm does not have this cost. This means that a great deal more of the energy that an ectotherm obtains from its food can go to growth, reproduction, and other functions. Ectotherms can also go for long periods of time without food. They do not have to continuously supply fuel (food) to their "metabolic furnace" to maintain body temperature. Ectotherms have a low-energy lifestyle compared to endotherms; they are fuel-efficient Hondas compared to gas-guzzling Cadillacs. Thus, it is incorrect to think of ectothermic amphibians and reptiles as being somehow physiologically inferior to endotherms. They are just different. Pough (1983) presents a fascinating discussion of amphibians and reptiles as low-energy systems.

Minnesota Amphibians

Two of the four main taxonomic groups of amphibians are found in Minnesota: salamanders and anurans (frogs and toads).

Salamanders

Five species of salamanders belonging to four families (Ambystomatidae, Plethodontidae, Proteidae, Salamandridae) are found in Minnesota. A tail plus two pairs of legs make salamanders superficially similar in appearance to lizards (which are reptiles) and some people confuse these two groups. Lizards can easily be distinguished from salamanders by their scales and external ear openings. Salamanders differ from frogs and toads in that the tail is retained throughout life and the head and trunk are distinct.

Salamanders are also unlike frogs and toads in their manner of reproduction. Salamanders do not use vocalizations as part of their breeding behavior; instead color, ritualized movements, and chemical secretions all play a role in their mating activities. The males of all Minnesota species deposit spermatophores (sperm packets), which are picked up by females using their cloacae (urogenital openings); fertilization then occurs inside the female's body.

Minnesota salamanders exhibit an interesting diversity of ecological "lifestyles." Several species (blue-spotted salamander, tiger salamander, central newt) exhibit the familiar amphibian life cycle: eggs laid in water, embryonic development and hatching, aquatic larval stage, and metamorphosis into an adult form. Most larvae metamorphose in the latter part of the summer in which they were laid as eggs. Depending on the species and local conditions, some individuals overwinter as larvae and metamorphose their second summer. Adult blue-spotted and tiger salamanders are terrestrial except during the aquatic breeding period. Some species, such as the central newt, may metamorphose into an aquatic form (newt) lacking gills that is capable of reproducing or into a nonreproductive terrestrial form (eft). The eft can metamorphose into a newt at a later time to reproduce. These transformations are controlled by local ecological conditions such as the availability of suitable aquatic breeding and feeding habitat. (Fig. 1)

The mudpuppy is completely aquatic. This species looks like a larval salamander throughout its life and never loses its gills. At the other extreme, the red-back salamander is terrestrial and even lays its eggs on land in a damp site. There is no aquatic larval stage in this species. All development occurs in the egg, and the young red-back emerges from the egg as a miniature version of the adult.

Free-swimming salamander larvae are carnivores. They initially eat small aquatic invertebrates and can handle larger prey such as tadpoles and other salamander larvae as they grow larger. Adult salamanders eat a variety of invertebrates. Mudpuppies are known to occasionally eat fish and fish eggs.

Many salamanders secrete noxious fluids from glands in their skin. If you have ever handled a tiger salamander you know they are capable of secreting copious amounts of unpleasant, sticky fluid. These secretions make salamanders distasteful to many predators. For example, raccoons and painted turtles will quickly reject red-back salamanders in experimental "taste tests." Salamanders are not fully protected by this chemical defense and some mammals, birds, and snakes do feed on them readily.

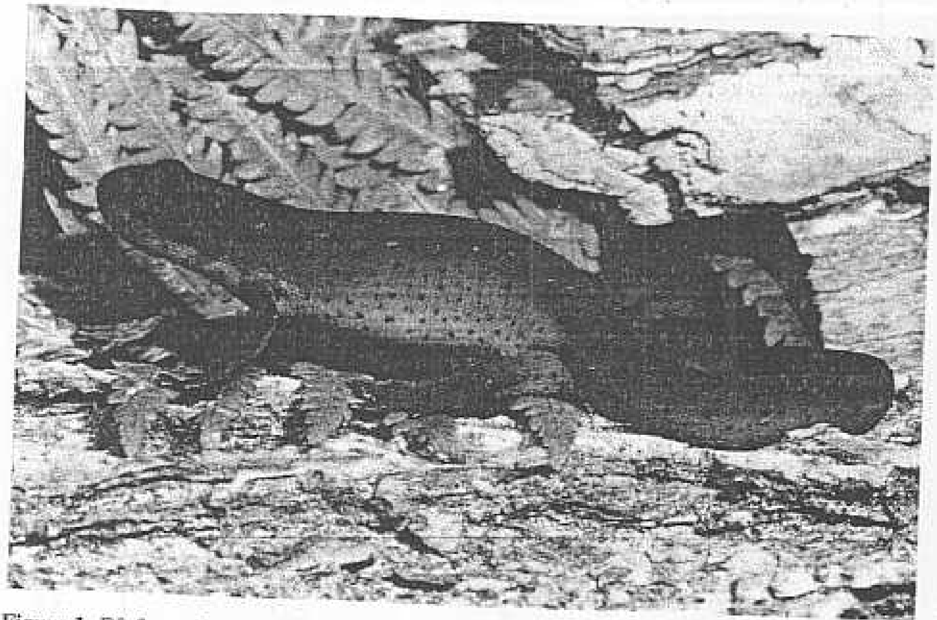

Figure 1. Eft form of central newt from the Chippewa River in Wisconsin. Central newt larvae have two life history options: they may either metamorphose and leave the pond as land-dwelling efts, or they may stay in the water as aquatic newts. The eft stage may last two to three years. Central newts can switch between newt and eft stages during their lifetime. Photo by Barney Oldfield.

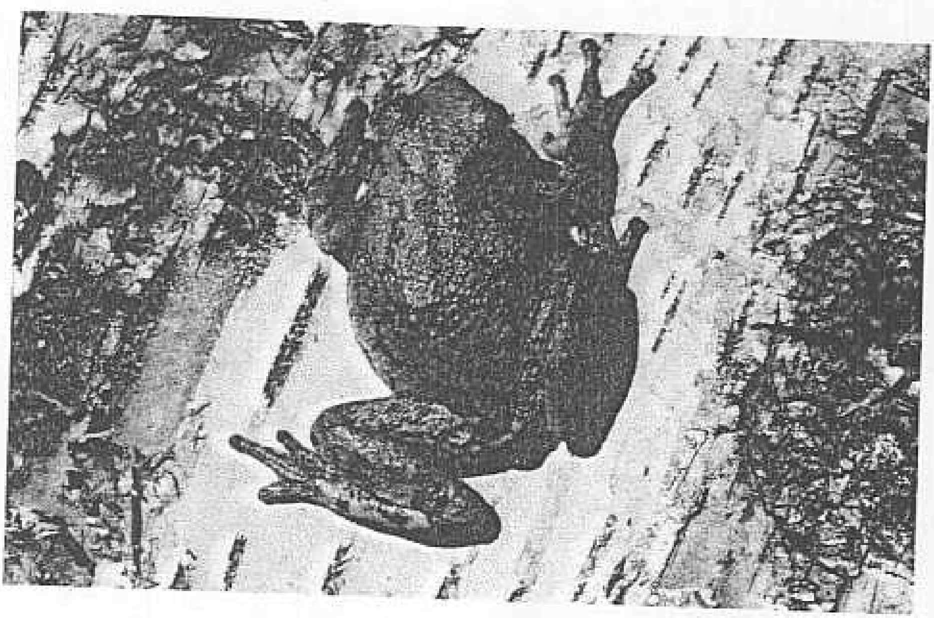

Figure 2. The gray treefrog (pictured) and Cope's gray treefrog cannot be reliably distinguished by appearance, but they have distinctive calls.

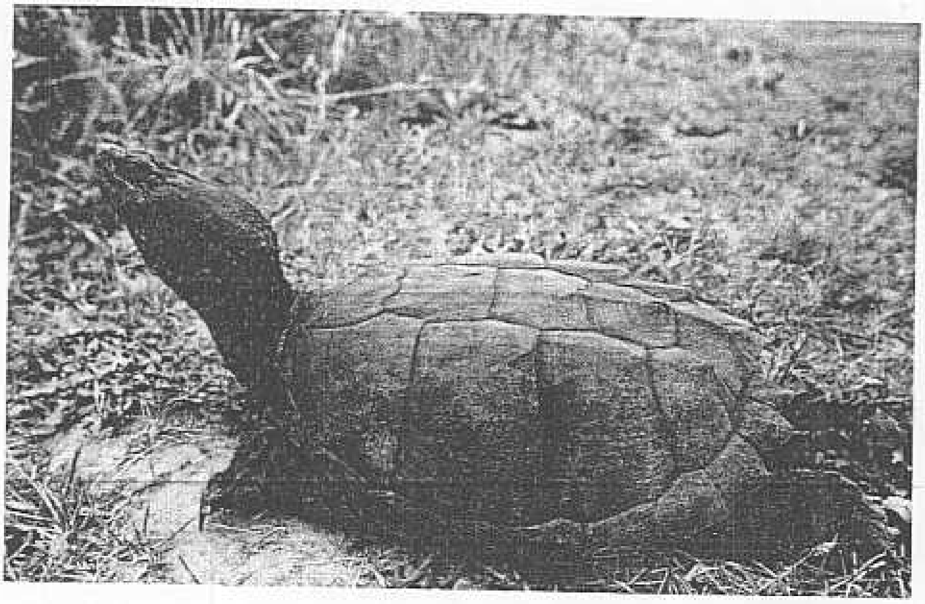

Figure 3. Snapping turtles are common throughout Minnesota. Heavy commercial trapping can harm local populations because snappers take five to seven years to become sexually mature. These turtles are extremely pugnacious on land and should be treated with respect. Photo by Barney Oldfield.

Figure 4. Northern prairie skink from Nobles County. These skinks are secretive burrowers that prefer dry, open areas with grass, sand, or gravel surfaces. They are found throughout Minnesota, except the northeast, and may be common locally though rarely noticed. Photo by Barney Oldfield.

Anurans

Fourteen species of frogs and toads belonging to three families (Bufonidae, Hylidae, and Ranidae) are found in Minnesota. Anurans lack tails, their heads and bodies merge without a pronounced neck, and their legs are well developed for hopping or leaping (Fig. 2). It is difficult to confuse them with other animals. Anurans can be conveniently classified into general categories on the basis of appearance: treefrogs (climbing forms with enlarged toepads), toads (squat, hopping forms with warty skins), and the classic frog (streamlined bodies with large powerful hindlimbs for jumping). It is technically correct to refer to any anuran as a frog.

Minnesota anurans vary in their dependence on water. Some species, such as the mink frog and bullfrog, are never found far from permanent bodies of water. Others are more terrestrial (e.g., wood frog, northern leopard frog, American toad); they will be found at breeding pools for short periods in the spring, but disperse to summer feeding areas and often are found far from the nearest body of water.

Unlike the silent salamanders, male frogs vocalize during their breeding activities. The male's call sends several messages to other frogs. It indicates what kind of frog he is (his species), advertises his presence to males and females of the same species, and says that he is looking for a mate. Each species has a unique call and generally only individuals of that species respond to it. Mixups do occur, however; in certain areas, individuals of different closely related species do mate and even produce hybrid offspring. This is not common and most encounters between different species are doomed to failure. I observed a spring peeper attempting to mate with a much larger wood frog. These star-crossed lovers were obviously wasting time and effort. Species vary as to the commencement and duration of the breeding season and the calls are a useful survey tool during that period. Chapter Three has more information on frog calls.

All Minnesota anurans lay their eggs in water and have external fertilization, but the form in which the eggs are deposited varies considerably. For example, the wood frog lays large, globular egg masses in dense clusters; the American toad lays long strings of eggs; and the spring peeper lays eggs singly or in clusters of two to three. The larvae that emerge from these eggs develop and metamorphose in the breeding pond. A peaceful looking breeding pond is a deadly arena of competition and predation for the tadpoles and salamander larvae that live there. Ecological studies with wood frog larvae in Alaska showed that 98 percent of larvae that hatched did not survive through metamorphosis (Herreid and Kinney, 1967).

Because Minnesota anurans breed in water, one might think that this is the way frogs reproduce in general. Nothing could be further from the truth; worldwide, anurans exhibit an incredible diversity of reproduction. There are tropical species that lay their eggs in foam nests over water; the eggs hatch and develop into tadpoles in the foam. When they are large enough, the tadpoles drop into the water below. In some species the eggs are deposited on the back of one of the parents. The embryos hatch and go through early tadpole development on the back. The parent visits ponds and as the tadpoles mature they drop off. In other species the embryos hatch, develop, and metamorphose into froglets on the parent's back; there is no free-swimming aquatic stage. An Australian species was recently discovered in which the eggs are swallowed and develop in the mother's stomach—the metamorphosed froglets emerge from her mouth! Somehow, the female is able to turn off her digestive secretions and the stomach becomes a brooding chamber.

Anuran larvae are called tadpoles, or sometimes, polliwogs. Both these terms derive from middle English and translate literally as "toad head" (tadpole) and "wiggling head" (polliwog). Scientists prefer tadpole. These terms are not correctly applied to salamander larvae which are unimaginatively called salamander larvae. Mature salamander larvae closely resemble adults and the morphological transformation that occurs at their metamorphosis is rather minimal compared to the dramatic transformation of anuran metamorphosis.

Tadpoles are primarily herbivores, grazing on the vegetation of the ponds and streams they inhabit. If you examine the belly of a tadpole you can see the long, coiled intestine. This lengthy digestive apparatus is necessary for processing plant material. At metamorphosis the digestive tract becomes dramatically shorter and the tadpole becomes a carnivorous adult eating primarily invertebrates. Large bullfrogs can take larger prey such as mice, baby turtles, and even young waterfowl.

Frogs are found in the diet of many birds, mammals, and snakes, who apparently consider them good "munchies." Some species have potent chemical defenses. For example, Minnesota toads secrete toxic substances from the skin that deter many potential predators. As with the salamanders, these chemical defenses are not perfect. Raccoons eat toads; they flip them, eviscerate them, and leave behind the distasteful upper body skin. Many tropical frogs also have toxic skin secretions and are brightly colored, apparently to warn would-be predators.

Minnesota Reptiles

Three of the six major groups of reptiles are found in Minnesota: turtles, snakes, and lizards.

Turtles

Nine species of turtles representing three families (Chelydridae, Emydidae, Trionychidae) are found in Minnesota. Turtles are the only reptiles encased in protective shells, varying from the hard bony dome of the Blanding's turtle to the flexible, leathery shield of the softshell turtles. Turtles lack teeth but possess horny beaks.

Turtles usually mate in the spring. All turtles must lay their eggs on land, embarking on terrestrial excursions that make the females vulnerable to predation and cars. Some Minnesota species lay two clutches of eggs a year, so that one can observe hatchling turtles at a variety of times over the season. Turtles that hatch late in the season may emerge at that time or overwinter underground and come out the following spring.

Fascinating information on turtle reproductive biology has emerged recently. Laboratory and field experiments and observations show that, in some species, the sex of hatchling turtles is determined by the incubation temperature of the eggs (e.g., Vogt and Bull, 1982). This phenomenon has been demonstrated for several species found in Minnesota (false map turtle, map turtle, Ouachita map turtle, painted turtle, snapping turtle) by researchers at the University of Wisconsin. Low temperatures (25 °C) produce mainly males; high temperatures (31 °C) produce mainly females. This discovery raises many interesting ecological and evolutionary questions. For example, do female turtles manipulate the sex of their offspring by choosing different egg deposition sites (e.g., exposed, sunny spots versus shaded spots)?

Minnesota turtles are mainly aquatic. Only the wood turtle is semiterrestrial, preferring small, fast-moving streams in relatively undisturbed forests. The aquatic turtles fall into two main groups: river and stream turtles (map turtles and softshells) and turtles of lakes, ponds, and marshes (snappers and painted turtles) (Fig. 3). Turtles have catholic diets. Depending on the species, they feed on a wide range of plant and animal material.

Adult turtles are usually safe from predation. They can, however, be successfully attacked by raccoons and dogs during their terrestrial excursions. Humans, whether hunting or in automobiles, are also a major threat. Baby turtles are vulnerable to a wide variety of predators (fish, snakes, raccoons).

Lizards

If you are interested in lizards, Minnesota is not the place to visit. There are only three species found in the state, in two families (Scincidae and Teiidae). The depauperate Minnesota lizard fauna should not mislead you; worldwide, there are more lizard species than any other kind of reptile (3000 species).

A scaled body and four legs quickly distinguish a lizard in Minnesota. Elsewhere, this task is not always so easy because some species have lost one or both pairs of limbs. One such legless lizard—the western slender glass lizard (*Ophisauraus attenuatus*)—is found in Wisconsin. Lizards differ from snakes in having visible external ear openings, movable eyelids, and often (all Minnesota species), tail autonomy. Tail autonomy, which is frustratingly familiar to would-be lizard catchers, is the ability to shed a portion of the tail when molested. The wiggling detached tail is often all the predator catches.

Minnesota lizards usually mate in the spring, shortly after emergence from overwintering. Eggs are laid in cavities, in logs, or on the ground and hatch later in the summer. The reproductive behavior of our two skink species is especially noteworthy: the females brood their egg clutches. Because lizards are ectothermic, they are not capable of metabolically generating heat for the developing eggs, so this brooding behavior should not be confused with incubation. Brooding is probably important in guarding against predators and in egg hygiene.

The northern prairie skink is the most abundant and widely distributed lizard in Minnesota. It is found in open areas with sandy soil (Fig. 4). Six-lined racerunners are restricted to sandy soil areas, especially hillsides, in the southeastern corner of the state along the Mississippi River. The five-lined skink is known only from several granitic rock outcrops in the Minnesota River valley in Yellow Medicine, Renville, and Redwood counties and is the subject of recent ecological investigations by Jeff Lang (1982). Based in large part on Lang's study, it is the only Minnesota herp with "endangered" species status in the state. Minnesota five-lined skink populations are separated from the main body of the species' geographic range (eastern and southeastern US) by several hundred miles. Similar isolated populations are known from Iowa and South Dakota. This unusual distribution makes the Minnesota populations of special interest to biologists.

Minnesota lizards are carnivores and eat a wide variety of small invertebrates. They in turn are found in the diet of a number of birds of prey, mammals, and snakes.

Snakes

Snakes are the most diverse group of herps in Minnesota, numbering 17 species in two families (Colubridae, Crotalidae). In addition to their elongate, limbless body forms, snakes are distinguished by the absence of external ear openings and lack of movable eyelids. Snakes have a transparent protective shield covering their eyes, the brille. The hypnotic movement of the serpentine body invokes a fascination that has generated much mythology over the centuries and initiated the careers of more than a few herpetologists (Fig. 5).

The body form of snakes has given them access to a unique ecological niche, but also presents feeding problems. Snakes must capture and manipulate their prey without appendages. Depending on the species, snakes may simply lunge and grab food with their mouths or use their bodies to pin or constrict prey. A snake's head is the major manipulator of food, taking the place of hands. Many species' skulls are marvels of biomechanical engineering that allow ingestion of prey items larger than the snake. The most sophisticated feeding adaptation is the killing of prey by injection of venom.

Venomous snakes are a source of fascination and fear. They are in large part responsible for the negative public image of herps. The two poisonous snakes found in Minnesota—timber rattlesnake and massasauga—are pit vipers, which use a heat sensitive pit found between the nostril and the eye to locate prey. Their fangs function as natural hypodermic syringes through which venom is forced under pressure into the prey animal. The venom itself is toxic saliva produced by modified salivary glands. Pit viper venom digests muscle tissue, causes blood cell destruction, hemorrhaging, and swelling (Fig. 6).

Minnesota snakes breed in spring or fall; mating often occurs in or around hibernacula (communal overwintering sites). Some lay eggs (oviparous snakes; e.g., rat snake, gopher snake) in the early summer. Others give birth to living young (ovoviparous snakes; e.g., red-bellied snake, timber rattlesnake) in the late summer or fall. The developing embryos of the livebearing snakes are retained by the female and receive nourishment from their egg yolk. Recent studies indicate that embryos in the oviduct may also absorb nutrients from the mother.

In Minnesota, snakes are found in a variety of ecological settings, from aquatic—the northern water snake—to terrestrial—the forest-litter dwelling northern redbelly snake. All snakes are carnivores. Smaller snakes eat small invertebrates; larger snakes are capable of taking small mammals, birds, amphibians, and other snakes. Although some snakes are dietary generalists and will eat almost anything that happens by, many species are specialists, finicky eaters that prefer a restricted group of prey. In Minnesota the eastern hognose snake is a toad fancier. Although toads are repulsive to many potential predators because of their noxious secretions, the eastern hognose acts as if the irritating whitish fluid secreted by toads were a delicate white sauce. Snakes are a common item in the diet of many birds, mammals, and other snakes.

The Distribution and Abundance of Amphibians and Reptiles in Minnesota

There are 48 species of herps in Minnesota (19 amphibians and 29 reptiles; see Table 3). Six more species may be present as border entrants (species that occur in surrounding states in counties immediately adjacent to Minnesota). Herps are outnumbered in Minnesota by the other vertebrate classes (birds: 350 species; fish: 150; mammals: 60).

The low species number does not mean that herps are unimportant in the natural economy; indeed, some may be abundant locally. A collecting trip will often produce many individuals of one or two species. Also, herp abundance is seasonal. For example, during the spring and fall when amphibians move to breeding and overwintering areas respectively, roads often become slippery with the crushed bodies of thousands of frogs or salamanders. During the rest of the year the casual observer would hardly be aware of these animals.

Minnesota's low herp species richness is typical of the upper midwest. There are 54 species in Wisconsin, 25 in North Dakota, and 40 in South Dakota (Vogt, 1981; Wheeler and Wheeler, 1966). The number of herp species decreases as one travels northward; so, as expected, the greatest number of species is found in the tropics. The ardent herpetologist will have to go south and east in the United States for a richer herpetofauna (North Carolina: 138 species; Kansas: 91 species; Pennsylvania: 73 species; Kentucky: 104 species; Martof et al. 1980; Collins, 1974; McCoy, 1982; Moriarty, personal communication). The Minnesota herpetologist can take solace in Alaska's list: six species—all amphibians (Hodge, 1976).

Species richness follows patterns within the state, also. The southeastern corner along the Mississippi River valley is the unqualified herpetological "hotspot," hosting 38

Table 3

Amphibians and Reptiles of Minnesota: A Checklist with Status Determinations

Compiled by the Amphibian and Reptile Group, Endangered Species Technical Advisory Committee to the Commissioner, Minnesota Department of Natural Resources; Jeffrey W. Lang, Chairman. See Appendix II for definitions of status determinations.

REPTILES (29)

Turtles (9)

SC Snapping Turtle (*Chelydra serpentina*)
Painted Turtle (*Chrysemys picta*)
TH Wood Turtle (*Clemmys insculpta*)
TH Blanding's Turtle (*Emydoidea blandingi*)
Map Turtle (*Graptemys geographica*)
Ouachita Map Turtle (*Graptemys ouachitensis*)
False Map Turtle (*Graptemys pseudogeographica*)
Smooth Softshell (*Trionyx muticus*)
Spiny Softshell (*Trionyx spiniferus*)

Lizards (3)

Six-lined Racerunner (*Cnemidophorus sexlineatus*)
EN Five-lined Skink (*Eumeces fasciatus*)
Prairie Skink (*Eumeces septentrionalis*)

Snakes (17)

SC Racer (*Coluber constrictor*)
SC Timber Rattlesnake (*Crotalus horridus*)
Ringneck Snake (*Diadophis punctatus*)
SC Rat Snake (*Elaphe obsoleta*)
SC Fox Snake (*Elaphe vulpina*)
SC Western Hognose Snake (*Heterodon nasicus*)
SC Eastern Hognose Snake (*Heterodon platyrhinos*)
SC Milk Snake (*Lampropeltis triangulum*)
Northern Water Snake (*Nerodia sipedon*)
Smooth Green Snake (*Opheodrys vernalis*)
SC Gopher Snake (*Pituophis melanoleucus*)
SC Massasauga (*Sistrurus catenatus*)
Brown Snake (*Storeria dekayi*)
Redbelly Snake (*Storeria occipitomaculata*)
Plains Garter Snake (*Thamnophis radix*)
Common Garter Snake (*Thamnophis sirtalis*)
SC Lined Snake (*Tropidoclonion lineatum*)

EN = Endangered
TH = Threatened
SC = Special Concern

AMPHIBIANS (19)

Salamanders (5)

Blue-spotted Salamander (*Ambystoma laterale*)
Tiger Salamander (*Ambystoma tigrinum*)
Mudpuppy (*Necturus maculosus*)
Eastern Newt (*Notophthalmus viridescens*)
Redback Salamander (*Plethodon cinereus*)

Toads & Frogs (14)

SC Northern Cricket Frog (*Acris crepitans*)
American Toad (*Bufo americanus*)
Great Plains Toad (*Bufo cognatus*)
Canadian Toad (*Bufo hemiophrys*)
Cope's Gray Treefrog (*Hyla chrysoscelis*)
Spring Peeper (*Hyla crucifer*)
Gray Treefrog (*Hyla versicolor*)
Striped Chorus Frog (*Pseudacris triseriata*)
SC Bullfrog (*Rana catesbeiana*)
Green Frog (*Rana clamitans*)
SC Pickerel Frog (*Rana palustris*)
Northern Leopard Frog (*Rana pipiens*)
Mink Frog (*Rana septentrionalis*)
Wood Frog (*Rana sylvatica*)

POSSIBLE BORDER ENTRANTS

Slender Glass Lizard (*Ophisaurus attenuatus*)
Spotted Salamander (*Ambystoma maculatum*)
Tremblay's Salamander (*Ambystoma tremblayi*)
Woodhouse's Toad (*Bufo woodhousei*)
Four-toed Salamander (*Hemidactylium scutatum*)
Plains Spadefoot (*Scaphiopus bombifrons*)

species. As one travels north and west in the state, fewer species are found. Twenty-four species have been collected in the prairie counties around Granite Falls. Twenty-two species have been recorded from the Lake Itasca area in Clearwater, Becker, Hubbard, and Mahnomen counties. The extensive peatlands in Koochiching, Beltrami, and Lake of the Woods counties are poor herping areas, with only 12 species reported.

What are the reasons for these patterns of herp distribution in Minnesota? Climate and vegetation help determine which species can successfully inhabit an area. As one travels northward in the state, the average temperature drops and the herp activity season, as estimated by the number of frost-free days, shortens. In southeastern Minnesota there are about 160 frost-free days, in northern Minnesota about 100 days. Average precipitation decreases from east to west (average minimum rainfall in the southeast corner of the state = 32 inches; southwest corner = 26 inches; northwest corner = 30 inches; Breckenridge, 1944). These climatic factors, coupled with the distribution of soil types and topography, determine Minnesota's vegetation. The state can be roughly partitioned into three zones: the northern conifer/hardwood forest, the deciduous forest, and the prairie (Fig. 7). The structure and characteristics of these different communities offer different resources for herps.

Wheeler and Wheeler's (1966) analysis of the species distribution of the Dakotas and Minnesota clearly shows the influence of climate and vegetation on species diversity. I have graphically portrayed their data in figure 8. Their division of Minnesota into northern and southern sections by continuing the Dakotas' borderline eastward makes the regions shown more comparable in area and latitude. Note the climatic (rainfall and temperature) patterns already discussed. Concordant with these changes, the vegetation shifts from moist forest to drier grassland from east to west.

The total number of species declines as one travels from Minnesota to the Dakotas, accounted for by a decrease in the number of amphibian species (30 percent decrease from "North Minnesota" to North Dakota and 37 percent from "South Minnesota" to South Dakota). The number of reptile species changes only slightly in this east-west transect. The opposite is true if the northern and southern sections of each state are compared. The number of reptile species decreases by about 50 percent from south to north in both the Dakotas and Minnesota; the number of amphibian species changes little from south to north. These patterns suggest that the reptilian fauna is affected more by decreasing temperatures and the amphibian fauna more by increasing aridity. These trends make biological sense, given the basic differences in the physiological ecology of amphibians and reptiles.

A less obvious factor in species richness patterns is the geological history of our region. Four times in the past two million years glaciers have covered the region and altered the landscape. As the glaciers retreated, plants and animals recolonized the formerly glaciated territories. The pattern of species occurrence in Minnesota today is the result of the most recent recolonization during the last 10,000 years, complicated by climatic changes during the same period.

Seven thousand years ago, Minnesota's climate was warmer and drier than it is today. Prairie extended into regions now occupied by coniferous forest. A shift toward cooler and wetter conditions over the last several thousand years has resulted in new landscape features, such as the boreal peatlands of northern Minnesota. Animals, especially small sedentary ones such as herps, were forced to move or go locally extinct as the location and composition of vegetation zones changed. The distribution and abundance of herps in Minnesota was undoubtedly much different in the past. In recent times our species has played an increasingly important role—through habitat destruction—in the distribution and abundance of herps in Minnesota.

The five-lined skink provides a good example of the influence of history on current

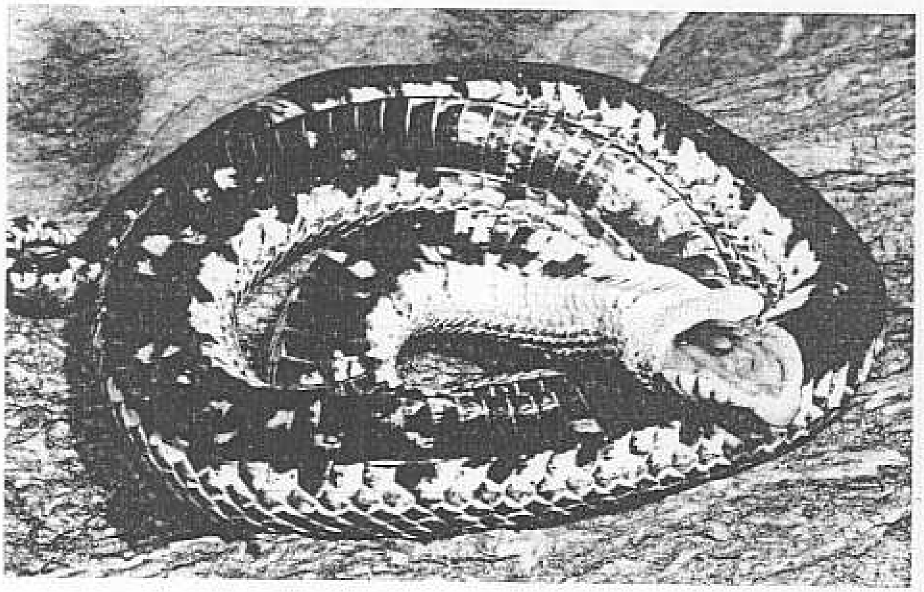

Figure 5. Western hognose snake from Anoka County. Hognose snakes defend themselves by hissing, flattening their bodies, and striking. If this fails to discourage the attacker, they will feign death, as this snake is doing. Photo by Barney Oldfield.

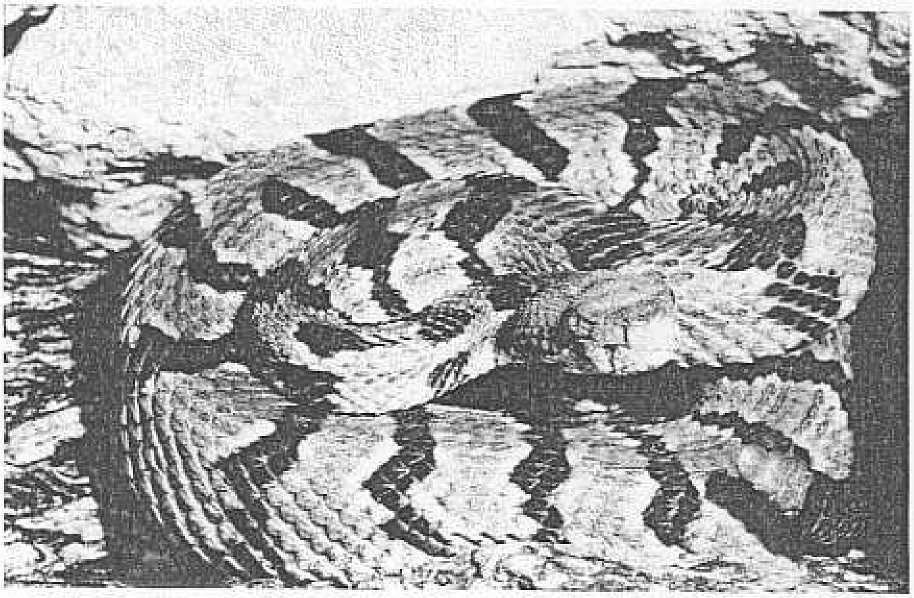

Figure 6. Timber rattlesnake from Maiden Rock, Wisconsin. Only two species of venemous snakes occur in Minnesota (the other is the massasauga), both in the southeastern corner of the state. These snakes are pit vipers, having two hollow, retractable fangs through which venom is injected.

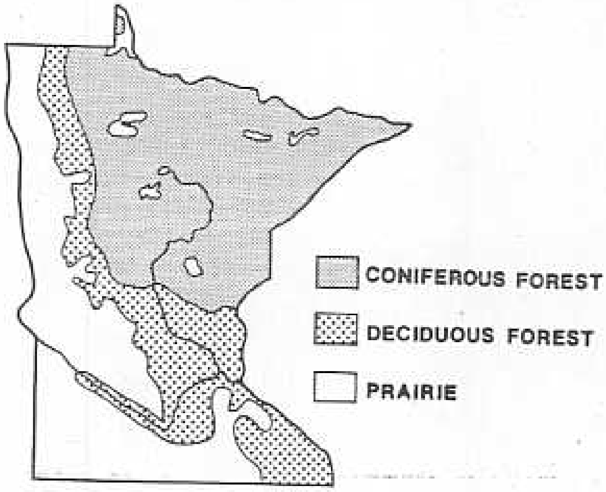

Figure 7. Major vegetation zones in Minnesota.

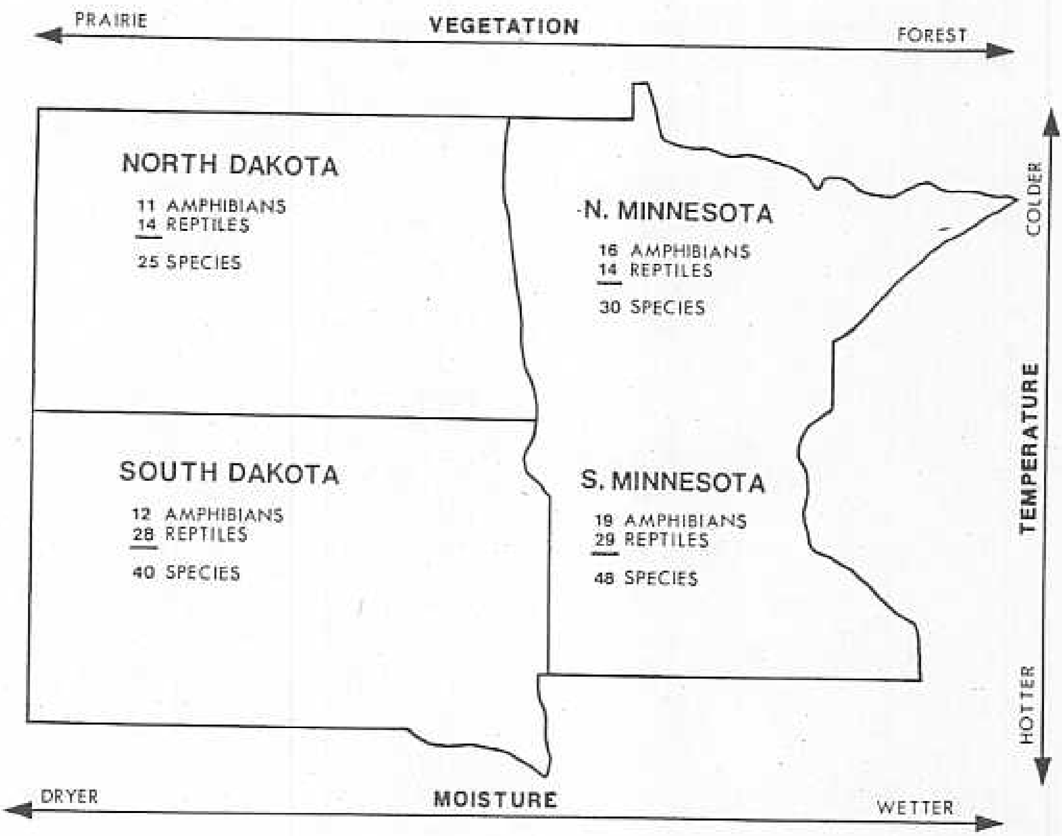

Figure 8. Patterns in the number of herp species in Minnesota and the Dakotas (modified from Wheeler and Wheeler, 1966). The observed patterns are correlated with changes in temperature, moisture, and vegetation. See text for further discussion.

species distributions in Minnesota. The presence of isolated populations of five-lined skinks in western Minnesota suggests that this species was once more widespread in the state. As changing conditions eliminated the species in most of the state, the western populations were left behind as remnants.

An examination of three different habitats in Minnesota, the western prairie, northern boreal peatlands, and southeastern Mississippi River valley, provides further insight into the reasons for current herp distribution. The dry, open grassland of the prairie is a structurally simple environment where sun and wind have a major impact. Low rainfall puts this environment "off-limits" for many species of moisture-requiring amphibians, resulting in the observed east-west decline in amphibians. Prairies are also the most heavily disturbed of Minnesota's native habitats, and this has undoubtedly affected the recent distribution and abundance of the prairie species.

The extensive boreal peatlands of northern Minnesota are a herpetologically "depressed" area of the state. My survey of central Koochiching and eastern Beltrami counties revealed only 12 species. By comparison, 22 species of herps are known to occur in the vicinity of Itasca State Park 75 miles to the south (Karns, 1979).

Several factors help explain this impoverished species list. The peatlands are characterized by fens—wet, open areas dominated by grasses and sedges, and by *Sphagnum* moss-dominated bogs. The water-saturated soils of the peatlands prohibit colonization by terrestrial burrowing herps, such as the northern prairie skink, which prefers open areas with sandy soil and is found to the south and northwest of the peatlands.

On the other hand, the peatlands might seem to be a paradise for moisture-loving amphibians. The peatlands certainly have plenty of water (in many respects the area is like a vast, shallow, subsurface river); but permanent, deep bodies of water that do not freeze to the bottom during winter are scarce. Mink frogs, which prefer true lake habitat, are not found in the peatlands, but are common in the lake districts surrounding them. It is likely that the distribution of water in the peatlands (water saturated soils, but few lakes) prohibits colonization by the mink frog and other species with similar requirements.

Water quality may also influence amphibian distribution in the peatlands. I found that bog water associated with *Sphagnum* bogs (acidic, darkly colored, high humic content) was toxic to amphibian embryos (Karns, 1984).

The southeastern corner of Minnesota along the Mississippi River valley is the herp "hotspot" of Minnesota. This is not surprising; more rain falls there than anywhere else in the state. The vegetation is dominated by hardwoods. There are also extensive bottomlands with marshes and sloughs, bluffs and ravines, and patches of prairie on southern exposures. The many types of habitat can be used by a wide range of species with differing ecological requirements.

Another factor explaining the high species count in this area is the Mississippi River valley itself. Many species reach the northwestern limits of their geographic range in Minnesota by using the Mississippi River valley as a movement corridor. The ecological diversity and milder climatic conditions of the river valley allow these species to "make it" in Minnesota. Examples include the smooth softshell turtle, six-lined racerunner, blue racer, black rat snake, and cricket frog.

The distribution of amphibians and reptiles in Minnesota is a complex phenomenon that depends on many interacting factors. The next time you overturn a rock and startle a snake or lizard, contemplate the diversity of biological and geological forces that have led to that individual's presence under that particular rock on that particular day.

The Herpetological Calendar in Minnesota

Phenology is the study of the timing of biological events in relationship to climate. The severe Minnesota climate imposes a stringent schedule on the herpetofauna. A calender of annual herpetological events in Minnesota is presented in Table 4.

Minnesota is a large state notable for its climatic variability. The exact timing of herp events can thus vary considerably across the state. For example, I have heard wood frogs calling in the Twin Cities area in late March, but not in Central Koochiching County (40 miles south of International Falls) until late April.

The timing of events can vary considerably even in the same area during the same year. An amphibian breeding site at the edge of a woods with a southern exposure may be active days before a less exposed woodland pond nearby. Awareness and observation of the annual progression of the biological calender is one of the great delights of natural history study.

TABLE 4
Major Events of the Herpetological Calender in Minnesota

SPRING-EARLY SUMMER (late March-June)

1. Herps emerge from overwintering sites.
2. Amphibians move to breeding areas in large numbers.
3. Main period of anuran and salamander reproduction.
4. Many reptiles mate shortly after emergence.
5. Turtles lay eggs.
6. Snakes lay eggs

SUMMER (July-August)

1. Most species are at summer feeding ranges.
2. Some late breeding frogs still call (bullfrog, cricket frog, mink frog).
3. Young-of-the-year salamanders and frogs appear as metamorphosis is completed.

LATE SUMMER-FALL (August-October)

1. Live-bearing snakes give birth.
2. Some salamanders and snakes breed.
3. Herps move to overwintering sites.

WINTER (November-March)

1. Most species are dormant at overwintering sites.
2. Some aquatic species (mudpuppy, central newt) remain active all winter.

The active season

Herps are active during the warm months of the year. Many species have a similar timetable of behavior: breeding during spring or early summer, feeding and growing during summer, and preparing to overwinter during fall.

The warm months of the year are also the active season for the herpetologist. Familiarity with the herp calender will help you plan your herp field excursions. You should be on the lookout for special, short-lived events such as the overland egg-laying journeys of turtles and breeding periods of anurans.

Winter ecology

What happens to herps during winter? In Minnesota that's a topic that warrants discussion. In fact, a number of important studies on the winter survival of herps have been done in the state.

The long, cold winter in Minnesota limits most herp species to a restricted window of activity when reproduction, feeding, and growth must be accomplished. Small, sedentary ectotherms like herps do not have high, stable body temperatures that permit year-round activity, nor can they opt to avoid winter by long-distance migration. They must locate habitats that are physiologically suitable for winter survival and stay there.

If an organism's tissues freeze, the ice crystals that form rupture cells and destroy the metabolic machinery of life. To avoid this fate, the organism must overwinter at a site where temperatures do not drop below freezing, or use physiological mechanisms that afford protection against below-freezing temperatures. Both strategies are exhibited by Minnesota herps.

Temperature and moisture are the key factors in determining the suitability of an overwintering site. Moisture, as might be expected, is especially important for overwintering amphibians. The more aquatic species such as the northern leopard frog and mink frog overwinter underwater, "breathing" through their skin. Experiments done by Bill Schmid at the University of Minnesota (Schmid, 1965) indicate that these species could probably not tolerate drier, terrestrial overwintering sites. Overwintering leopard frogs have been observed by scuba divers lying on the bottom, and are often found concentrated at favorable spots. This behavior, of course, makes them vulnerable to frog hunters.

Gib Hedstrom, a semi-retired professional frog catcher, recalled a particularly good frogging site at Wesport Lake near Sauk Centre (Wheeler, 1979). In the "good old days" (1930's) there were culverts around the lakes where the water was about three feet deep and "laid solid with leopard frogs from the month of December to February." He remembers that "a great frog picker" could pick 150 to 300 pounds a day (at eight cents per pound). The market for these frogs was biological supply houses which sold them to schools and research laboratories. In 1975 the DNR closed commercial frog harvesting in Minnesota due to dwindling population levels of the leopard frog.

Other amphibian species overwinter on land. One strategy is to burrow (or find burrows) below the frostline. John Tester and Walter Breckenridge of the University of Minnesota investigated the overwintering behavior of Canadian toads (Tester and Breckenridge, 1964). They followed radioactively labeled toads with a geiger counter. These toads burrowed downward, keeping ahead of the frostline, to depths of more than four feet. Soil consistency, temperature, and soil moisture appeared to be important factors in their movements. The toads favored particular sites called mima mounds where they could be found by the thousands. Mima mounds are island-like mounds of soil that dot the prairie in certain areas of northwestern Minnesota. The toads may be an important factor in the development and maintenance of the mima mounds.

Some frogs overwinter at or near the ground surface. For example wood frogs, chorus frogs, and gray treefrogs overwinter in shallow depressions in the leaf litter. Temperatures in these sites can fall below freezing, especially if snow cover is light or absent. How do these frogs survive? Bill Schmid at the University of Minnesota has performed pioneering experiments in the physiology of overwintering in amphibian species that remain near the ground surface. He has found that wood frogs, spring peepers, and gray treefrogs can tolerate below-freezing temperatures (-4 to -9 °C). The frogs appear to have a double line of defense against low temperature. First, they can tolerate freezing of a considerable portion of their body fluids (about 35 percent). Second, a biochemical "antifreeze," glycerol, helps them resist freezing, a phenomenon known as supercooling. Many other organisms (e.g., insects) also survive below-freezing temperatures by using such biochemical antifreezes to help prevent freezing. Thus, these frogs both supercool and tolerate some freezing of body water. The problem of cell destruction due to freezing is apparently avoided by "allowing" only water between cells and in the urinary bladder to freeze (Schmid, 1982).

Reptiles also seek overwintering sites with favorable thermal conditions for winter survival. Research on the physiology of reptilian overwintering shows that many species are quite cold hardy and do not require sites with temperatures significantly above freezing (Gregory, 1982). Also, most reptiles appear to be capable of supercooling (defined above) to some degree. The general importance of supercooling to overwintering reptiles is not fully established and requires more research. Reptiles in hibernacula (overwintering sites) that remain above freezing avoid the problem of tissue destruction. Other species (e.g., hatchling turtles in shallow nest burrows) undoubtedly face freezing temperatures and may supercool.

Minnesota's turtles overwinter as adults, in or near water. Depending on the species, they may burrow in river banks or the river bottom, simply lay on the bottom (behind logs or rocks), or use muskrat or beaver burrows. They often congregate at favorable sites, becoming vulnerable to turtle trappers.

Lizards seek crevices or burrows or dig their own burrows. A quarryman from Granite Falls told me of quarrying out huge slabs of rock in the winter and finding five-lined skinks tucked in crevices far below the surface.

Snakes usually use natural cavities (ant mounds, rock crevices, rodent burrows) that offer favorable thermal conditions. Snakes often congregate at these sites in considerable numbers, with several species present. Jeff Lang (1971) investigated the use of ant mounds as hibernacula in northern Minnesota. An excavated mound contained 299 snakes of three species (smooth green snake, red-bellied snake, and young eastern garter snakes) distributed from the surface to a depth of 264 cm (104 inches). Unfortunately these communal hibernacula, with up to thousands of snakes, make snakes particularly vulnerable to collectors or people who wish to eliminate snakes from a local area.

Snakes may be faithful to a particular hibernaculum year after year, traveling up to several kilometers to return to it. What advantages do communal hibernacula offer? Several ideas have been proposed (Gregory, 1982). Suitable hibernacula may be scarce in a given area and force snakes to congregate at the few available sites. There may be thermal advantages to gathering in large masses, but there is little support for this idea. Communal hibernacula may facilitate mating activity. The majority of snake breeding occurs in the spring shortly after emergence; the congregation of a large number of males and females during the winter ensures that mates will be readily available in the spring. More research is needed to fully understand the phenomenon.

Snakes' use of hibernacula results in a number of phone calls to the Bell Museum's Wildlife Information Line every spring. The unfortunate homeowner whose home is under seige by dozens or hundreds of snakes is desperate. The local garter snake population has discovered a house with suitable entry ways that can be used as a winter refugium. When spring arrives, some snakes are seen as they try to leave. Sealing up the possible snake entry points after they leave is the best solution to this problem.

Some herps, including mudpuppies and central newts, remain active all winter under the ice (Vogt, 1982). Snapping turtles and painted turtles have been seen moving under the ice (Breckenridge, 1970; Vogt, 1982). Green frogs sometimes overwinter as tadpoles and are known to feed during the winter (Vogt, 1982).

Even species overwintering on land may be active during the winter. Canadian toads continue burrowing all winter just ahead of the dropping frostline (Tester and Breckenridge, 1964). Snakes sometimes move around in their hibernacula (Gregory, 1982), and red-back salamanders in Indiana feed on ants during the winter (Caldwell, 1975). Thus, depending on the species and the situation, winter is a period of more herp activity than one might expect.

TWO
How To Find and Observe Amphibians and Reptiles

A primary pleasure of natural history study is going into the field to observe animals in their natural state. These excursions provide exercise, education, and fun. Herping requires that you overturn logs and stones or that you get wet and muddy in search of squirming salamanders, ferocious frogs, and, of course, leaping lizards. Herping allows an outlet for the childlike sense of wonder that many people have about nature, so that they can get out and "poke around" their corner of the natural world.

Unfortunately, herping can also be very frustrating. Success depends on the season, temperature, rainfall, time of day, species being hunted, and habitat being searched. Failure is likely under some conditions (e.g., salamander hunting in a forest during a summer drought). Under favorable conditions, almost every step or overturned log may reveal a herp (e.g., looking for salamanders in the same forest noted above, but in the spring after rains). Choosing times and places that will maximize your chances of herping success requires a knowledge of herp biology. The activity calender for Minnesota herps (Table 4) provides a general guide for planning herp field activities. Chapter Three gives specific recommendations on where and when to maximize herping success.

Careless herping can lead to deterioration of habitat. Inconsiderate herpers leave every rock and fallen tree overturned, strip bark off fallen trees, and rip up moss. Such activity destroys essential microhabitats used by herps and many other organisms. Restore areas that you have visited to their original condition. Most formally designated natural areas (wildlife refuges, state and national parks, county nature preserves) forbid collection of wild plants and animals, limiting herpers to observation (see Appendix II on Minnesota herp laws; Czajka and Nickerson, 1974, provide information on collecting in other states).

If you catch a herp in the wild and have no legitimate reason for keeping it, let it go where it was captured. This last point is important: releasing the herp outside an area with which it is familiar may decrease its survival chances. Keeping a garter snake as a pet or raising tadpoles through metamorphosis are real pleasures; however, the sad fact is that most people are poor herpkeepers and the fate of many captives is neglect and slow starvation.

If you are planning to keep herps in captivity, do some research in advance. Find out about feeding, temperature and cage requirements, and the legality of keeping the animal you have caught. Several good references are provided in Appendix III. Pet stores can also be a source of information, but many pet dealers do not know much about herps. Two Twin Cities' area pet stores specialize in herps and the Minnesota Herpetological Society is also a valuable source of information on herp care. Information on these sources is provided in Appendix I.

Basic herping involves searching an area for any and all herps. The main tool employed is the human hand. Dick Vogt (1982) has referred to this basic activity as "search-and-seize" herping.

Terrestrial Methods

Searching

A slow walk through an area, scanning the substrate for any movement or herp-like shapes, will reveal things that go unnoticed by a casual observer. Herps that are on the surface may "freeze" at your approach and rely on their cryptic coloration and motionless stance to avoid detection. Alternatively, your presence may cause an active flight response, so that all you capture is a glimpse of a snake disappearing into the leaf litter or the blur of a lizard as it vanishes into the crevice at the base of a tree.

Overturn rocks, logs, and garbage as you walk. Any kind of debris may serve as a shelter for herps, since most do not excavate their own burrows and hiding places. This means that good herping spots are not always found in idyllic natural surroundings (Fig. 9). The densest northern prairie skink population I have encountered in Minnesota was located in an old garbage dump in Anoka County, with skinks in beer cans, rubber tires, and sofas.

There is a right way and a wrong way of overturning objects to look for herps. Always pull the object toward you, creating a shield between you and whatever you have uncovered (Fig. 10). If that turns out to be a poisonous snake or a skunk, you are protected and can simply drop the overturned object. Be realistic in appraising your ability to move objects. I have been in the field with herpers who perform isometrics with small boulders and large fallen trees in vain attempts to move the immovable. Again, keep the microhabitat intact by replacing anything you have disturbed.

Stevadore hooks, potato rakes, sticks, and crowbars are useful in the field for overturning objects and probing debris. I have found potato rakes to be particularly useful in areas with a lot of small objects to overturn. These tools save wear-and-tear on the hands and back. Gloves protect the hands from abrasion, especially in rocky areas.

Seize and examination

Very little equipment is needed for capturing herps. Most herps, with a little practice and quick reflexes, can be captured by hand. For immobilizing snakes and other herps, a snake stick is very handy.

A snake stick is simply a pole with a piece of flattened metal at one end. The L-shaped end of the pole is used to pin the animal (Fig. 11). Snake sticks can be made from old golf putters by filing down the putter head, by attaching a metal hook to a wooden pole, or, most simply, by attaching a piece of angle iron to one end of a pole. Potato rakes also make fine snake sticks. Commercial snake tongs and snake sticks are also available (see Appendix I).

The first rule of snake capture is to immobilize the head. If you forgot to bring your snake stick, pin the snake by gently stepping on it and using a stick (or your other foot with some contortion) to pin the head (nonvenomous snakes only). Grab the snake behind the head gently but firmly, and support the snake's body with the other arm for safe examination (Fig. 12). Some species (e.g., eastern garter snake) may attempt to spray you with a foul smelling cloacal discharge.

A word of caution is appropriate concerning venomous snakes. Minnesota has only two species of poisonous snakes (timber rattlesnake and massasauga) found in a limited area in southern Minnesota. Learn to identify these snakes and know where they occur. Be observant in rattlesnake country. Do not place your feet or hands in places you cannot see. NEVER attempt to handle a poisonous snake without good reason. The chance of being bitten if you are cautious is very small. More information on Minnesota's poisonous snakes and treatment of snakebite is given in Appendix IV.

Fast moving lizards can be difficult to catch. Noosing is a technique that works well with some species. Stiffen a piece of fine thread by waxing and attach it to the end of a pole or old fishing rod. Slip the noose over the head of a basking lizard and, with a well-timed jerk, you have your lizard. Another capture technique for lizards and other hard-to-catch herps is a blowgun made of a four-foot piece of aluminum conduit. With a cork as the projectile the animal is stunned but usually not damaged. I have little personal experience with this technique, but Gibbons (1983) claims that an experienced blowgunner can hit an 8-cm lizard at 12 m almost every time. Gibbons also notes that olives make excellent projectiles. Slingshots can be used effectively, if you are willing to risk damaging a specimen.

Most smaller herps can be caught by slapping your hand down on them. Be careful not to come down too hard! After whole-body-slap-down seizure, the captive must be carefully extricated from beneath your hand, if, indeed, you didn't miss it.

Most lizards, anurans, and salamanders can be held for examination by gently grasping a rear leg while supporting the body with the other hand. It is important that the animal have something to hold onto for security. Large frogs should be restrained by holding the animal around the waist so it can't kick free; large salamanders can be held by encircling the trunk of the body with your fingers and letting the head protrude.

Snapping turtles and western spiny softshells are pugnacious critters, prone to snapping, biting, and scratching. Snappers can be held by the tail away from your body. Vogt (1982) recommends holding western spiny softshells by inserting the thumb and forefinger into the anterior portion of the hind leg sockets. I have held these species successfully by grasping both edges of the shell midway between the front and hind limbs. Other Minnesota turtles are better tempered and can be picked up easily, but beware of scratching claws.

Transport

If you are planning to carry the catch, obtain a suitable herp container. The traditional method of transport is a sturdy, tightly woven, fabric sack that is about half as wide as it is long. It is handy to have several sizes. A 24 × 50 cm sack can be sewn from pillow case material. Heavy-duty plastic bags are favored by some collectors. Plastic containers are excellent for small, delicate specimens and for aquatic captives. Breadbags or similar plastic bags are convenient for small specimens. Inflate the bag like a balloon and tie it shut; the air cushions the occupants.

Remember the thermal and moisture requirements of your captives while in transport. Provide your catch with moist litter or wet the sack occasionally (especially for amphibians). Store sacks and containers under reasonably cool conditions during transport; the temperature in a hot, closed car can kill herps—especially salamanders—in minutes. Remove your captives from transport containers to more desirable quarters as soon as possible.

Miscellaneous equipment

A field notebook or pad for recording observations is a must for the serious herper. Many notetakers like to jot down observations during the actual fieldwork on a handy notepad and enter the information in a permanent field notebook later. A good field notebook should allow someone else to reconstruct where you went and what you did on a particular day. Chapter Four discusses the information that should be recorded for each specimen encountered.

Additional equipment that can provide an added dimension to your field excursions are a fast-reading cloacal thermometer (for taking substrate and herp temperatures) and a small scale for weighing specimens (Pesola scales are excellent for this purpose). Another handy tool is a reptile sex probe, a small, blunt probe that can be gently inserted into either side of a snake's or lizard's cloaca to determine sex. All male snakes and lizards have two penises (hemipenes) that sit in pouches at the base of the tail, a pouch on each side. The probe can be inserted deeper into a male's cloaca, because of the pouches. These items can be purchased through various supply houses (see Appendix I). Herps provide an interesting challenge for the photographically inclined herper (see Bibliography for good books on field photography).

A fully equipped herpetologist is indeed a formidable creature, loaded down with all sorts of heavy-duty landscape-altering and data-recording equipment. You do not really need all this gear. My working outfit is a small knapsack (with herp guide, camera, notepad, etc.), a supply of herp sacks and small plastic containers (if I am collecting), and a snake stick.

Field clothing is a matter of common sense. Wear sturdy, comfortable clothes and footwear appropriate for the conditions (i.e., do not go into an insect infested, poison ivy laden forest in shorts, tank-top and sneakers). If you are seriously interested in fieldwork, I strongly recommend investing in a good sturdy vest with lots of pockets; they are very handy.

Aquatic Methods

Wetlands, ponds, and streams pose new problems for the herper. Some aquatic herping, such as grabbing frogs sitting at the edge of a pond or turtles basking on logs, can be done without getting wet. Approach basking herps as slowly and quietly as possible; they are extremely wary and will often slip into the water at the slightest disturbance. Binoculars are a good tool for turtle identification; spotting scopes used by ornithologists are excellent for turtle watching from a distance. Turtle watching is where herping and birding intersect.

Obtaining an aquatic or semiaquatic species for examination will usually mean getting wet. In the summer or fall, when the water is warm, I prefer to wade about in old tennis shoes. In the spring, when the water is cold (or at other times if you do not like getting wet), some sort of waterproof footgear is necessary. Options include calf- or knee-length rubber wading boots, hip waders, and chest waders. If you want to go all the way, snorkle or use SCUBA gear.

The problem in aquatic search and seize is getting the animal out of the water. A variety of equipment is available for use in this challenging endeavor.

Nets

Dipnets, a basic tool of aquatic ecology, come in a variety of sizes and shapes including small aquarium models, rectangular bait nets, and triangular crabbing nets. I have found 3/16- 1/4-inch mesh to be a good general purpose size for larger nets. It is convenient to have an assortment of nets available and let the situation dictate their use. Conant (1975) describes a handy net made from a kitchen strainer for catching salamanders. A trip to your local bait shop or sports store will probably satisfy your net needs (see also Appendix I).

Seines

A small seine—about 1.2 m × 1.2 m—is easily handled by two people and is excellent for fairly open shallow water. Seine in early spring for breeding salamanders, later in the year for salamander larvae, tadpoles, frogs, and occasionally snakes and turtles. The two seiners march through the water holding the seine net rigid between them and the bounty of the waters is theirs.

Plunge trays

In areas where aquatic vegetation is particularly dense, nets are difficult to use because they are easily fouled. I have successfully collected amphibians in such situations using plastic refrigerator storage trays (20 × 20 × 5 cm). Plunge the tray into the water every every few steps and quickly pull it out. The water sucked into the tray during the plunge contains aquatic invertebrates, tadpoles, salamander larvae, and occasionally adult amphibians. Pour the water from the tray through an aquarium dipnet, separate the animals from the vegetation and debris, and transfer the animals to a plastic holding jar. One can easily construct a tray with a wire-mesh bottom that will allow water to drain out.

Minnow traps

Plastic minnow traps effectively catch breeding frogs in the spring. Place several traps near an active chorus and there will usually be a number of frogs in the traps the next morning. The method works even better if the trap is "baited" with a calling male. Tadpoles and salamander larvae may also be caught in minnow traps if the trap mesh size is sufficiently small (or the tadpoles sufficiently large).

Muddling for turtles

Turtles can be caught by "muddling" or "noodling" (Plummer, 1979). After you see a turtle slip into the water from its basking perch, use your hands to probe in the muck and rocks around the area of the perch. When you feel something hard that moves, grab it and pull up your catch.

Dick Vogt has considerable experience muddling; although he has occasionally connected with the business end of large snapping turtles, he has never been bitten. You can probe for turtles by "sounding" with a stick or pole if you do not relish the idea of sticking your hands into dark, unseen, underwater crevices. Turtle traps (Fig. 21) can also be made or bought (see Appendix I).

Snorkling and SCUBA

Diving can provide unique glimpses of turtles, salamanders, and frogs. Bill Schmid at the University of Minnesota has used SCUBA to observe overwintering leopard frogs in Minnesota lakes. Max Nickerson at the Milwaukee Public Museum snorkels to search for hellbenders (larger relatives of our mudpuppies) in Ozark streams (Nickerson and Mays, 1972).

Nighttime Methods

If herping is confined to daytime, many nocturnal species will elude detection. A good source of illumination is the major requirement for nighttime herping. Headlamps are useful because they free your hands for collecting, but the beam is usually narrow and tied to your head movements. I like to supplement a headlamp with a powerful waterproof hand lantern or flashlight.

Car cruising

If prowling around at night in the woods is not for you, cruising in your automobile is a way to herp in relative comfort. Secondary roads through relatively undisturbed areas are best for this activity. Drive slowly and watch carefully for any movement or herp-like shapes. With a little practice you will learn to distinguish toads from rocks and snakes from sticks.

Car cruising can be an extraordinarily productive procedure on warm, rainy spring and fall nights when breeding and overwintering movements are taking place. One rainy, fall night I observed an average of a herp-a-minute during a two-hour cruise along a sandy road in northern Minnesota. Warm summer nights are good for snake cruising, because the road retains heat and thus attracts snakes. Dusk and nighttime are best for car cruising, though daytime car cruising can also be productive.

Figure 9. Herpetologists are often found in strange places doing strange things. This graveyard in southeastern Indiana is a site where Kirtland's snake is found. This species is found in the midwest from Ohio through Illinois, but not in Minnesota.

Figure 10. Overturning a stone. This herper is doing it the right way: he is lifting the stone from behind, thereby using it as a shield. If something nasty awaits, the stone can be dropped. This stone revealed a green frog, seeking a cool, damp spot on a hot summer afternoon.

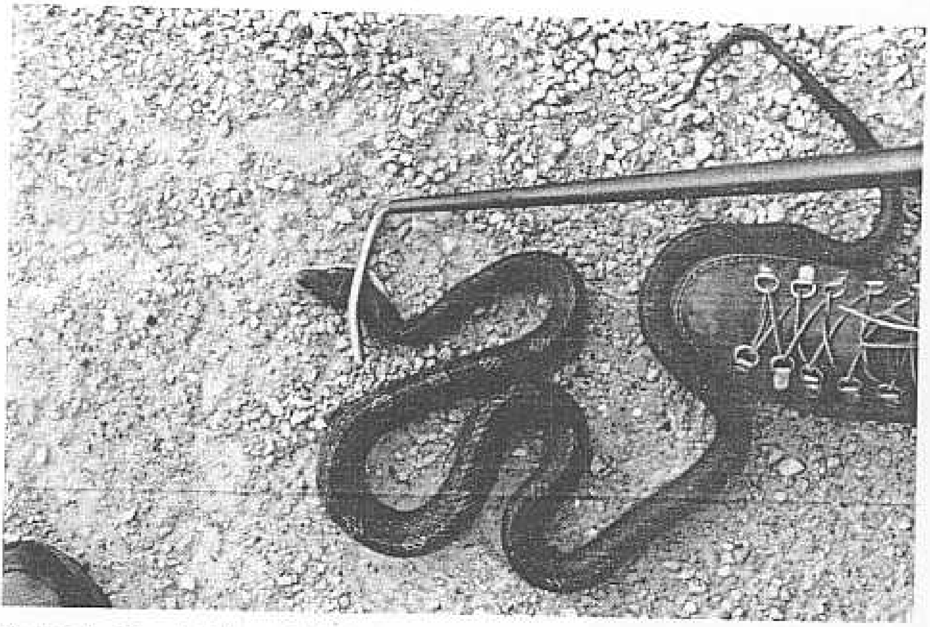

Figure 11. Pinned! A herpetologist's eyeview of a successful capture. The herper is using a snake stick to immobolize a black rat snake. Once the head is pinned, the snake can be safely picked up by grasping it behind the head and holding the body. This commercially made stick is a modified golf putter.

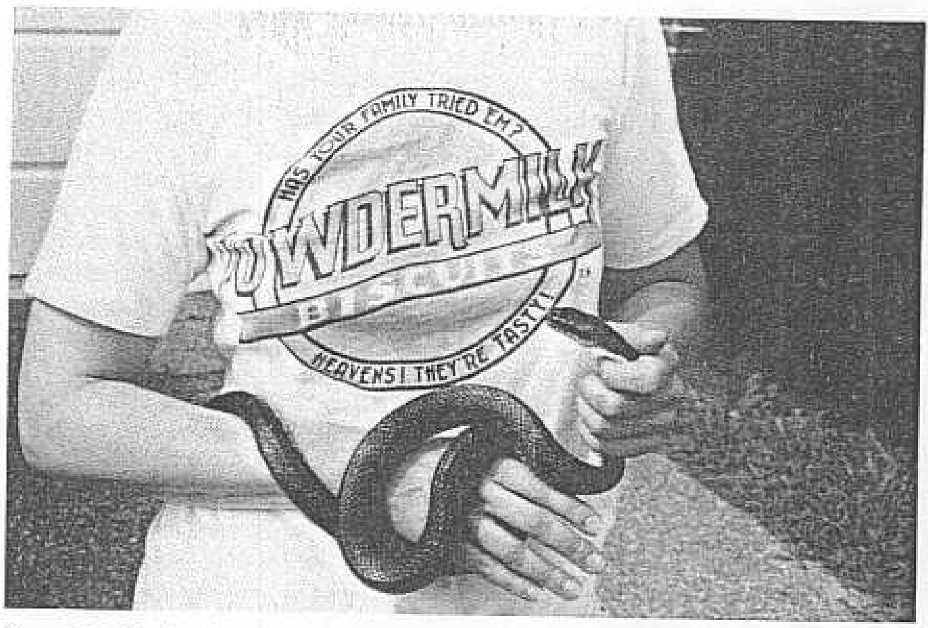

Figure 12. Herping is a popular pastime in Lake Wobegon, Minnesota. This herpetologist needed a few powdermilk biscuits to give her the courage to get up and do what needed to be done, in this case demonstrating the proper way to hold a snake. This black rat snake is held firmly behind the head, with the body supported.

Unfortunately, many people consider running over small critters a form of amusement, so roadkills are common. With practice, one can identify roadkills without slowing down. Herp roadkills increase during the spring and fall movement periods, and can be a valuable source of information.

Frog calls

The spring breeding period of frogs and toads allows the combination of nighttime and aquatic herping. You will need wading gear and a headlamp (or waterproof flashlight or lantern). Carry a dipnet and containers or herp sacks if you plan to collect frogs or eggs.

Locate breeding choruses by driving or walking through an area with appropriate water resources (wetlands, woodland ponds, streams) and listening for calling activity. Frog calls are much easier to learn than bird calls, since there are relatively few species, the calls are simple, and the different species follow a seasonal sequence of calling. Field guides contain descriptions of the calls and records are available that have recordings of several Minnesota species (see Appendix III). The Nongame Program of the Minnesota Department of Natural Resources has produced an excellent slide and tape show of the frogs and toads of Minnesota.

The vocalizations of anurans are acoustical beacons that beckon the herper to participate (in a voyeuristic way at least) in the mating rites of spring. Some species will call during the day, but activity increases at dusk as more species chime in. Calling activity will be low on particularly cold or windy nights.

What you can expect to hear on a favorable calling night depends on the time of year. The peak of breeding activity for different species occurs at different times. For example, in south-central Minnesota, wood frogs, chorus frogs, and spring peepers are early spring breeders. American toads, northern leopard frogs, and gray treefrogs call later, and green frogs later still. Figure 13 is a frog calling calender compiled by Ray Anderson and Deborah Jansen at the University of Wisconsin - Stevens Point based on observations in central Wisconsin. I have found this calender to be a good general guide for anuran calling activity in central and southern Minnesota.

The duration of breeding activity also varies among species. Wood frogs and American toads are examples of explosive breeders; they breed during a short period of time and then disappear. They deposit most of their eggs in one or several areas within the breeding pond, so their breeding acitvity is confined to relatively small areas. At a particular site, wood frogs may call for 4-14 days in the early spring, depending on weather conditions (Fig. 14). American toads breed later and are even more explosive. Almost all their breeding activity at a particular site may occur on a single night.

During the peak of these explosive events an anuran orgy takes place; calling may continue day and night and involve hundreds of frogs. Easy to locate because of the racket they produce, these gatherings can be approached closely without shutting down chorus activity. The explosive breeding periods provide the best opportunity during the year to observe amphibian behavior.

Other species (e.g., gray treefrog, green frog, spring peeper) are prolonged breeders that call for four to eight weeks at a given site, depending on weather conditions. Males, who defend areas where mating occurs, are usually spaced out over the breeding pond, often concealed by vegetation. Observing these prolonged breeding species can thus be challenging.

Pinpointing the source of a call, especially the higher pitched calls (e.g., spring peeper) that have a ventriloquial quality, is difficult. Two people working together can use triangulation to zero in on the caller. Move slowly and avoid creating ripples that will disturb the caller. Once you see a calling spring peeper or chorus frog you will develop a search image that will help you pick out these small frogs against the background clutter. For example, the inflated vocal sac of the chorus frog is as big or bigger than the frog itself and will appear as golden bubbles the size of grapes on the surface of the water.

When disturbed, a few individuals may cease calling, followed by the entire chorus shutting down almost instantaneously. If this happens, and the area of the pond you are standing in is suddenly silent, remain still. Calling will usually begin again in a few minutes. Your disturbance may shut down the chorus for long periods of time if it is a rather cool night or if the density of calling frogs is low.

Properly dressed, wading through an April woodland pond in search of a wood frog orgy or the commonly heard, but rarely seen, chorus frog or spring peeper is a lot of fun. There are no biting insects out yet and you will usually have the woods to yourself. A hot shower feels great after such an expedition.

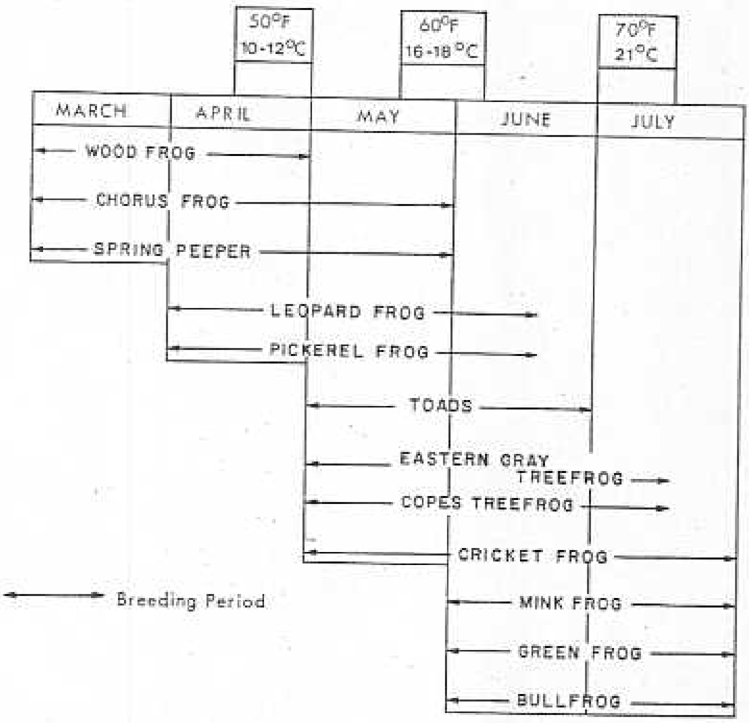

Figure 13. Breeding activity calendar for frogs and toads from central Wisconsin to central and southern Minnesota (modified from Hine, 1982). The horizontal lines indicate, for various species, the expected breeding period, when males will be calling. The three boxes on top of the chart show recommended listening periods (with minimum water temperatures) for hearing a diverse collection of frogs.

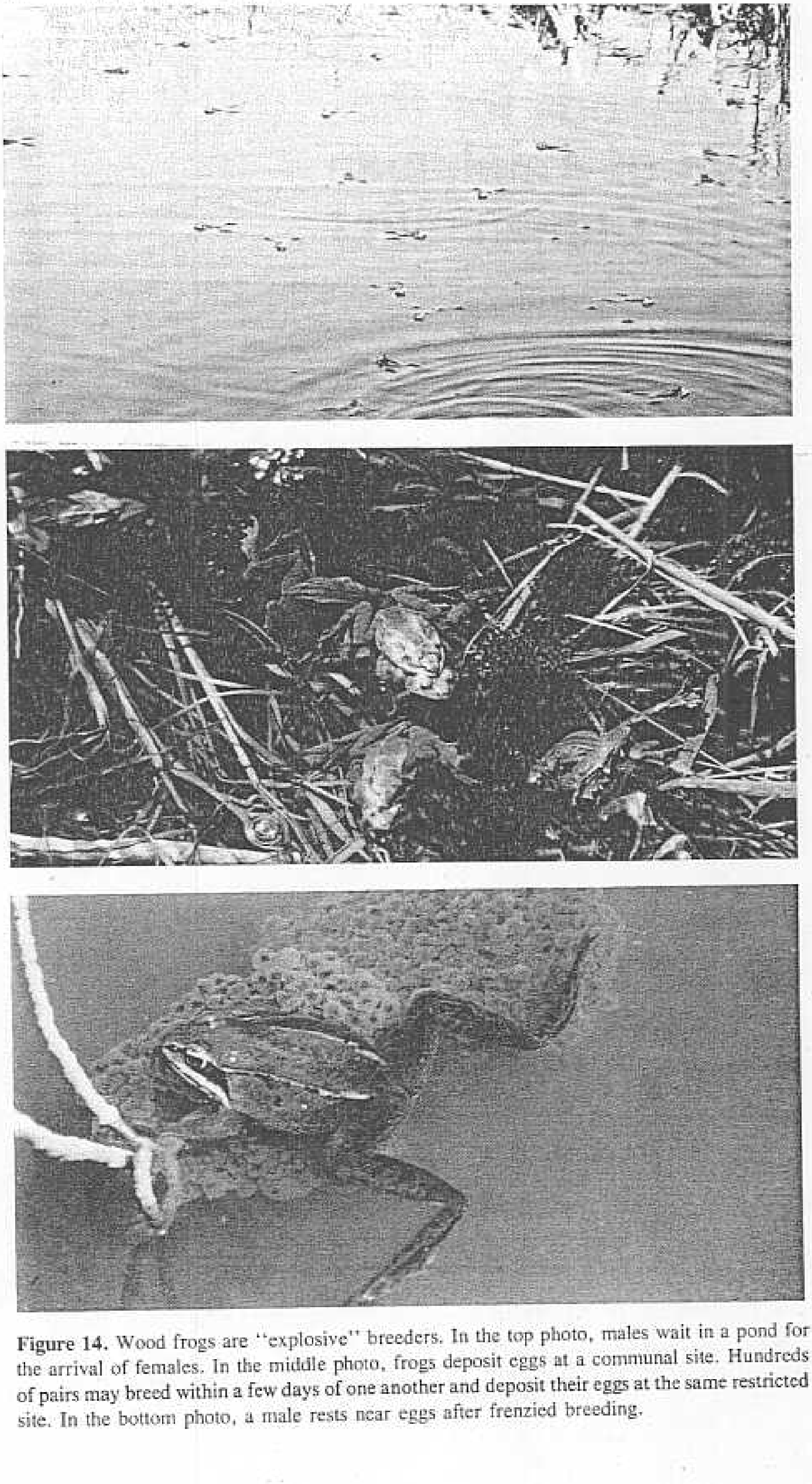

Figure 14. Wood frogs are "explosive" breeders. In the top photo, males wait in a pond for the arrival of females. In the middle photo, frogs deposit eggs at a communal site. Hundreds of pairs may breed within a few days of one another and deposit their eggs at the same restricted site. In the bottom photo, a male rests near eggs after frenzied breeding.

THREE

Survey Methods for the Assessment of Amphibian and Reptile Communities

An inventory of plant and animal communities is fundamental to understanding an area's ecology and a necessary prelude to intelligent management and conservation. In this chapter I discuss a number of methods used by herpetologists to describe amphibian and reptile communities in a given area, including general, time-constrained, and quadrat search-and-seize methods, the use of pitfall and funnel traps, drift fence techniques, nighttime road cruising, breeding call surveys, and the assessment of amphibian breeding site quality.

The methods are geared toward assessing the entire herp community in a given area. Often, a study focuses only on a particular species, which will define the techniques employed. Much of what I say concerning herp community assessment will be useful in these more restricted studies.

Before beginning a survey, decide the questions you want to answer and realistically appraise the time, effort, and money you are willing to spend. Among the basic questions that a general herp survey should address are:

1) WHAT KIND?
   = Species Richness: What species are present?
2) HOW MANY?
   = Relative abundance: How many individuals of each kind of species are present?
3) WHERE ARE THEY?
   = Habitat utilization: How do the species present use the available habitat?

Beyond these basic questions are a multitude of more detailed ecological questions (e.g., estimates of population sizes, reproductive success of individuals, food habits, etc.) that are beyond the scope of this introduction to field herpetology. References are provided in Appendix III.

Where and When to Survey Herps

The goal of a survey is to collect as much information as the investigator's constraints of time, effort, and money allow. With any survey technique, consideration of basic herp biology can reduce frustration and increase the quality and quantity of the data collected. Obviously, the best opportunity to collect and observe herps is when they are active. Three main factors are important: seasonal activity, weather, and habitat.

Seasonal activity

The herp activity calender (Table 4) indicates the major annual life history events for herps in Minnesota. This schedule provides a general framework for a survey program. A survey based on a single collecting period of one or a few days may be of little value because of the seasonality of herp activities and the influence of weather. Ideally, a survey consists of a series of data collecting periods distributed throughout the herp activity season to coincide with periods of maximum activity and optimal weather conditions. Often, constraints of time or money make such an ideal plan impossible. All things considered, spring and early summer (late March-June) is the best time to concentrate survey effort. During this period all herp species should be active. Vogt and Hine (1982), based on their experience in Wisconsin, recommend the following schedule of four census periods:

1) Early April: salamanders and frogs moving
2) Late April-Early May: frogs and snakes moving, fewer salamanders
3) Late May: lizards and snakes moving, fewer salamanders and frogs
4) Late May-June: turtles nesting, lizards and snakes

Ordinarily, survey periods should last several days and coincide with rainy periods. My Minnesota experience agrees well with Vogt and Hine's Wisconsin recommendations. The precise timing of the survey must be adjusted for a particular area. As noted earlier, salamanders and frogs may be moving in late March in southern Minnesota, but not until late April in northern Minnesota. This schedule should also be modified according to the distribution of water resources in the survey area (see Habitat below).

It is not necessary to engage in a long spring-through-fall survey to produce a reasonable species list for an area. But, population estimates or habitat use descriptions based only on survey periods coinciding with spring (or fall) movements can be misleading. Herps often concentrate in large numbers during these periods and travel through habitat where they are not normally abundant. A survey that includes summer sampling is more likely to give a true picture of habitat use than a survey conducted during reproduction or overwintering movements.

The appearance of young-of-the-year herps may produce a dramatic increase in the numbers of certain species near natal ponds when metamorphosis or hatching occurs during the summer. These juvenile additions to local populations are easily recognized; I recall walking at Wood Lake Nature Center (Minneapolis) one July evening when it was difficult to avoid inadvertently stepping on tiny, newly metamorphosed American toads.

Weather

Weather superimposes patterns of local activity onto general seasonal activity patterns. Temperature, precipitation, humidity, soil moisture, and wind all influence herp activity. Of these, temperature and precipitation have the most noticeable effects. The following general guidelines are based on Vogt and Hine's observations (1982) in Wisconsin and my own survey experiences in Minnesota.

Amphibian movements are strongly correlated with periods of high humidity or rainfall, so they can most efficiently be collected within 24 hours after rains. Expect to see amphibians if air temperatures are greater than 4 °C. I have seen wood frogs crawling towards breeding ponds in the early spring; even if prodded, they were apparently too cold to hop.

Lizards and snakes also move after rains, but they usually do not respond as rapidly as amphibians. They will wait for temperatures to increase, 21 °C or higher being good for lizard and snake activity. Overcast, humid days above 15 °C are ideal for catching aquatic turtles in terrestrial habitats (Vogt and Hine, 1982). Sunny days without too much wind are excellent for observing turtles basking near water. Extremely windy conditions curtail herp activity in general, even if air temperatures are favorable.

Thus, for both amphibians and reptiles, it is wise to concentrate survey effort on periods associated with rain. Herps will often seem to disappear during drought. In northern Minnesota, I have gone for many days without trapping herps during dry weather followed by dramatic increases in captures after even light rains.

Habitat

The location of water resources within the survey area helps determine how and when to census. Vogt and Hine (1982) provide the following general guidelines for upper midwestern states:

1) Within 3 km of permanent water: expect all major herp groups.
2) Farther than 3 km from permanent water, but within 3 km of temporary ponds, marshes, or streams, or in heavily wooded area: expect snakes, lizards, frogs and salamanders.
3) Open grassland or savannah more than 3 km from any open water: expect only lizards and snakes.

For example, if you were conducting a survey in a prairie far from any water, early to mid-spring sampling periods would be unnecessary (lizards and snakes are not very active then). It would be better to concentrate sampling effort in late spring-early summer (mid-May to June), using techniques most efficient for snakes and lizards.

Clearly, an appreciation of herp biology can reduce wasted effort and increase the amount of data collected. Effective herp study requires a naturalist's common sense. Thus, early April in southern Minnesota is an appropriate time to observe amphibian movements and frog chorusing, but the effort would be wasted on a particularly cold, windy night.

Search-and-Seize Methods

General search-and-seize

Objective: Species list.

Method: The simplest way of generating a species list for an area is to keep track of herps encountered by chance during routine activities on the survey area. This opportunistic collecting can be supplemented by active searching for herps as time and interest permit.

Comments: This method is effective if there are no time constraints on the survey and the observer resides on the survey area or visits it often. The observer must take the time to identify and record what he or she or others find on the survey area. Little time or effort is put into such a cumulative survey. The diligent observer will also develop qualitative impressions of relative abundance and habitat use.

The value of this method depends on the quality of the observer and the number of "contact hours" spent on the survey area. The resident manager of a park who is interested in herps, but who lacks time or money to devote to a quantitative survey could use this method effectively to assemble a species list.

Data presentation: Data are presented as a species list. General notes should be provided on sampling effort and qualitative estimates of relative abundance and habitat use.

Time constrained search-and-seize

Objectives: Species list, relative abundance, habitat use.

Method: Campbell and Christman (1982) describe this method. Select habitat to be sampled (e.g., oak forest, marsh); then search intensively each habitat using the basic techniques described earlier. No spatial boundaries are set other than staying within the habitat. Set a time limit for the search; one to six man-hours might be used depending on the situation.

Record the species identity of each specimen captured or observed and any other biological information of interest. Release animals at the site of capture/examination unless collections are being made for some specific reason. Avoid recounting animals by not searching areas twice.

Comments: This method, a step up from general search-and-seize, allows a quantitative comparison of species richness, relative abundance, and habitat use between sites. Because only time, not area, is constrained, this method is somewhat less rigorous for comparative purposes than the quadrat method (see below). Depending on the searcher(s) and the habitats sampled, there could be significant differences between sites in the amount of area searched. On the other hand, biases introduced by choice of quadrat site are avoided. As always, the biases introduced by season, time of day, and weather must be considered.

Data presentation: The data can be expressed in a number of ways depending on what you are interested in showing. See Table 5 for more examples.

1) number species/total search time/habitat
2) number individuals/total search time/habitat
3) number individuals/unit search time/habitat
4) number individuals/species/unit search time/habitat

(Example: 95 herps collected in six man-hours in a forest would be presented as 15.8 individuals/hour in a forest.)

Quadrat search-and-seize

Objectives: Species list, relative abundance, habitat use.

Method: Campbell and Christman (1982) describe this method. The idea is to select sites of a specific size (quadrats) within the survey area and attempt to collect all herps within the quadrats in some set period of time.

Study the survey area and choose the habitats to sample. Choose one or more sites not near each other within each habitat. Choose a standard size quadrat for your sampling (100-1000 m^2 is typical). Mark quadrat boundaries using nylon string and stakes. The seasonal and weather induced biases discussed earlier must be considered in planning the survey program.

On census days, one or more herpers intensively search the quadrats using the basic techniques described earlier. Record the species identity of each specimen captured or observed on the quadrat during the search. Record other biological information of interest. Keep animals as you collect them to avoid counting them twice. Return specimens to the quadrat if you are not collecting them for scientific purposes. Decide in advance the amount of time to be devoted to each quadrat so that a similar amount of sampling effort will be expended. Campbell and Christman (1982) write that six man-hours per quadrat is sufficient to guarantee that a quadrat has been "cleaned out."

Comments: For quadrat comparisons to be valid, it is important that approximately the same effort be put into each quadrat censused. The method is biased by the season, time of day, weather conditions on the census day(s), and skill and tenacity of the searchers. Another bias is in the quadrat itself; it may not be representative of the habitat. These biases can be minimized by doing several quadrats in a habitat and having several sampling periods.

Data presentation: Data presentation is similar to the time constrained method except effort is expressed per unit area.

1) number species/total area searched
2) number individuals/total area searched
3) number individuals/unit area searched/habitat
4) number individuals/species/unit area searched/habitat

(Example: 100 green frogs collected in a 1000-m^2 quadrat in a marsh would be presented as 0.1 green frogs/m^2 in a marsh.) Table 5 presents a hypothetical set of data and how it might be presented.

TABLE 5

Presentation of hypothetical data for three herp survey methods. For the purposes of demonstration, each of the three sampling methods is considered equally effective.

HABITAT	SPECIES			SAMPLING EFFORT		
	Number of Species	Identity	Number of Animals	Quadrat* (# animals/sq. m.)	Time** (# animals/hr.)	Fence*** (# animals/trap day)
FOREST	3	A	50	.05	8.3	5.0
		B	25	.03	4.2	2.5
		C	20	.02	3.3	2.0
			95	.10	15.8	9.5
MARSH	2	A	100	.10	16.7	10.0
		D	10	.01	1.6	1.0
			110	.11	18.3	11.0
PRAIRIE	1	E	75	.08	12.5	7.5
TOTAL	5	A,B,C,D,E	280	.09	15.6	9.3

*Quadrat = Quadrat search-and-seize method, one 1000 m^2 quadrat in each habitat.

**Time = Time constrained search-and-seize method, six man-hours in each habitat.

***Fence = Drift fence trapping, one drift fence array in each habitat, drift fences open 10 days in each habitat.

Trapping Techniques

The survey methods discussed so far require the active participation of an observer/collector. There are trapping techniques that eliminate some of the problems and biases of manual survey methods (but also produce complications of their own).

The manual methods are relatively inexpensive, requiring time and manpower rather than equipment. The trapping techniques described below can demand a considerable expenditure of funds (depending on the extent of the sampling program). They represent a sound investment because the equipment will last for years if properly cared for. Trapping is probably not the technique of choice if you are involved in a short term survey program.

Pitfall and funnel traps

A pitfall trap is a container, open at the top, and buried in the ground so that the opening is flush with the surface. Animals that fall in are trapped unless they can climb out. A funnel trap is a container that rests on the substrate with one or more funnel-like openings (usually one at both ends). Animals can enter the funnel trap through the opening, but once inside find escape difficult (Fig. 15).

Isolated pitfall and funnel traps, deployed by herpetologists who choose trap sites aided by specialized knowledge of herp behavior and habitat use, can catch herps (e.g., Shields, 1985). But uninformed scattering of pitfall or funnel traps around the environment by the nonspecialist would be ineffective.

Drift fence trapping

Objective: Species list, relative abundance, habitat use.

Methods and materials: Pitfall and funnel traps work most effectively when combined with an artificial barrier. The barrier intercepts animals moving through the environment and directs them toward the traps placed along the barrier. These artificial barriers are called drift fences and are the standard tool for sampling herp communities.

Drift fence trapping is by no means restricted to herps. Large numbers of arthropods and some small mammals will also be caught. I have even recovered birds from drift fence pit traps!

Recently, three review papers on the use of drift fences as a herpetological sampling tool have appeared (Campbell and Christman, 1982; Gibbons and Semlitsch, 1982; and Vogt and Hine, 1982). The following discussion is based on these papers and my own drift fence experience in Minnesota and Indiana.

Ideally, drift fence material should be an effective herp barrier, strong, flexible, lightweight, durable, and inexpensive. Among the materials employed by herpetologists are aluminum flashing, sheet metal, aluminum window screen, chicken wire, hardware cloth, plastic-coated screen, tarpaper, and pressure-treated boards.

The authors cited above and I all prefer aluminum flashing (also called aluminum valley, valley flashing, valley tin). This is a thin gauge aluminum sheeting that can be purchased in rolls; it is relatively inexpensive, lightweight, flexible, and extremely durable. Aluminum flashing is available from hardware stores or building supply companies. Flashing comes in several gauges. I have found .019 gauge to be a good weight; thinner gauge flashing is flimsy and must be supported with stakes.

A drift fence is made by partially burying a length of aluminum flashing (usually 15-30 m) in the ground and placing pitfall or funnel traps adjacent to the fence. The fence should be about 50 cm tall, with approximately 10 cm buried in the ground. If needed, 1-m angle iron stakes (or stakes of other material) can be used to anchor the fence at both ends and elsewhere. The stakes are secured to the fence with wire or

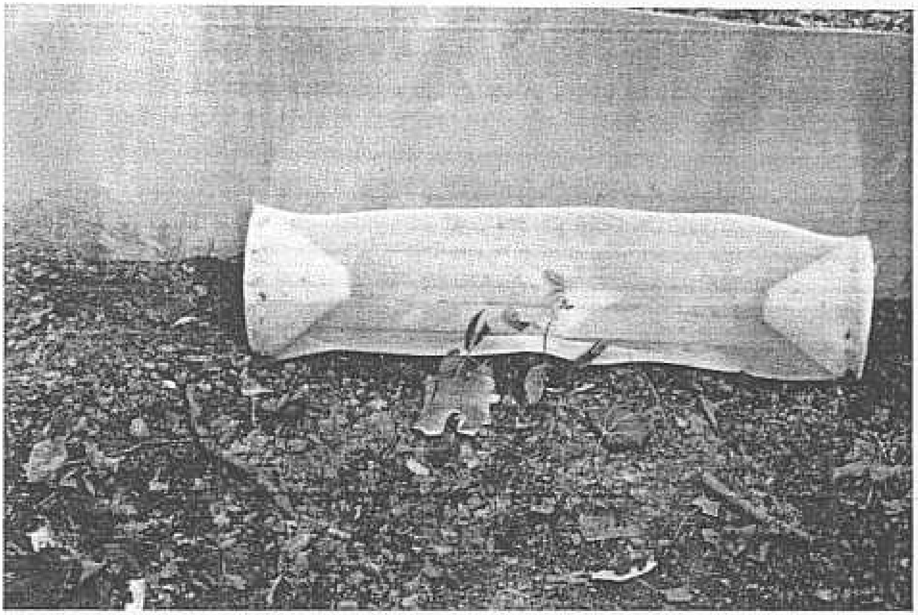

Figure 15. Funnel trap placed along a drift fence. The trap is a 76-cm long cylinder of aluminum window screen stapled to a wood strip, with plastic funnels at either end. Animals are removed with long forceps or tongs.

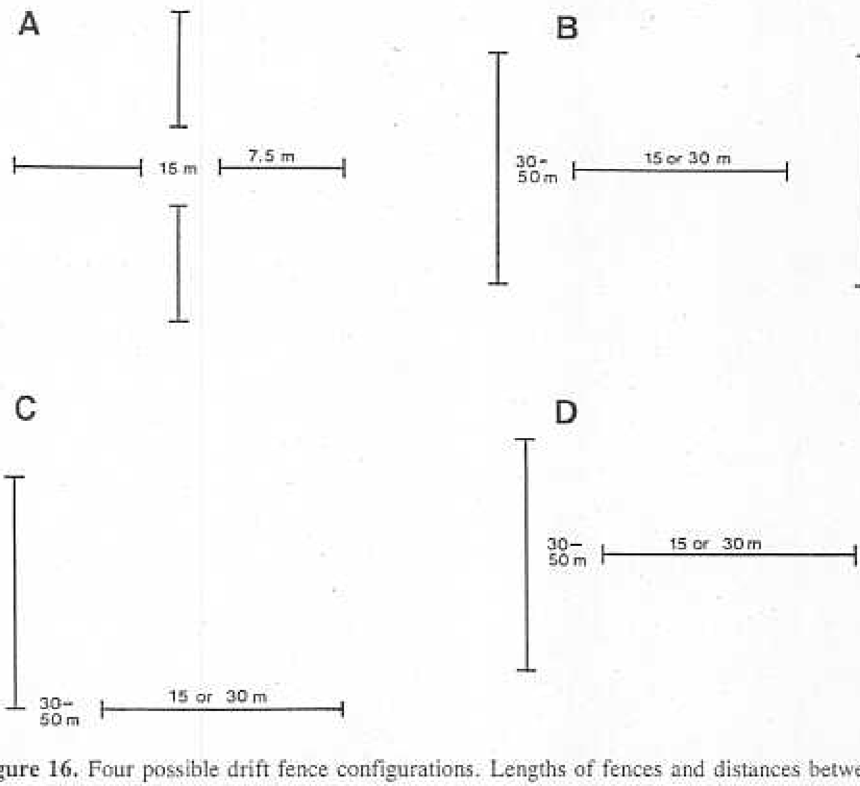

Figure 16. Four possible drift fence configurations. Lengths of fences and distances between them are shown. A is the "+" design (Campbell and Christman, 1982); B is the "H" design (Vogt and Hine, 1982); C is the "L" design and D is the "T" design (Karns, 1979). See text for discussion.

plastic cable. In most soil types, staking is not really necessary with .019 gauge because the fence is rigid enough to stand by itself when buried.

A number of options are available for pitfall and funnel traps: 20-*l* (5-gallon) plastic buckets, 7.6-*l* cans (two 3-lb metal coffee cans with or without funnel rims), aluminum window-screen funnel traps. Construction of these traps is described below. The 1985 cost of one 15-m fence unit with a standard array of traps would be:

One roll 15 m × 50 cm .019 gauge valley flashing	$45.00
Two 20-*l* buckets @ $3.00	6.00
Two 76 cm × 65 cm aluminum window screen funnel taps with plastic funnels	6.50
Eight 3-lb coffee cans (scrounged at no cost)	0.00
Total	$ 57.50

Details of Drift Fence Technology

Length, placement, and configuration of fences

Vogt and Hine (1982) tested the trapping effectiveness of various lengths of fence from 3 to 60 m. Not surprisingly, longer fences caught more animals. Their data show that fences less than 15 m long have capture rates too low to make them worthwhile. Campbell and Christman (1982) used four 7.6-m fence lengths in a "+" shaped array. I employed 15-m lengths singly or in pairs. Fifteen to 30 m for each trapping site seems to be a practical length of fence.

How should the fences be deployed? Consider an extensive patch of woodland habitat. If the investigator decides to place one 15-m strip of fence running in an east-west direction in the center of the woods, the sample will be directionally biased. Animals moving east or west would be unlikely to encounter the fence, and if they did would hit it edge on. A north-south fence would trap more animals moving in an east-west corridor. Gibbons and Semlitsch (1982) and Campbell and Christman (1982) suggest an open "H" design. I employed an "L" or "T" design (Fig. 16). All these configurations will help eliminate directional bias in trapping. Your budget and time constraints may not allow multiple fences at each trapping site. Do not let these directional considerations prevent you from using drift fences. One 15-m fence in the middle of a woods is better than no fence and will catch herps for you.

Where should the fences be deployed? Habitat borders are important zones to sample, especially aquatic/terrestrial boundaries when herps are moving (e.g., during amphibian breeding movements, feeding forays, overwintering movements, turtle nesting movements). A "+," "H," "L," or "T" shaped fence array can be put in at habitat boundaries. One segment of the fence array should be parallel to the habitat boundary and the other(s) perpendicular. However, because most movement is probably across the habitat boundary, one fence parallel to the border may be sufficient, depending on the investigator's interests.

For example, I constructed a drift fence adjacent and parallel to the edge of a pond in northern Minnesota; I caught 48 wood frogs in late April, 1979. Forty-five were caught on the side of the fence facing the adjacent woodland and three on the pond side. This strongly side-biased trapping was due to spring breeding movements. My primary goal was to intercept amphibians coming to breed, so fences set perpendicular to the border would not have been effective. Breeding ponds have been entirely surrounded by drift fences in a number of studies (e.g., Semlitsch, 1985; Stenhouse, 1985), a technique that gives a complete record of movements into and out of the pond.

The interior of patches of relatively uniform habitat should also be sampled, using the directionally unbiased fence arrays shown in figure 16, if possible.

The number of fence sites employed within and between habitats will depend on your budget and manpower constraints. Figure 17 shows the placement of drift fences for a hypothetical piece of land and illustrates some of the points discussed above.

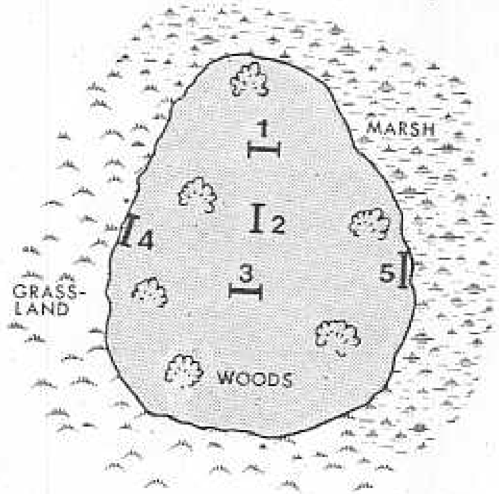

Figure 17. Hypothetical drift fence placement. The primary objective is to sample the large wooded area. Fences are not to scale with the habitat. An "H" array (1,2,3) is placed in the center of the woods where there is no clear habitat gradient. Single fences (4,5) are placed parallel to habitat border on edge of grassland and marsh to intercept movement between habitats (modified from Vogt and Hine, 1982).

Pitfall and funnel traps

How many and what kind of traps should be used with drift fences? Campbell and Christman (1982) use 20-*l* plastic buckets and funnel traps in all situations (Fig. 18). They suggest putting one bucket at the end of each fence arm, cutting a slot into it so that the fence overhangs it slightly. A masonite board (raised 5-8 cm) placed over the buckets provides shade and protection. Thus, half the bucket is on each side of the fence. This technique reduces the number of buckets needed, with the drawback that one doesn't know on which side of the fence animals were intercepted. Gibbons and Semlitsch (1982) also use 20-*l* plastic buckets in all situations. They place buckets on both sides of the fence and at both ends of each fence segment in the array (Fig. 18). Vogt and Hine (1982) suggest various trap arrays depending on the distance of the fence from standing water, thus taking into account the fact that amphibians drop out of the sample with increasing distance from open water (Fig. 18).

To construct a smaller (7.6-*l*) pitfall trap, attach two #10 cans (3-lb coffee cans). Remove the lids from both ends of one can and one end of the other. Attach the cans with duct tape. This makes a durable, 36-cm deep pitfall trap. If the site is particularly wet, you may want to put silicone sealer around the inner seam. A one-pound tub of butter with the bottom (and the butter) removed makes an effective funnel rim.

There are a number of possible funnel trap designs for use with drift fences (Vogt and Hine, 1982; Campbell and Christman, 1982). I have used 76-cm long funnel traps made from aluminum window screen.

To construct a funnel trap, cut a 65-cm long strip of 76-cm wide screen. Roll this into a 20-cm diameter cylinder (Fig. 15). Attach to a 70-cm fir strip with a staple gun. Campbell and Christman construct funnels from aluminum screening. I use 20-cm diameter plastic funnels with the bottom cut off (leaving a 4-cm opening). Attach the funnels to the screen cylinder with a pliers type staple gun. Animals are easily remov-

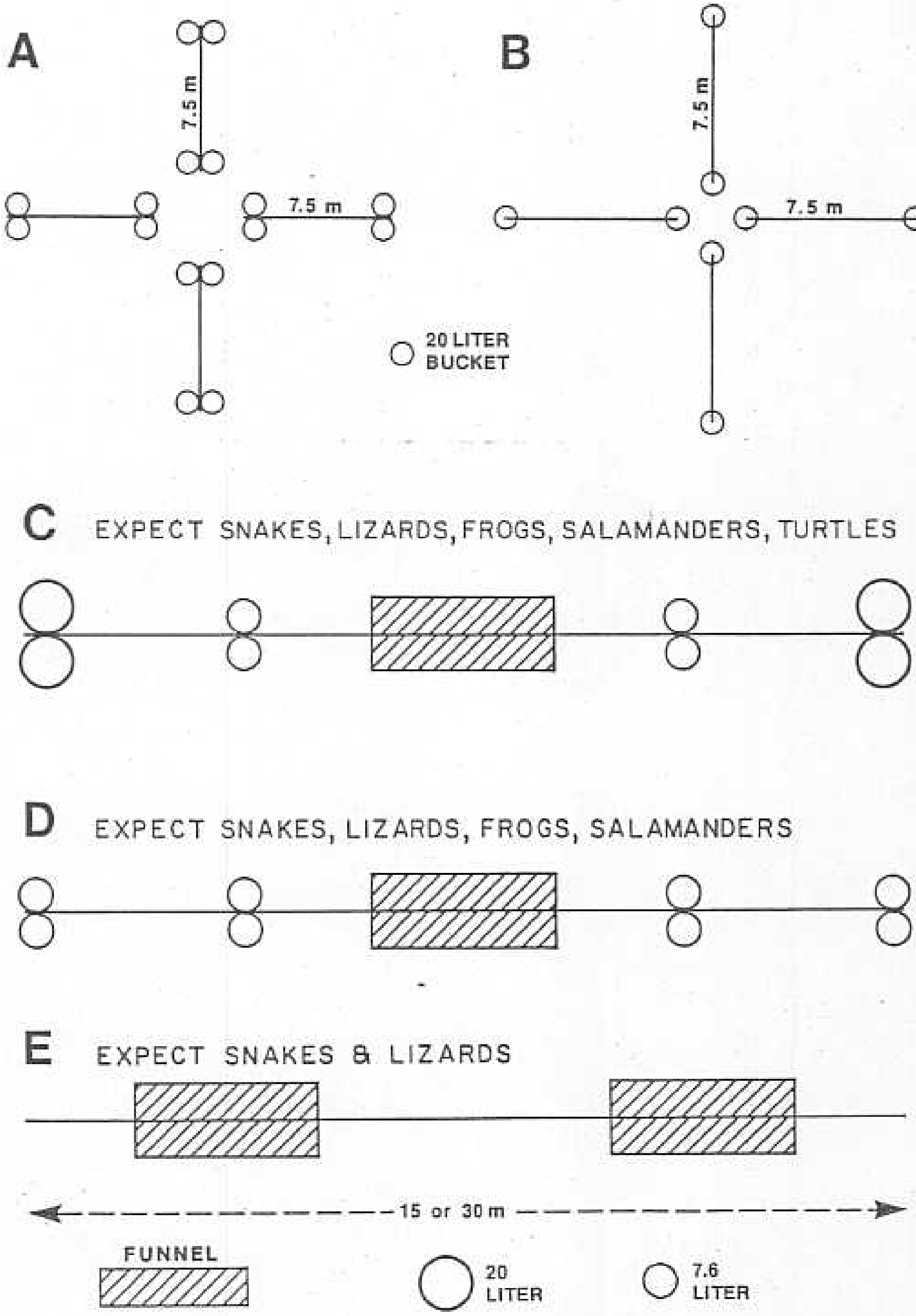

Figure 18. Possible placement of pitfall and funnel traps along drift fences. Design "A" (Gibbons and Semlitsch, 1982) and design "B" (Campbell and Christman, 1982) show "+" shaped arrays of 7.5-m fences with 20-*l* buckets only. Designs "C," "D," and "E" (Vogt and Hine, 1982) show possible deployment 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.

ed from the trap with long forceps. Alternatively, one funnel can be clipped onto the screen and completely taken off to remove animals.

Vogt and Hine (1982) tested the effectiveness of funnel and pitfall traps for catching amphibians and reptiles. Their results indicate that:

1) 20-*l* pitfall traps are required for adult turtles and are effective for small snakes, lizards, and amphibians.
2) 7.6-*l* metal cans are effective for amphibians and lizards. The addition of a funnel rim on the cans may improve the trap by inhibiting the escape of climbing herps (such as treefrogs). I found these cans (with or without rims) to be useful for small snakes as well.
3) 3.8-*l* cans are ineffective except for some hatchling turtles and toads.
4) Funnel traps are the most effective for lizards and the only reliable means of catching large snakes. They are also excellent for amphibians.

Vogt and Hine conclude that a system of 20-*l* and 7.6-*l* pitfall traps and funnel traps is needed to catch the array of herps encountered. None of the traps used in their study were extremely effective in trapping treefrogs. Recently, I have successfully caught treefrogs in the funnel traps described above.

Installation of fences

In addition to aluminum flashing and traps you will need a shovel, tin snips, and a square-headed axe. A post-hole digger is convenient for the pitfall holes, but a shovel will get the job done. If you plan to stake the fence you will need wire or plastic cable and an ice awl (or some other sharp object to puncture the aluminum). Use an appropriate length of rope to stake out a guideline for digging the fence trench.

You are now out in the field, you have all your equipment and have made the "theoretical" decisions about fence placement. The final placement nevertheless depends on practical considerations. You need a fairly level, open stretch to dig a 15- to 30-m trench. This is not always easy to find. Reconnoitor the study area in advance and have exact sites located and flagged.

Stake out the guideline and then dig a trench with one vertical side for easy fence placement. Remove soil in intact clumps if possible and keep these clumps to aid in refilling the trench and securing the fence. The trench should be about 10 cm deep and 20 cm wide. The fence can be cut with tin snips to accommodate immovable roots or rocks. Smaller roots can be chopped out of the way. Try to avoid cutting the fence.

After excavating the trench, but before putting in the fence, dig the holes for the pitfall traps. When trap holes are done, position the fence in the trench and secure with the dirt clumps or loose soil. Plant the pitfall traps in their holes and secure them in place with loose soil. The openings should be flush with the ground. Wedge the traps tightly against the fence for added bracing. For upland sites, Vogt and Hine (1982) recommend drilling holes in the sides of the can 2.5 cm from the bottom and leaving a space beneath the can to allow for overflow in case of heavy rains.

Brace the fence with angle iron stakes, if necessary. I prefer to place stakes next to the fence without actually attaching them to the fence. If you prefer, punch holes in the fence with an ice awl or other appropriate instrument and then pound the stakes into the ground and tie them to the fence with wire or cable.

Drift fences are easy to install at dry upland sites, but troublesome in wetlands or at aquatic/terrestrial boundaries. In northern Minnesota, I worked in water-saturated peat soils. The drift fence can be anchored in this kind of environment with a little wet, mucky effort; the real problem is with the pitfall traps. They will fill with water if they are not made waterproof. The 7.6-*l* cans can be made watertight by applying silicon sealant (available from hardware stores) around the inside seam between the two cans. The 20-*l* plastic buckets are, of course, already waterproof. Waterproof cans are buoyant and will pop out of their holes unless kept in place by weighting them down with stones, filling them with 5-8 cm of water, and wedging them firmly against the fence with wet soil. Once the wet soil "sets" around the fence they should remain in place.

Clay soils underlain by hardpan are very prone to flooding; these sites should be treated as water-saturated sites.

Fence maintenance

Maintenance chores are best accomplished during routine checking of fences for animals (Fig. 19). Keep vegetation around the fence well-clipped for ease of access and to prevent herps from being deflected from the fence. Erosion around cans and fences, especially after heavy rains, is common; fill in as necessary. Heavy rains can also make bailing necessary. Check water levels in cans routinely and maintain at desired levels.

A piece of sponge should be placed in each pitfall trap to prevent dessication if the water in the can evaporates or as an "island" in case of flooding. Wet sponges should go in funnel traps, too. Masonite boards propped against the fence will shield funnel traps that are susceptible to dessication.

Data collection

Prepare a data form for general use in checking trap sites. On the data form, provide spaces for:

1) location of the trap site
2) date
3) name of collector(s)
4) weather notes
5) other observations
6) a systematic method of recording data for each individual trapped with spaces for:
   a) species identification
   b) position and side of the fence where animal was caught
   c) additional biological information being collected (e.g., size, weight, sex)

Chapter Four contains a discussion of data collection techniques beyond species identification and the number of individuals caught.

After recording the data, release the captured animals a few meters away on the opposite side of the fence unless you have some specific reason for keeping specimens. Animals can be marked to avoid recounting the same animal. The simplest way to mark frogs, toads, salamanders, and lizards is toe-clipping in which one or more digits are removed with scissors or a nail clipper. Belly scales on snakes can be clipped. Turtles are marked by making notches on the margin of their shells with a file. For general survey purposes, when you need to know that you caught an animal previously, but not its individual identity, there is no need to use individual-specific codes; a general mark will suffice. Ferner (1980) provides information on the theory and practice of marking amphibians and reptiles for field study.

Sampling schedule

How often should fences be checked? Campbell and Christman (1982) checked their fence arrays weekly. Vogt and Hine (1982) suggest at least every other day and within 24 hours after heavy rains. Gibbons and Semlitsch (1982) recommend daily checking and even more frequent checking during amphibian breeding movements. I checked fences every two to four days and as soon as possible after rains.

When and for how long should fences be open for sampling? Leaving fences open for the entire season of herp activity may not be practical for a variety of reasons. The other option is a series of sampling periods designed to maximize trapping effort based on the basic considerations of herp biology discussed earlier in this chapter.

If you decide upon a series of sampling periods, fences should be open for three to five days during each period (Vogt and Hine, 1982). If you have some flexibility on when to open traps, make an effort to include periods with some rain, because of rain's well-documented influence on herp movements. Cover pitfall trap openings and remove funnel traps during periods when the fences are not being checked. In my northern Minnesota peatland survey, my fences were open continuously from 15 April to 15 June and for the first two weeks of each month from July through October.

Comments

Drift fence technology is relatively expensive and requires more initial planning and effort compared to simpler search-and-seize methods. Once the initial investment of time, money, and labor is made, however, drift fences continuously sample an area. This "24-hour-per-day herpetologist" avoids the sampling biases introduced by the quality of the collector and timing of the search that are unavoidable with search-and-seize methods.

Drift fence surveys do have problems that the potential investigator should recognize. In search-and-seize surveys, the collector combs the environment searching for herps. With drift fence methods, the "collector" (the fence) is stationary and the herps must come to the "collector" to be counted. The success of the fence in capturing animals will depend on characteristics of the organism's form and natural history, and on drift fence placement.

Some herps are not sampled effectively by drift fences regardless of the fence configuration and type of traps employed. Treefrogs, with their "adhesive" toepads, can crawl out of containers. In three years of drift fence trapping in northern Minnesota I caught one gray treefrog in drift fences although they were extremely abundant in the area. Large snakes also present problems. Gibbons and Semlitsch (1981) note that large snakes can easily escape from 20-*l* containers; adult black racers, abundant in their Georgia study area, were rarely caught in their drift fences. Funnel traps, however, do catch large snakes (Vogt and Hine, 1982; personal observation).

On the other hand, some herps may be overrepresented in a sample. Shields (1985) presents evidence that southern leopard frogs use pitfall traps as a refuge from dry conditions, terrestrial predators, and cold nights. Based on my experience in northern Minnesota, I suspect that breeding anurans vocalizing from within pitfall traps attract other anurans to the drift fence.

Herps that engage in periodic mass movements, that have large home ranges, or whose daily activities involve frequent movement—such as active foraging versus sit-and-wait predators—will more likely encounter drift fences. These factors coupled with fence placement provide a clue to what a fence can catch. Thus, a drift fence array set in the vicinity of an amphibian breeding pond in the spring will provide an excellent sample of the local amphibians that breed in temporary pools (except treefrogs). A more sedentary species in the same area that lays its eggs in moist terrestrial conditions will probably be underrepresented in the sample. Gibbons and Semlitsch (1982) provide an excellent discussion of the biases inherent in drift fence trapping and the interpretation of drift fence results.

Data presentation

The unit of sampling effort used with drift fence trapping is the number of days a particular fence or fence array is open (trap-days). Thus, using drift fence data one could report:

1) number species/total number trap-days/total survey area
2) number individuals/total number trap-days/total survey area
3) number individuals/trap-day/habitat
4) number individuals/species/trap-day/habitat

(e.g.: Five spring peepers caught in 10 trap-days in a marsh = 0.5 peepers/trap-day in a marsh). See Table 5 for more examples of drift fence data presentation.

Miscellaneous Survey Methods

Nighttime road cruising

Objectives: Species list, relative abundance, habitat use.

Method: Secondary roads at night can be used as transects to sample herps. This technique is an old standard for herpetologists. As described earlier, one drives at low speed along secondary roads that pass through habitats of interest. A record is kept of herps crossing the road and roadkills (Fig. 20). Nighttime road cruising for herps becomes a quantitative survey tool by simply recording the distance traveled in each habitat and the species identity and number of herps observed in each habitat.

Comments: This economical and convenient—as long as suitable roads are available—survey method can provide considerable data for the time and manpower investment.

As with other herping methods, the timing of the cruise with respect to season, weather, and time of night will influence the productivity of the method. Herps that are primarily diurnal (active during the day) will probably not be encountered (e.g., northern prairie skink), and more active, nocturnal herps with large home ranges will probably be encountered most frequently. As noted earlier, large numbers of herps can be observed by nighttime cruising on warm, rainy spring and fall nights in suitable habitats when breeding and overwintering movements are taking place.

Given suitable cruising conditions, the success of this method is a function of the distance traveled. For example, Campbell and Christman (1982) detected 38 species in 2,903 km of nighttime road cruising in a Florida survey, approximately the same number of species they detected using drift fences in the same area (43 species in 7,432 trap days using 30 "+" shaped fence arrays). Three of the species they found by cruising were not detected by any of the other techniques they employed.

Information on patterns of habitat use and relative abundance can be obtained if particular areas are repeatedly cruised. For example, my nighttime cruising in northern Minnesota showed that American toads were far more common than wood frogs during the summer in relatively dry, black spruce bog forests, but less abundant in wetter, tamarack swamps. Data from drift fence arrays in these habitats corroborated the nighttime road cruise findings.

Data presentation: The basic unit of sampling effort in nighttime road cruising is distance traveled (kms or miles). Thus data could be presented as:

1) number species/total distance traveled
2) number individuals/total distance traveled
3) number individuals/distance traveled/habitat
4) number individuals/species/distance traveled/habitat

(e.g.: 55 wood frogs collected in three km of driving through a bog habitat would be reported as 18.3 wood frogs/km in a bog.)

Breeding call surveys

Objectives: Species list of breeding anurans, identification of important breeding areas, relative abundance of different species present.

Method: Anuran breeding sites can be located in the early spring by car cruising or walking through areas where there is sufficient standing water for breeding to occur. Select a number of census sites; these sites will be used throughout the survey in a given year and in future years if the survey is continued. It is important that the sites be accessible and well marked on topographic maps for future reference.

During a survey, go to each site, stop, and remain quiet for a few minutes (to allow chorus activity to resume if you have disturbed it). Then listen for some predetermined period of time (5-10 minutes is reasonable). Record the identity of the species that are calling.

In addition to identifying species, one can make a subjective quantitative index to score the relative intensity of calling activity. Rate the level of calling for each species at each site from 0-5 (or some other scale of your choosing). I have used the following scale:

0 = absence of calling
1 = single individual calling
2 = occasional calling by several individuals
3 = low intensity, relatively frequent calling
4 = medium intensity, continuous calling
5 = high intensity, continuous calling

Such an index allows comparison within and among sites for different species.

Due to differences in the commencement and duration of calling by different species, it is necessary to visit sites several times. Keep in mind that even in the same general area there can be considerable variation in the onset of breeding because of microclimatic differences among sites. In northern Minnesota I observed a two week time lag in the initiation of spring calling at a shady woodland pool compared to a nearby pool with open, sunny southern exposure. Surveys should be performed only on nights when temperature and weather conditions are favorable for calling.

Ray Anderson and Deborah Jansen at the University of Wisconsin-Stevens Point have been censusing the anurans of Portage County in central Wisconsin for several years (Hine, 1982). They have developed a calling calender that indicates the approximate period of calling activity for different species and suggests the time periods and temperatures most favorable for call surveys (Fig. 13). I have found this calender to be a good general guide for anuran calling activity in central and southern Minnesota.

Anderson and Jansen believe that a mimimum of three surveys (one during each of the periods indicated on the graph) is necessary to hit the peaks of calling of the different species. Their survey program includes ten wetland sites and a five minute listening period at each site. Obviously, more surveys conducted over the course of the breeding season will provide more detailed information and help eliminate weather induced biases and differences due to microclimatic variation among pools.

Comments: An obvious prerequisite for call surveys is the ability to identify the calls of Minnesota species. This is a simple task compared to bird calls; there are relatively few species in a given area, the calls are simple, and there is a definite sequence to calling activities (see Appendix III for information on how to learn frog calls).

Frog calls cannot be used to census the number of frogs in a breeding population (Vogt and Hine, 1982). Only males call and not all the reproductive males at a site may call. Also, in active choruses, counting all the individual callers would be impossible. The call intensity index suggested is too subjective and dependent on sampling conditions to reliably indicate slight differences in relative abundance among sites. Nevertheless, large differences can be discerned using a call intensity index.

Call surveys are a simple way of determining what species of anurans occur in the survey area and which sites are important to breeding populations. If done consistently during the active season, call surveys can provide information on the status of different species in the areas sampled.

Data presentation: Breeding call surveys produce a species list. For quantitative comparison among sites, the call intensity index must be used. For example, in my Minnesota peatland study, I suspected that breeding amphibians used bog-water pools less than fen (peatland equivalent of a marsh) pools. I chose seven bog and six fen sites to census and conducted 7-10 surveys per site over the breeding season. The results were clearcut: Fen sites were favored by breeding amphibians (average call intensity scores, wood frog: bog = 1.3; fen = 3.3; chorus frog: bog = 0.7; fen = 3.7; data from Porter Ridge Bog, Koochiching County, 1979). These results were further supported by drift fence trapping and counts of wood frog egg masses. The standard error or standard deviation should be supplied with mean values.

The value of frog call surveys: Frog call surveys can be an important tool in assessing environmental deterioration. Ruth Hine (1982) of the Wisconsin Department of Natural Resources points out that anurans are sensitive to changes in water quality and quantity. A serious decline in frog populations is a warning that something is happening in the environment and warrants further investigation.

Some population declines may, of course, be perfectly natural. For example, cold winters with relatively light snowfall could cause heavy winter mortality in species that overwinter in shallow surface depressions. Severe spring droughts can prematurely dry up breeding pools and kill off the larval population for that year. This would reduce the size of breeding populations in future years.

The cause of population declines may be difficult to establish. Northern leopard frogs underwent a dramatic decline in abundance in the United States and Europe in the past decade, posing a serious problem because they are used widely in scientific research and in biological education. Populations are now increasing, but despite intensive study by researchers in Wisconsin, Minnesota, and elsewhere, no single reason for the decline has emerged. Degenerative liver changes indicated that a toxic substance might be involved (Hine et al., 1981). Disease, natural or man-induced, is also suspected; frog autopsies indicated that a high percentage of frogs sampled during the decline had a form of kidney cancer. The cancer seems to be absent in currently expanding populations (McKinnell et al., 1979). More recently, cricket frogs are now undergoing a mysterious and dramatic decline in Wisconsin. Regular call surveys may pick up such changes early in their development.

The Wisconsin Phenological Society initiated a "Frogwatch" program in 1981, conducting 27 surveys that year in 22 counties throughout Wisconsin. If made an annual event, like the Audubon Christmas Bird Count, the program would provide valuable information on anuran populations in Wisconsin. It would be worthwhile to develop a similar program in Minnesota.

The assessment of breeding site quality

Objectives: The presence of calling frogs does not necessarily mean that a site is suitable for successful amphibian reproduction. The following methods can be used to monitor the quality of ponds for embryonic development, hatching, and larval survivorship.

Method: The conspicuous globular egg masses of wood frogs and northern leopard frogs, laid in clusters at "communal" deposition sites, are a useful survey tool. Use calling activity to locate egg laying, then stake each breeding site so that it can be relocated. The number of egg masses in the typical clusters at any given site can be counted. The census should be done at least twice at each site over the relatively brief breeding period to insure that the majority of eggs laid are counted. Data can be presented as egg masses/unit area if an estimate of the area searched is made.

As eggs develop at the sites, the globular egg masses lose their shape, making them difficult to count. Thus, egg mass surveys must be done early in the season. This method provides a direct measure of the ecological importance of a pond.

Blue-spotted and tiger salamanders also lay eggs in globular masses that can be observed and counted. Blue-spotted females usually deposit eggs in several small clusters of one to 35 eggs, attached to vegetation. Tiger females usually lay a loose globular mass of 18 to 110 eggs near the bottom of a pond.

Egg masses can be monitored for normal development and hatching. This is especially easy for explosive breeders (e.g., wood frog, American toad) that lay large numbers of eggs in conspicuous sites (Fig. 14). Large numbers of dead eggs in many egg masses may indicate a problem with water quality.

Breeding ponds can be sampled for the presence and abundance of larval populations using dipnets, seines, trays or modified minnow traps. These methods are most effective early in the season when young larvae are most abundant. As the season progresses larval populations decline due to disease, predation, and competition. Larger, older larvae are more wary and harder to catch. Different species of larvae will be most abundant at different times depending on when breeding occurs.

For example, I used the "tray-plunge" method (Chapter Two) to sample amphibian larvae. I chose a number of sites of known breeding activity and performed 30 tray plunges at each site. I preserved the larvae for later identification and presented the data as larvae/plunge (see Appendix III for information on how to identify larvae).

Drift fencing can be used to determine if successful amphibian reproduction is occurring. Drift fences set parallel and immediately adjacent to known breeding areas will catch recently metamorphosed salamanders and anurans emerging from the natal ponds in the summer. The productivity of different breeding sites can be compared using this method.

Aquatic methods

I have already described a number of techniques that are useful for collecting or observing herps in aquatic situations (Chapter Two). These methods can be turned into quantitative survey tools by keeping track of the amount of time or effort put into the sampling.

For example, turtle traps might be deployed at several ponds and left out for four days with periodic checking (Fig. 21). The capture rate over this period of time would provide a basis of comparison among sites.

Specialized studies

This book is a guide to general surveys, but a study may focus on only one species or one group. The project's objectives and the species' characteristics will then determine the methods employed.

For example, Jeff Lang of the University of North Dakota investigated the ecology of the state-endangered, five-lined skink in western Minnesota. The lizard's favored habitat is granitic outcrops. Drift fences are not appropriate in such habitat; Lang successfully collected lizards by overturning the rocks they use for shelter. This was no easy task: the lizards favored the larger rocks that could only be moved by two or three people while another person stood by, ready to grab the surprised lizard before gravity won the contest with the rock. Lang (1983) has written an interesting popular account of his skink study.

Another specialized project is the Spring Peeper Program undertaken by the Hennepin County Park Reserve District. Spring peepers do not breed in a number of county reserves although conditions seem suitable and it is likely they bred in those reserves

Figure 19. Two young herpetologists check a drift fence. The fence is 30 m long and made of 50-cm wide aluminum flashing. The boys are examining one of the funnel traps along the fence, which is adjacent and parallel to a small stream.

in the past. The Wildlife Department of the District decided to introduce spring peepers into three different Park Reserves. The primary reason for the project was aesthetic; the District wanted to restore a pleasing component of the regional fauna.

Three years into the program, it appears to be working. Frogs were collected by hand and by drift fences from known peeper breeding areas in the Carlos Avery Wildlife Management Area. Five hundred eighty-nine peepers were released in 1983 in the three county reserves. The "Peeper Project" is an excellent example of herping techniques being employed for a specialized purpose. I have never heard of a similar program, though wildlife managers are always introducing large mammals or birds to some area where they have been extirpated.

Comparison of Methods

The most extensive comparative study of herpetological survey methods is that of Campbell and Christman (1982). They compared quadrat search-and-seize, time constrained search-and-seize, and drift fence array methods in four habitats (Table 6). These researchers expended approximately equal effort on each of the methods. The drift fence array was clearly the overall winner, in terms of both number of species detected and number of animals caught. Drift fences were somewhat less effective than hand collecting in detecting species of treefrogs in the flatwoods and hummocks, where several species of broadleafed shrubs and trees harbored them.

Campbell and Christman detected 38 species by cruising roads at night in several habitats, including three species not collected any other way. Drift fences were slightly superior to nighttime road cruising in detecting species if all 30 arrays deployed in 11 habitats were included; 43 herp species (1,644 specimens) were collected in 7,432 trap-days in the 11 habitats. This comparison indicates that nighttime road cruising is a powerful (and cheap) species detection tool. Drift fence arrays, however, can provide more comparative information on relative abundance and habitat use.

Figure 20. Cruising for roadkills and live herps crossing the road can be an effective, cheap way of herping. The author is examining a snake roadkill in southeastern Minnesota.

Figure 21. Turtle trap deployment. This commercial turtle trap is a nylon mesh cylinder supported by metal hoops with a funnel opening at one end. After baiting it, the author is staking it to the bottom of the pond near a western painted turtle basking site.

One might think that these various formal methods in combination would have detected most of the species present in the areas sampled. However, Campbell and Christman found an additional 25 species by opportunistic search-and-seize herping. Thus, they point out that the various formal survey methods do "not fully replace the snake collector with potato rake and cloth sack if maximal information on faunal composition is one of the objects of the survey."

In Wisconsin, Vogt and Hine (1982) compared species lists generated by drift fence trapping with those generated by other methods and found drift fences to be effective. In one area, with a five year history of search-and-seize herping, drift fences detected two species suspected to be in the area but never collected (tiger salamander and slender glass lizard).

Vogt and Hine's survey also showed clearly the impact of extended dry periods on herp surveys. At the Spring Green site, 12 species were known to be present. In two months of drought, drift fence trapping revealed only two species and 15 hours of intensive search-and-seize revealed only three species.

I recently conducted a herp survey on a large tract of land for a private consulting firm. Due to budget constraints my survey involved only 10 hours of on-site inspection. I employed the time constrained search-and-seize method in different habitats in the area. It was necessary to perform the survey in late summer, which as discussed earlier, is not a "prime time" for herp surveys. Ten species were known from previous late summer or fall surveys using drift fences, opportunistic collecting during routine field work, and nighttime road cruises. My short-term survey produced only four of the ten species.

These comparisons make it clear that a variety of techniques, used under a variety of conditions, are needed to develop an accurate picture of the herpetofauna of a given area. If a short-term survey is performed when conditions are not favorable, one may find very little. If the author of a report based on such a survey is not aware of the problems inherent in herp sampling, the report may be an extremely biased and inaccurate account of the herpetofauna. An author aware of the problems can at least point them out and draw up a hypothetical species list based on known distributions and habitat requirements. It is important in such reports that information based on collected specimens, positive species identification without actual collection, and speculation on species presence and distribution be distinguished. Unfortunately, this is not always the case.

General search-and-seize and nighttime road cruises are cheap ways to obtain information. If relied on exclusively, with sufficient time and effort put into the search under the necessary variety of conditions, these two methods can provide a fairly complete species list. However, these methods are less valuable for comparisons of relative abundance and habitat use than drift fencing in the hands of nonspecialists. If your project warrants the expense or you (or your organization) will be involved in future surveys, then drift fence technology is a sound investment.

TABLE 6

Comparative herpetological survey data from the Cross Florida Barge Canal Study (modified from Campbell and Christman, 1982). Data collected at four different sites using three sampling methods.

Habitat	Quadrat		Time-constrained		Drift fence array	
	Animals	Species	Animals	Species	Animals	Species
Slash pipe	30	9	31	14	110	7
Xeric hammock	39	6	106	18	59	10
Oak sandhills	30	6	75	13	119	16
Pine scrub	23	8	48	10	148	19
TOTAL	122	15	260	24	436	29

FOUR

Methods for the Preparation of Herpetological Study Specimens

Scientific Collections

Many people oppose killing animals for any purpose, including for use as museum study specimens. They view scientific study collections as "dead zoos." Is it really necessary, they ask, to kill animals, put them in jars with foul smelling liquids or stuff them and file them away in drawers, and number each one? After all, the museum may have many examples of frogs or warblers. Animals can be observed in the field and species lists compiled without removing animals from their habitats and killing them. These are legitimate questions that deserve consideration. Unfortunately, these questions often go unaddressed by scientists or are dismissed with a patronizing statement concerning the needs of SCIENCE.

Well-maintained scientific collections are valuable resources for several reasons. These collections are the primary source of information concerning the diversity of life on earth. From such collections, organisms are named and classified, and evolutionary relationships are sorted out. Scientific studies using the collections form the basis of the field guides that the naturalist carries.

Teaching collections at museums, colleges, and universities acquaint many students—including tomorrow's botanists and zoologists— with the diversity of flora and fauna.

A scientific collection is an invaluable tool for conservation. The collection is a documented record of past occurrences that can be used to map species distributions, show changes in species status, and indicate alterations in the environment. For example, the herpetological collection at the Bell Museum of Natural History provided the majority of species distribution data used to assemble the Department of Natural Resources (DNR) status report on Minnesota's amphibians and reptiles. (Fig. 22)

My defense of scientific collections does not mean that herps should be collected in large numbers at every opportunity. If Minnesota museums contained excellent samples of the Minnesota herpetofauna from diverse areas, the need for more specimens would be slight. The herpetofauna of many areas of the state is not well documented, however, and even short-term collecting can greatly increase the species list for a particular site. For example, during his five-lined skink study in western Minnesota, Lang (1982) incidentally documented 15 new county records in a four-county area. The Minnesota Herpetological Society, with financial support from the Nongame Program of the Minnesota DNR, has initiated a series of collecting trips to herpetologically neglected areas of Minnesota. These expeditions are making important contributions to our knowledge of the Minnesota herpetofauna.

If you are going to collect herpetological information, the value of your labor will be greatly increased if you make a representative collection of the species encountered, preserve it, and deposit it in an appropriate institution where it will be properly curated and cataloged. Such a reference collection, referred to as a voucher collection, allows your identification of specimens to be verified. It's a way to leave a permanent contribution to Minnesota herpetology.

A voucher collection need not be large; a few examples of each species encountered is sufficient. Include juveniles and adults, if possible, and any unusual individuals. A

Figure 22. The herpetological collection room at the Bell Museum of Natural History in Minneapolis. This collection is the primary source of information on the herpetofauna of Minnesota. Photo by John Slivon.

large collection is necessary only when a species-specific research project requires a large sample size (e.g., a study of geographic variation in the morphology of wood frogs).

Where should specimens be deposited? The Bell Museum of Natural History has the most extensive herp collection in the state and contains most of the specimens cited in Breckenridge's (1970) treatment of the Minnesota herpetofauna. Voucher collections can also be donated to other state institutions with trained personnel who maintain scientific and teaching collections, such as the Science Museum of Minnesota and the biology departments at private and state colleges. Ideally, one institution in the state, active in research on the Minnesota herpetofauna, would be the repository for collections from all over the state. Unfortunately, no institution in the state has an active research program on the distribution and abundance of the Minnesota herpetofauna.

If you have just completed a herp survey and want to make a voucher collection, or if you wish to prepare a teaching collection for a school or nature center, you may want to learn how to preserve your specimens. If you have collected voucher specimens and have accurate locality data but neither the resources nor the inclination to preserve your collection, call the Nongame Program at the Minnesota Department of Natural Resources or the Bell Museum at the University of Minnesota for arrangements to maintain your collection. (See Appendix I for addresses and phone numbers.)

The preservation of herp specimens depends on a fluid technology. Specimens are treated with chemical solutions that preserve the body tissues and, unless the specimen is very large, the whole organism is usually kept. Thus, each specimen is a treasure-trove of data, capable of providing information on diet, reproduction, and internal and external morphology.

There are six important steps in the preservation of herps for scientific collections:
1) Collect field data
2) Kill the specimens
3) Label the specimens
4) Fix the specimens (the actual preservation process)
5) Store the specimens
6) Ship the specimens

The information on preservation methods presented here comes from two main references (Hall, 1962; Pisani, 1973). Pisani's guide is available through the Society for the Study of Amphibians and Reptiles (see Appendix III).

Field Data

A specimen without accurate locality data is like a bicycle without wheels; you have something in your hands, but you can't really do very much with it. The more information you have on each specimen, the more valuable the specimen is. Field notes (or copies) should be donated to the receiving institution along with the specimens.

Pisani (1973) lists the following information that should accompany each specimen collected:

Vital:
1) Locality
2) Date
3) Name(s) of collector(s)

Helpful:

4) Time of collection
5) Temperature and weather notes
6) Additional observations
    - -other species collected and/or observed
    - -notes on species abundance
    - -notes on color and unusual morphology
    - -notes on microhabitat
    - -notes on behavior
7) Field number tag

If you do not have topographic or county maps, the locality should be recorded as specifically as possible with respect to major roads, incorporated towns, and natural landmarks using standard state maps. A metropolitan locality can be described using city and street names.

United States Geological Survey (USGS) 7.5′ topographic maps and county maps will allow you to describe collecting areas with great precision by indicating the range, township, and section of your collecting site (Fig. 23). Township, range, and section boundaries appear as a grid system overlay on the map. Townships are numbered south to north and ranges are numbered east to west. (i.e., T101N, R40W = township 101 north, range 40 west). A township is made up of 36 one-square-mile sections. Each section can be split further into quarters or halves, which can in turn be split. The DNR prefers localities to be specified as finely as possible, at least to the quarter-quarter section (see Fig. 23 for an example; see Appendix I for information on where to obtain topographic maps). The goal of locality data is to allow someone else, using your information, to relocate your collection site (Moriarty, 1985).

If you are collecting at several localities in a day, mark the bags or containers from these sites to avoid later confusion. Do not mix catches from different localities and

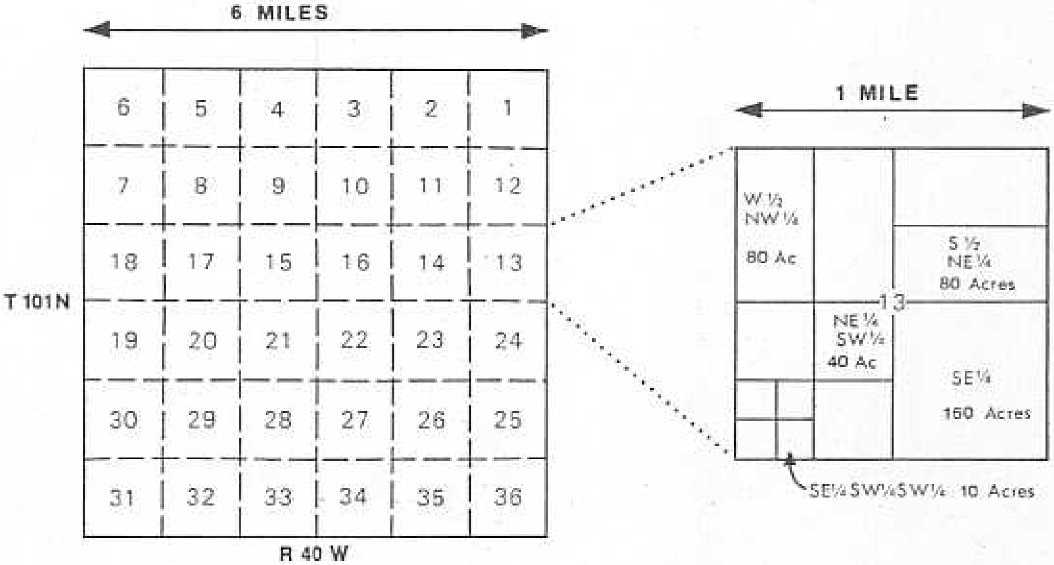

Figure 23. How to indicate specific areas within a section of a township. United States Geological Survey topographic maps are overlayed with a grid of township and range lines. The larger square is one township showing the 36 one-square-mile sections. Section 13 of the township is blown up and different ways of dividing it into quarters and halves are indicated. Identification to the quarter/quarter section is advised (modified from Moriarty, 1985).

expect to remember later what came from where. Also do not mix prey with its predators (e.g., frogs with a garter snake).

Dates can be a source of trouble. Write out the month or put the month in roman numerals to avoid confusion between day and month. Field tags will be discussed shortly.

Killing

The ideal killing agent should work quickly and leave the animal in a limp, relaxed condition. If the animal reacts vigorously to the killing agent, the result will be contorted specimens that make study difficult.

Reptiles

Nembutal (aqueous sodium pentobarbital) is the best killing agent for reptiles. Injection in or near the heart kills quickly and leaves muscles relaxed. Unfortunately, *Nembutal* is a controlled substance and available only to appropriately licensed individuals or institutions. If you do have access to this drug, it should be diluted before use for herps under five pounds at one part *Nembutal* to five parts water (to ten parts water for very small herps). Use the *Nembutal* which comes as a clear thin liquid, rather than the dark brown, syrupy elixir form.

Pisani (1973) notes that chloroform is a good killing agent for turtles. Keep the turtle in a closed container with a chloroform-soaked rag for 15-30 minutes; remove promptly upon death to avoid stiffening. Although suitable for turtles, chloroform will cause severe contortion in other reptiles. Pisani suggests that trichlorethylene (cleaning fluid) can be used full strength in a manner similar to chloroform with most reptiles without the contortion.

Amphibians

The best killing agent for amphibians is *Chlorotone* (hydrous chlorobutanol), which can be obtained from chemical supply companies by recognized institutions. If available,

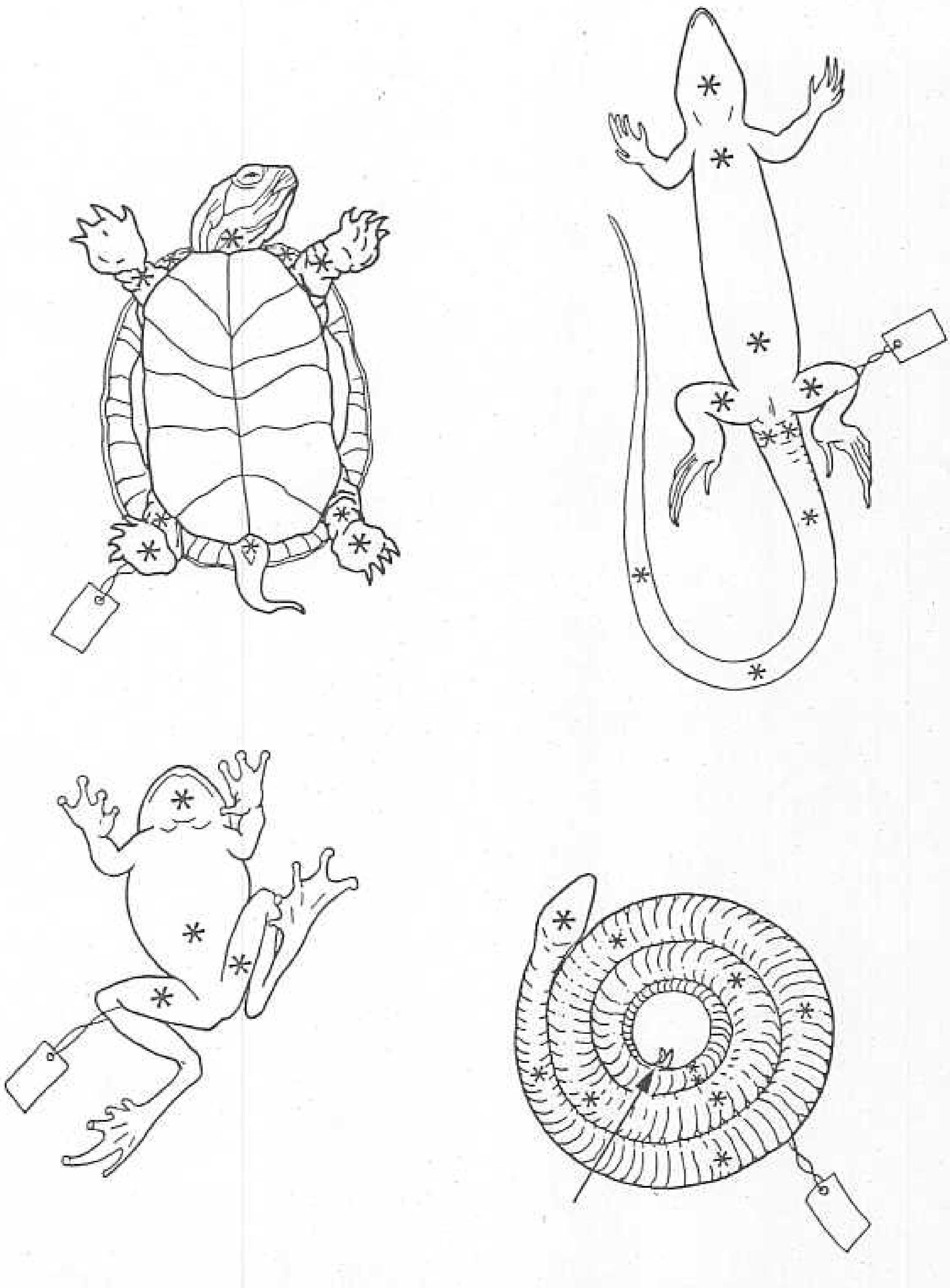

Figure 24. Suggested injection points and desired hardening positions for frogs, turtles, lizards, and snakes. Treat salamanders like lizards. Proper placement of tags is shown. Injection points are on the ventral (belly) sides of the animals. Arrow indicates an everted hemipenis. Final positioning in the fixative should be with the dorsal (back) side up, to insure desired form (modified from Cogger, 1975).

dissolve a teaspoon of *Chlorotone* in a quart of water. Amphibians placed in the water will succumb in a few minutes and be left in a relaxed condition.

Temperature extremes can kill herps quickly. One way is to place them in sacks and immerse them in 40° to 50° C water. They will usually not be too badly contorted if removed promptly upon death. Herps in sacks placed in a freezer will also quickly succumb, though often with contortion. Pisani also notes that injection of preservative into the heart followed by immersion into alcohol (15-25% for amphibians and 50-60% for reptiles) is an effective method of killing but usually results in severe contortion. In spite of contortion, freezing is probably the best killing method for the nonspecialist without access to the preferred drugs.

Labeling

Label the specimen after killing, but before fixing. Professional herpetologists usually give each specimen a numbered field tag, which insures that the specimen will not be confused with others. The field number is different from the collection catalog number that is eventually assigned to the specimen.

Individual tagging is worthwhile on extended collecting trips or if you accumulate many specimens, but may not be necessary if you are collecting locally and will be delivering your preserved specimens to an institution for curation and storage. In that case, keep specimens from different sites clearly marked and separated. A different container for each locality is the best idea. Label every container clearly and fully. Group tagging is not recommended museum procedure but will save the nonspecialist some work.

There are two options if you plan to label your specimens individually:

1) Field-number tag. In the field, label the specimen with a small tag with only a number and perhaps initials on it (e.g., DRK 36). Collection data must be entered in a field notebook under the tag number.

2) Locality-and-field-number tag. Label the specimen with a larger tag on which the locality, date, and collector(s) are written. A field number is usually put on the tag as well which refers to an entry in the field notebook where more information can be obtained. Separate field number and locality tags might also be used.

The tag must be made of a material that will not fall apart in the preservative. Tags are available from biological supply houses, but you can make your own less expensively using the highest quality and weight of 100% cotton and linen fiber paper (e.g., Dennison Paper Co. "Resistall Index Bristol," 100% rag, 110 lb. wt.). Some collectors use plastic tags imprinted by plastic label-maker "guns."

Ink that will not run in water, the fixative, or the storage fluid must be used. "Pelikan" brand drawing ink is a good choice (several other brands of "waterproof" ink will run).

Tags should be fastened to the animal securely. Pisani (1973) suggests using square knots. For most lizards, frogs, toads, and turtles the tag can be tied below the knee; for very small lizards, frogs, toads, and salamanders it can go around the waist. Tags should be tied around snakes at the thickest part of the body (Fig. 24).

Fixing

Fixing refers to the process of preserving the animal by using chemicals that penetrate the tissues of the organism and prevent postmortem decomposition. The goal is to preserve the original appearance of the specimen—its gross external morphology, cellular structure, and color. No fixative does all of these things equally well.

Formalin has proven to be the most effective all-around fixative for herps. It is not a controlled substance and can be obtained commercially from drugstores and chemical supply houses. Commercial *Formalin* is actually formaldehyde gas dissolved in water in a 37-40% solution. The percentage will be indicated on the label. To use this solution as a fixative, mix one part 37-40% commercial *Formalin* (straight from the bottle) to nine parts water. Ordinary tap water can be used as the diluting agent. Do not buy *Formalin* that has a white precipitate in the bottom of the bottle; it is an old solution.

Formalin is no fun to work with. It is poisonous, has a very irritating odor, can cause skin rashes, and has been implicated as a carcinogen. Use only in a well-ventiliated room and wear rubber gloves. (See Appendix III for information on the Consumer Product Safety Commission's report on the risks associated with exposure to specimens preserved in formaldehyde.)

An alternative to *Formalin* is ethyl alcohol (the drinking kind). However, this substance is very expensive (due to federal taxes) and can be difficult to obtain. As a fixative it should be used full strength for reptiles and at 75% strength for amphibians. Isopropyl alcohol (rubbing alcohol) can be used in an emergency, but is not as good a preservative. The rubbing alcohol bought in stores is usually 70% isopropyl alcohol and should be used full strength as a fixative.

If you have a dead animal and want to preserve it, but do not have access to proper fixatives, freeze it. Herpetologists in desperate situations (e.g., a valuable dead specimen, no fixatives, the middle of the Sonoran desert) have used liquor, brine, and shaving lotion as emergency fixatives.

The fixative must be introduced into the body cavity to insure complete preservation. There are two main methods: 1) injection by syringe, or 2) slitting open the body.

If you have access to syringes, injection is the cleanest technique. It is convenient to have a 10 and 35 cc syringe and several sizes of needles (18 to 26 gauge). The goal of injection is to retain the original form of the animal. Avoid bloating the specimen by injecting too much fluid. If syringes are not available, slit open specimens with a sharp scalpel, razor, or scissors. Slits must be deep enough to allow the preservative to enter the body cavity. All slits should be pulled open to facilitate entry of the fixative.

Frogs, toads, salamanders

Inject fixative into the body cavity (Fig. 24). Inject the head and limbs of large specimens. If you are not injecting, a single belly slit is sufficient for most specimens. No injection or slitting is necessary for small specimens.

Amphibian larvae and eggs

Tadpoles, salamander larvae, and amphibian eggs can be put live into 10% *Formalin*. This will kill the animals and preserve them with little distortion. Change the *Formalin* after 24 hours. Very large tiger salamander larvae can be treated as adult salamanders.

Lizards

Inject fixative into the body cavity (Fig. 24), limbs, and the head region on larger specimens. Injection does not work well with the tail (except at the base). Instead make a series of slits on the underside of the tail from the cloaca to the tail tip. Use sharp instruments; tails are easily broken off with too much abuse. If syringes are not available, slit open the belly of the lizard on one side of the midline of the body.

It is standard practice to evert one of the hemipenes of male lizards and snakes during fixation, to permit easy sexing of specimens (male lizards and snakes have two penises kept in pouches at the base of the tail). Inject fixative into one side of the base of the tail and simultaneously apply pressure with the thumb just behind the cloaca. The injection and finger pressure will usually pop out one of the hemipenes. Tie and knot a loop of thread around the base of the hemipenis to insure that it stays extended (Fig. 24).

Make a series of injections in the belly from the head to the base of the tail. An inch or two apart is sufficient on larger snakes (Fig. 24). Remember: fill the body cavity with sufficient fluid without bloating the specimen. The tail should be slit and one hemipenis everted on males as described above. If syringes are not available, make a series of slits along the length of the belly.

Turtles

A proper specimen will have its head extended, which can be accomplished by hooking the upper jaw of the dead turtle on a branch or pole and letting it hang for a while. Inject turtles in the neck, limbs, and tail (Fig. 24). Inject into the body cavity where the limbs join the body. If syringes are not available, make deep slits in these areas. Injection of sufficient fixative into the body cavity will "pop out" retracted limbs and the head. For large turtles, cut off the head and limbs, remove the internal organs for preservation. Clean and dry the shell.

Positioning

Immediately after injection or slitting, the specimen should be placed in a position that facilitates later examination. The fixative hardens the specimen into the desired position; afterward the animal feels like a semi-rigid rubber model of the original. Positioning is not possible with specimens contorted during the killing process.

To position and harden specimens you need shallow trays with covers, white paper towels, and fixative. The plastic trays used for refrigerator storage are excellent for this purpose (and cheap). Cover the bottom of the tray with paper towels (do not use colored towels because the specimens can be stained) and moisten with a fixative. Position the specimens as desired, cover them with another layer of paper towels, and pour in sufficient fixative to completely cover the specimens.

Standard positions for fixing are shown in figure 24. Animals should be placed belly down with limbs and digits spread out. Very small snakes can be laid out straight; larger snakes can be looped into a coil. Very large snakes (e.g., 1.5 m gopher snake) are conveniently coiled by looping them into jars filled with fixative. Long lizard tails can be bent around the side of the animal to save space.

Storage

Although Formalin is good for the initial fixing and hardening of the specimens, its nasty properties make it less than ideal for long-term storage of most specimens. If possible, specimens should be stored in either ethyl or isopropyl alcohol. Formalin should be used as a storage fluid for egg masses and amphibian larvae.

Both types of alcohol are usually sold at 95% concentration (except isopropyl purchased as rubbing alcohol, which is at 70% concentration) and must be diluted with water (distilled is better than tap) for use as storage fluids. A range of dilution strengths are seen in the herpetological literature: 50-75% alcohol for reptiles and 40-60% alcohol for amphibians. I prefer dilutions in the upper end of these ranges. Remember to adjust your dilution based on the initial alcohol concentration (e.g., 1 liter of 75% storage solution made with 95% isopropyl alcohol will require 788 ml of alcohol and 212 ml of water).

Pisani (1973) recommends keeping the specimens in the fixative for at least 24 hours. If the specimens are to be transferred to alcohol for storage, they should be soaked in water for 24-48 hours to prevent dehydration.

Glass jars with spring tops and rubber gaskets (used for canning) make excellent containers. Regular screw-top glass jars are also good, but fluid loss through evaporation will be greater. Evaporation can be retarded by covering the jar opening with a square of heavy-duty plastic before screwing on the top. Plastic containers, though useful in the field, conceal the specimen in storage. Containers must be watched for drops in fluid levels over time. Fill jars to the brim initially. Do not expose specimens to full sunlight for long periods.

Proper curation of study specimens involves attention to many details including labeling of specimen jars, cataloging of specimens, and other tasks that go beyond the scope of this booklet (Slevin, 1927, and Anderson, 1960 provide a full discussion of curating a herpetological collection).

Shipping

If you have amassed a preserved herp collection, you should donate it to an appropriate public institution; private collections are of little scientific value. If personal delivery is difficult, preserved material can be easily shipped by mail.

To prepare animals for shipping, wrap specimens in cheesecloth or white paper towels. An easy method of packing is to lay out a length of wrapping material, space out specimens along the strip, and then roll the wrapper up. Soak these specimen bundles in storage fluid and put them into heavy-duty waterproof plastic bags. Avoid packing specimens too tightly. Specimens packed in this manner will remain in good condition for weeks.

If your specimens are not individually tagged, make up your shipping bundles by locality and include an alcohol- and water-proof card with locality, collector, and date with each bundle. It is very important that the receiver of your collection be sent unambiguous, accurate information on the specimens. Close the bag with a heavy duty rubber band or by knotting it shut. Place this sealed bag into another plastic bag for extra protection against leakage, and seal the second bag.

Paint cans, heavy duty cardboard boxes, or wood boxes can be used for shipping. Pack the specimen bundles like any fragile commodity going through the postal system—prepare for the worst. Surround the specimen bags with plenty of buffering material (e.g., plastic packing chips, rags, etc.).

Place three address labels with the specimens: put one in the box or can with the specimens; tape a second label to the outside of the shipping can or box; fasten a third label on the outer paper wrapper (if used).

Under separate cover, send a letter explaining the circumstances surrounding the collection and a copy of your field notes. Keep a copy of this information for yourself in case the letter should be lost.

Postscript

This book is an assemblage of techniques from the literature filtered through my own experience of successes and failures. Every herpetologist has little "tricks of the trade" that make life easier. As you pursue your herpetological interests, you will undoubtedly develop various shortcuts, devise instruments for various chores, and discover new, useful products for herpetological field work. *Herpetological Review* (see Appendix III) publishes brief notes on new techniques. If you happen to devise a "better mousetrap," consider sharing it with the scientific community via *Herpetological Review*.

LITERATURE CITED

Anderson, R.M. 1960. *Methods of Collecting and Preserving Vertebrate Animals.* Bull. No. 69, Biology series No. 18. Natl. Museum of Canada, Ottawa. 164 pp.

Breckenridge, W.J. 1970. *Reptiles and Amphibians of Minnesota.* Minneapolis: University of Minnesota Press (3rd printing).

Campbell, H.W. and S.P. Christman. 1982. Field techniques for herpetofaunal community analysis. Pp. 193-200 in N.J. Scott, Jr. (ed.), *Herpetological Communities.* Wildlife Res. Report 13, Fish and Wildlife Service, U.S. Dept. of Interior.

Caldwell, R. 1975. Observations on the winter activity of the red-backed salamander, *Plethodon cinereus*, in Indiana. Herpetologica 31(1):21-22.

Cogger, H. 1975. *Reptiles and Amphibians of Australia.* Sydney, Australia: Reed.

Collins, J.T. 1974. *Amphibians and Reptiles in Kansas.* Publ. Educ. Ser. Univ. Kansas, No. 1. 283 pp.

Conant, R. 1975. *A Field Guide to the Reptiles and Amphibians of Eastern and Central North America.* Boston: Houghton-Mifflin Co.

Czajka, A.F. and M.A. Nickerson. 1974. *State Regulations for Collecting Reptiles and Amphibians.* Milwaukee Public Museum, Special Publications in Biology and Geology, No. 1.

Duellman, W.E. 1962. Directions for preserving amphibians and reptiles. Pp. 37-40 in E.R. Hall (ed.), *Collecting and Preparing Study Specimens of Vertebrates.* Misc. Publ. Univ. Kansas Mus. Nat. Hist. No. 30:1-46.

Ferner, J.W. 1979. *A Review of Marking Techniques for Amphibians and Reptiles.* Herp. Circul. No. 9, Society for the Study of Amphibians and Reptiles.

Gibbons, J.W. 1983. *Their Blood Runs Cold: Adventures with Amphibians and Reptiles.* University, AL: The University of Alabama Press.

Gibbons, J.W. and R.D. Semlitsch. 1982. Terrestrial drift fences with pitfall traps: an effective technique for quantitative sampling of animal populations. Brimleyana No. 7:1-16.

Goin, C.J., O.B. Goin, and G.R. Zug. 1978. *Introduction to Herpetology.* 3rd ed. San Francisco: W.H. Freeman.

Gregory, P.T. 1982. Reptilian hibernation. Pp. 53-154 in Gans, C. and F.H. Pough eds., *Biology of the Reptilia.* Vol. 13.

Herreid, C.F. and S. Kinney. 1967. Temperature and development of the wood frog, *Rana sylvatica*, in Alaska. Ecology 48(4):579-590.

Hine, R., B.L. Les, and B.F. Hellmich. 1981. *Leopard frog populations and mortality in Wisconsin, 1974-1976.* Tech. Bull. No. 122, Wisc. Dept. Natl. Res., Madison, WI.

Hine, R. 1982. Creatures of the night. Wisc. Acad. Rev. 29(1):21-23.

Hodge, R.P. 1976. *Amphibians and Reptiles in Alaska, the Yukon and Northwest Territories.* Anchorage: Alaska Northwest Publishing Co. 89 pp.

Karns, D.R. 1979. The relationship of amphibians and reptiles to peatland habitats in Minnesota. Minn. Dept. Nat. Res., Minerals Div., St. Paul, MN.

Lang, J.W. 1971. Overwintering of Three Species of Snakes in Northwestern Minnesota. M.S. Thesis, Univ. of North Dakota, Grand Forks.

________ 1982. Distribution and abundance of the five-lined skink (*Eumeces fasciatus*) in Minnesota. Final Report: Nongame Program, Minn. Dept. Nat. Res.

________ 1983. Blue Devils in Rock Valley: Our search for the five-lined skink. The Minn. Vol. July-Aug. 1983:40-46.

________ 1984. The Reptiles and Amphibians of Minnesota: Distribution Maps, Habitat Preferences, and Selected References. Final Report: Nongame Program, Minn. Dept. Nat. Res.

Martof, B.S., W.M. Palmer, and J.R. Bailey. 1980. *Amphibians and Reptiles of the Carolinas and Virginia.* Chapel Hill, NC: The University of North Carolina Press.

McCoy, C.J. 1982. *Amphibians and Reptiles in Pennsylvania: Checklist, Bibliography, and Atlas of Distribution.* Spec. Publ. Carnegie Mus. Nat. Hist. No. 6, 91 pp.

McKinnell, R.G., E. Gorham, F. Martin, and J. Schaad. 1979. A major reduction in the prevalence of the Lucke renal adenocarcinoma associated with greatly diminished frog populations. J. Natl. Cancer Inst. 63:821-824.

Minton, S.A., Jr. 1969. *Venemous Reptiles*. New York: Scribners.

Moriarty, J. 1985. The use of the U.S. General Land Survey (Township/Range) System. Minn. Herp. Soc. Newsletter 5(2):4.

Nickerson, M.A. and C.D. Mays. 1972. *The Hellbenders*. Publ. in Bio. and Geo. No. 1. Milwaukee Public Museum.

Pisani, G.R. 1973. *A Guide to Preservation Techniques for Amphibians and Reptiles*. Herp. Circul. No. 1. 22 pp. Society for the Study of Amphibians and Reptiles.

Plummer, M.V. 1979. Collecting and marking. Pp. 45-60 in Harless, M. and H. Morlock (eds.), *Turtles: Perspectives and Research*. New York: Wiley-Interscience.

Pough, F.H. 1983. Amphibians and reptiles as low-energy systems. Pp. 141-188 in Asprey, W.P. and S.I. Lustick (eds.), *Behavioral Energetics*. Columbus: Ohio State University Press.

Schmid, W.D. 1965. Some aspects of the water economies of nine species of amphibians. Ecology 46(3):261-269.

________ 1982. Survival of frogs in low temperature. Science 215: 697-698.

Semlitsch, R.D. 1985. Analysis of climatic factors influencing migrations of the salamander *Ambystoma talpoideum*. Copeia 1985 (2):477-489.

Shields, M.A. 1985. Selective use of pitfall traps by southern leopard frogs. Herp Review 16(1):14.

Slevin, J.R. 1927. The making of a scientific collection of reptiles and amphibians. Proc. Calif. Acad. Sci., 16(9):231-259.

Stenhouse, S.L. 1985. Migratory orientation and homing in *Ambystoma maculatum* and *Ambystoma opacum*. Copeia 1985(3):631-637.

Tester, J.R. and W.J. Breckenridge. 1964. Winter behavior patterns of the Manitoba toad, *Bufo hemiophrys*, in northwestern Minnesota. Annal. Acad. Scient. Fenn., Ser. A., IV Biol. 71/31:423-431.

Vogt, R.C. 1981. *Natural History of Amphibians and Reptiles of Wisconsin*. Milwaukee, WI: Milwaukee Public Museum. 205 pp.

Vogt, R.C. and R.L. Hine. 1982. Evaluation of techniques for assessment of amphibian and reptile populations in Wisconsin. Pp. 201-217 in Scott, N.J., Jr. (ed.), *Herpetological Communities*. Wildlife Res. Report 13, Fish and Wildlife Service, U.S. Dept. of the Interior.

Vogt, R.C. and J.J. Bull. 1982. Temperature controlled sex-determination in turtles: Ecological and behavioral aspects. Herpetologica 38(1): 156-164.

Wheeler, C. 1979. The ups and downs of frog picking. *Minnesota Daily*, Weds. 25 April, 1979:11-13.

Wheeler, G.C. and J. Wheeler. 1966. *The Amphibians and Reptiles of North Dakota*. Grand Forks, ND: The University of North Dakota Press, 104 pp.

APPENDIX I

Herpetological Resources

Equipment

Most of the equipment described in this booklet can be obtained from local hardware, department, and sporting goods stores. Listed below are several mail-order companies useful in finding various items for field research. If nothing else, their catalogs are fun to browse through. *Herpetological Review*, published by the Society for the Study of Amphibians and Reptiles (see Appendix III), carries advertisements of companies carrying herpetological supplies.

Forestry Suppliers, Inc.
205 West Rankin Street
PO Box 8397
Jackson, MS 39204-9987
(800) 647-5368
(Forestry Suppliers carries pesola scales for weighing herps.)

Carolina Biological Supply Company
Burlington, NC 27215
(800) 334-5551
(Carolina offers a wide range of equipment and educational materials in biology.)

Nylon Net Company
PO Box 592
Memphis, TN 38101
(800) 238-7529
(Everything you ever dreamed about in terms of nets.)

Miller and Weber, Inc.
Precision Thermometers
1637 George Street
Ridgewood, Queens, NY 11385-5342
(718) 821-7110
(Makers of the "Cloacal Quick-Reading Rat-Reptile Thermometer"-$15)

National Tag Company
815 South Brown School Road
Vandalia, OH 45377
(513) 898-1334
(Suppliers of high quality identification tags; smallest order accepted is 2000 tags; cost, $100.)

Furmont Reptile Equipment
Fuhrman Diversified
905H South 8th Street
La Porte, TX 77571
(713) 474-4832
(Suppliers of professional herpetology equipment; their catalog includes snake sticks, reptile sex probes, reptile bags, etc.)

Herp Care

As mentioned earlier, many pet stores are not knowledgeable concerning the care of amphibians and reptiles. Listed below are two pet stores with considerable herp experience. The Minnesota Herpetological Society is also an excellent source of information concerning herp care.

Pet Cetera
6519 Nicollet Avenue
Richfield, MN
(612) 861-8868

Twin Cities Reptiles
540 Winnekta Ave. No.
Golden Valley, MN 55427
(612) 593-0298

Minnesota Herpetological Society
(see below)

Maps

County maps may be purchased at many bookstores in the Twin Cities area.

U.S.G.S. 7.5' topographic maps are for sale at the Minnesota Geological Survey, some outdoor stores (e.g., Eastern Mountain Sports) or the Map Store (120 S. 6th St., Minneapolis, MN 339-4117).

Institutions and People

It can be difficult to obtain information on amphibians and reptiles in Minnesota. Listed below are places and people you can contact that may be able to answer your questions. I especially draw your attention to the Minnesota Herpetological Society. This is a young, strong organization dedicated to the care, conservation, and study of amphibians and reptiles in Minnesota. It holds monthly meetings at the University of Minnesota with speakers, live herps, and a variety of other events. As noted above, this organization is an especially good source of information on care of captive herps. The Bell Museum and Science Museum can usually point you in the right direction if you have herp questions.

Minnesota Herpetological Society
Bell Museum of Natural History
10 Church Street SE
University of Minnesota
Minneapolis, MN 55455

Bell Museum of Natural History
10 Church St. SE
University of Minnesota
Minneapolis, MN 55455
(612) 624-4112

Science Museum of Minnesota
30 E. 10th St.
St. Paul, MN 55101
(612) 221-9488

Minnesota Department of Natural Resources
Nongame Division
Section of Wildlife
Box 7
500 Lafayette Ave.
St. Paul, MN 55146
(612) 297-2276

There are few professional herpetologists in Minnesota. Listed below are individuals at various institutions in Minnesota whose research interests have included amphibians and reptiles. Vogt (1982) provides a similar list for Wisconsin.

University of Minnesota-Twin Cities

Dr. Walter Breckenridge (Professor Emeritus and Director Emeritus, Bell Museum). Now retired, Breckenridge made major contributions to our knowledge of the Minnesota herpetofauna (e.g., Breckenridge, 1970).

Dr. Robert McKinnell (Department of Genetics and Cell Biology). A developmental biologist and oncologist interested in developmental biology of amphibians, cloning of frogs, cancer in frogs; investigated recent collapse of leopard frogs in Minnesota.

Dr. Philip Regal (Curator of Herpetology, Bell Museum, Department of Ecology and Behavioral Biology). An evolutionary ecologist, Regal's interests include evolution, and physiological and behavioral ecology of amphibians and reptiles.

Dr. William D. Schmid (Department of Ecology and Behavioral Biology). A physiologist who studies physiological ecology of amphibians, particularly tolerance of amphibians to hydration and low temperature.

Dr. John Tester (Department of Ecology and Behavioral Biology). Vertebrate ecologist, investigated behavior and ecology of toads in western Minnesota.

Dr. James C. Underhill (Department of Ecology and Behavioral Biology). Ichthyologist, collected herps in many parts of the state, interested in stripe polymorphism in wood frogs.

Science Museum of St. Paul

Dr. Bruce Erickson (Curator of Paleontology). Paleoherpetology, especially of crocodilians.

University of Minnesota-Morris

Dr. Dennis Hoppe (Division of Science and Mathematics). Amphibian ecology and behavior.

Minnesota Zoo

Brint Spencer (Senior Keeper of Reptiles).

Nongame Program, Minnesota Department of Natural Resources

Lee Pfannmuller (Staff Zoologist). Closely involved with herpetological research projects in Minnesota.

Carrol Henderson (Chief of the Nongame Program).

Other

Dr. Jeff Lang (Dept. of Biology, University of North Dakota, Grand Forks). Evolutionary ecology, behavior, and physiology of crocodilians. Has extensive experience with Minnesota herps, investigated ant mounds used as herp hibernacula, ecology of the endangered five-lined skink, recent distribution maps of Minnesota herps.

James E. Gerholdt (PO Box 86, Webster, MN 55088). Knowledgeable amateur herpetologist. Gerholdt offers tailor-made herpetological presentations for schools and other groups ("The Remarkable Reptiles"; (507) 652-2996).

Minnesota Herpetological Society. Several individuals in the Society are particularly knowledgeable about various aspects of herpetology and can be contacted through the society.

Delvin Jones — founder of the Society, excellent general resource person.

John Moriarty — experienced field herpetologist.

Barney Oldfield — experienced field herpetologist.

Bruce Delles — owner "Twin Cities Reptiles," knowledgeable about herp care and husbandry.

Bruce Cutler — good all-around naturalist.

APPENDIX II
Status of Amphibians and Reptiles in Minnesota

Legal Status
by Lee Pfannmuller
Staff Zoologist
Minnesota Nongame Program

All reptiles and amphibians that are officially classified as state endangered or threatened in Minnesota are legally protected. At present, this includes the five-lined skink (state endangered), wood turtle (state threatened) and Blanding's turtle (state threatened). Permits issued by the Department of Natural Resources are required to legally take, possess, import, transport, purchase, sell or dispose of these species. Regulations for issuing these permis are governed by the conditions set forth in Minnesota Statute 97.488 Commissioner's Order 2204. General conditions outlined in the order include permits for scientific and educational purposes and rehabilitation. Permits will also be issued for specimens legally acquired before the effective date of the Commissioner's Order (June 18, 1985) or before the species were listed as endangered or threatened (March 1984). Proof of having acquired the specimens before this date must first be submitted. Permits are not required for taking or possessing those reptiles and amphibians that are officially classified as state special concern, except as noted below. All other lizards, snakes, salamanders, and toads not officially listed in Minnesota are totally unprotected; at present, no regulations restrict their acquisition or possession. Numerous laws and orders, summarized below, govern Minnesota's frogs and turtles.

Turtles

Noncommercial. Any person permitted by law to take fish by angling may take, possess, buy, sell, and transport turtles. Turtles may not be taken by the use of explosives, drugs, poisons, lime or other deleterious substances or by the use of nets, other than landing nets or traps.

The possession limit for snapping turtles is ten, and the dorsal surface of the carapace must be ten inches or more across at its most narrow point.

Commercial. A $50 commercial turtle license is necessary to take, transport, purchase, and possess for sale unprocessed turtles within the state, without limit. Turtle traps, turtle hooks, and commercial fishing nets may be used by holders of commercial turtle licenses and are the only gear to be used.

Minnesota laws (1983, 1984) pertaining to turtles are found in: 1) Minnesota Statute 97.48, Subdivisions 4, 17 and 21; 2) Minnesota Statute 97.49, Subdivision 4; 3) Minnesota Statute 97.46, Subdivision 5, Paragraph 6; and 4) Minnesota Statute 101.45 Commissioner's Order 2131, 2137, 2059.

Frogs

Any person permitted by law to take fish by angling may take or possess frogs for bait purposes only. Frogs may not be taken for bait if they exceed six inches from the tip of their nose to the tip of their hind legs when the hind legs are fully extended. Bait frogs can be possessed, bought, sold, and transported in any numbers. Frogs may not be taken from April 1 to May 15.

No more than 150 frogs over six inches in length may be possessed in or transported through the state except by common carrier.

The taking, possessing, purchasing, transporting, or selling of frogs for purposes other than as bait within the state is prohibited. Scientific or special permits may be issued to educational and scientific institutions within Minnesota.

State laws pertaining to frogs are contained in Chapter 97, Section 97.48, Subdivision 21, Chapter 101, Section 101.44 and Section 101.441, of the Minnesota Statutes and in Commissioner's Order 1381 and 1912.

Amphibian and Reptiles Classified as Endangered, Threatened or Special Concern

ENDANGERED

Eumeces fasciatus (Five-lined Skink)

THREATENED

Clemmys insculpta (Wood Turtle)
Emydoidea blandingi (Blanding's Turtle)

SPECIAL CONCERN

Chelydra serpentina (Snapping Turtle)
Coluber constrictor (Racer or Blue Racer)
Crotalus horridus (Timber Rattlesnake)
Elaphe obsoleta (Rat Snake or Black Rat Snake)
Elaphe vulpina (Fox Snake)
Heterodon nasicus (Western Hognose Snake)
Heterodon platyrhinos (Eastern Hognose Snake)
Lampropeltis triangulum (Milk Snake)
Pituophis melanoleucus (Gopher Snake or Bull Snake)
Sistrurus catenatus (Massasauga)
Tropidoclonion lineatum (Lined Snake)
Acris crepitans (Northern Cricket Frog or Blanchard's Cricket Frog)
Rana catesbeiana (Bullfrog)
Rana palustris (Pickerel Frog)

ENDANGERED:

A species threatened with extinction throughout all or a significant portion of its range.

A species threatened with extinction within Minnesota and dependent on a scarce, sensitive and/or exploited habitat in Minnesota and neighboring states.

THREATENED:

A species likely to become endangered (based on the criteria listed for the endangered category) within the foreseeable future.

SPECIAL CONCERN:

A species, that although not endangered or threatened, is extremely uncommon in Minnesota, or has unique or highly specific habitat requirements and deserves careful monitoring of its status.

A species on the periphery of its range which is not listed as threatened or endangered.

A species which was once threatened or endangered but now has increasing or protected, stable populations.

A species whose breeding biology is affected by human activities.

Minnesota's Herpetofauna
by Jeff Lang
Chairman, Amphibian and Reptile Group, Endangered Species Technical Advisory Group

Priority species

The lists below were formulated by the Amphibian and Reptile Group of the Endangered Species Technical Advisory Committee to assist in establishing priorities for future studies. Conservation of the state's herpetofauna requires different management strategies for the various species. In some instances, habitat protection is desirable; for other species, direct protection by prohibiting collecting is sufficient. For a particular species, additional information on distribution or abundance is often a prerequisite for effective management or conservation. These lists should be used to establish the appropriate approach or combination of approaches for a particular species.

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1. *Habitat:* Each of these species appears to be tightly linked to or dependent upon a particular habitat(s). Protection, preservation, and management of habitat is considered critical to the species' survival.

   Wood Turtle (*Clemmys insculpta*)
   Blanding's Turtle (*Emydoidea blandingi*)
   Five-lined Skink (*Eumeces fasciatus*)
   Western Hognose Snake (*Heterodon nasicus*)
   Eastern Hognose Snake (*Heterodon platyrhinos*)
   Rat Snake (*Elaphe obsoleta*)
   Massasauga (*Sistrurus catenatus*)
   Lined Snake (*Tropidoclonion lineatum*)
   Northern Cricket Frog (*Acris crepitans*)
   Pickerel Frog (*Rana palustris*)

2. *Direct Protection:* Some species are currently collected/harvested/killed by humans. Regulation or prohibition of such activities is conssidered essential to the continued survival of these species. We recommend that the Department of Natural Resources:
   1) regulate the harvest of snapping turtles (*Chelydra serpentina*) and spiny softshell turtles (*Trionyx spiniferus*); prohibit commercial trade in all other species of turtle found in Minnesota;
   2) abolish all bounties or similar incentives for killing or destroying any snakes, particularly rattlesnakes;
   3) prohibit or regulate commercial trade in all species of snakes, especially those reaching adult lengths of 50 cm or more;
   4) assess the impact of using frogs as a commercial bait; and
   5) protect all species of amphibians and reptiles, with exemption for private collectors of nonlisted species and regulation of certain other collecting activities.

3. *Distribution:* Distributions of these species must be documented further before effective conservation may be undertaken; present data are inadequate. Regional surveys of the local herpetofauna would be desirable in the following areas: southeast Minnesota (DNR region 5), southwest Minnesota (region 4S), and east central Minnesota (region 3E).

   Wood Turtle (*Clemmys insculpta*)
   Smooth Softshell (*Trionyx muticus*)
   Five-lined Skink (*Eumeces fasciatus*)

Ringneck Snake (*Diadophis punctactus*)
Rat Snake (*Elaphe obsoleta*)
Massasauga (*Sistrurus catenatus*)
Lined Snake (*Tropidoclonion lineatum*)
Eastern Newt (*Notophthalmus viridescens*)
Redback Salamander (*Plethodon cinereus*)
Cope's Gray Treefrog (*Hyla chrysoscelis*)
Northern Cricket Frog (*Acris crepitans*)

4. *Abundance:* Estimates of the abundance and densities of certain species, including some common ones that occupy a range of habitats, are required to establish the necessary baseline data to assess the effects of various management strategies and environmental changes and alterations. Some of these species have recently undergone marked and largely inexplicable fluctuations in numbers in some habitats in Minnesota and nearby states (e.g., Hine et al., 1981; McKinnell et al., 1979). Understanding the population dynamics of these species is considered a necessary first step in any conservation program.
Snapping Turtle (*Chelydra serpentina*)
Painted Turtle (*Chrysemys picta*)
Redbelly Snake (*Storeria occipitomaculata*)
Plains Garter Snake (*Thamnophis radix*)
Common Garter Snake (*Thamnophis sirtalis*)
Tiger Salamander (*Ambystoma tigrinum*)
Northern Cricket Frog (*Acris crepitans*)
Pickerel Frog (*Rana palustris*)
Mink Frog (*Rana septentrionalis*)
Northern Leopard Frog (*Rana pipiens*)
Wood Frog (*Rana sylvatica*)

General Recommendations

The Amphibian and Reptile Group of the Endangered Species Technical Advisory Committee has made recommendations in four major areas: preservation of habitat, protection from collecting or harvest, research, and education.

1. *Protect, preserve, and manage critical habitat.* This approach appears to be the method of choice for the conservation of a number of amphibian and reptile species. It is relatively easy to define the preferred habitat(s) of a particular species, and protection, acquisition, easements, etc., are usually straightforward. Habitat preservation is synergistic because other species of plants and animals benefit, not just the target species.

   For many species, specific habitats may be characterized. In Minnesota, important aquatic habitats include rivers, streams and associated woodland (wood turtles, smooth softshells), springs and springfed streams (northern cricket frog and pickerel frog), and prairie sloughs and marshes (Blanding's turtles). Important terrestrial habitats include rock outcrops in the upper Minnesota River valley and southwestern corner of the state (five-lined skink, lined snake), bottomland and bluffland of the Mississippi River valley in the southeastern corner of the state (massasauga, rat snake), and oak barrens, sand plains, beach ridges and moraines (western and eastern hognose snakes). Although many of these habitats are small and getting smaller, *the survival of these species in Minnesota is directly dependent on preserving specific habitats.* For an example, see Lang (1982) or individual species' status sheets.

Undoubtedly, the dramatic loss of native woodland and prairie to agriculture has had detrimental effects on the distribution and abundance of herpetofauna in the state. In Iowa, it is estimated that two-thirds of the herpetofauna is endangered; if present trends continue, less than a third of the present species will remain in 50-100 years. In Minnesota, loss of native habitat is most severe in the southern half and western third of the state. In these areas, a number of species live in the wooded bottomlands of the Minnesota and Mississippi River valleys and numerous other valleys along their tributaries. *These woodland habitats are crucial to a number of species and should be protected from further destruction and degradation through agricultural, residential, or commercial development.* Hoppe (in letter, 1982) comments that redbelly snake populations in the Morris area have declined with the loss of woodland to residential development. Populations of other species (e.g., lined snakes and rat snakes) sensitive to the loss of woodland meadow edge have declined and will continue to decline as these habitats disappear; stable populations will remain only in areas resistant to cultivation, i.e., rocky steep terrain. In Minnesota, documentation of declining populations of amphibians and reptiles due to habitat loss is hindered by a paucity of baseline data for comparison with present populations.

2. *Protect herpetofauna by regulation of collecting or harvest.* Nearly all of the state's 48 species of amphibians and reptiles may now be collected or harvested in unlimited numbers without any regulations or prohibitions. Various restrictions are applicable only to one species of turtle (snapping turtle) and "frogs"; all other species are apparently *not* protected. The State claims ownership of wild animals and prohibits acquisition or destruction of them (Section 97, 43, p. 24, Minnesota Game and Fish Laws 1981-82); but it is not clear to what extent, if any, this provision provides legal protection for the state's herpetofauna. *We recommend that all species of amphibians and reptiles in Minnesota be protected; all harvesting should be either prohibited or regulated.* All such regulations should specify particular species and avoid generic terms such as "turtles" or "frogs" to reduce ambiguities. Some provision should be made for a limited number of each species (not listed as endangered or threatened) to be taken and possessed alive, but not bought or sold, by a resident of the state. Provisions should also be made for specimens acquired by captive propagation, that is, progeny originating from individuals bred in captivity. The restrictions in force in Missouri regarding overall protection with provisions for personal possession may be a useful guide in formulating a general protection for herpetofauna in Minnesota.

The major threat for many species, particularly the common and abundant forms, is an unregulated harvest of specimens for biological supply houses. In Wisconsin, where a number of supply houses are based, the harvest of herpetofauna for this purpose has been substantial (160,000 leopard frogs and 42,000 turtles/year; Vogt, 1981) and likely detrimental to populations of certain species (Hine, et al., 1981). In Manitoba, 60,000 garter snakes were collected during two weeks in September 1981. The estimated value of the harvest is about $25,000. Large numbers of frogs (limit: 50 tons) and lesser numbers of salamanders have also been harvested under regulations in recent years (Bob Grant, Manitoba Department of Natural Resources, personal communication).

There is reason to believe that certain species of amphibians and reptiles are harvested in Minnesota and transported to biological supply houses elsewhere, primarily in Wisconsin. For instance, a harvest of 6965 painted turtles was reported in Minnesota in 1978. This species is not desirable for food or the pet trade ($0.10/pound; average turtle = 1 pound); but the species is in demand, either dead or alive, through biological supply houses for educational or scientific use ($45/turtle). Some painted turtles from Minnesota, identified by their large size and shell configuration, have been sold by midwestern animal dealers (Michael Ewert, personal communication). In our judgement, certain species in Minnesota are susceptible to harvest and exploitation by biological supply houses based either within the state or in nearby states. These species include painted turtles, garter snakes, tiger salamanders, and leopard frogs. *We recommend prohibition of commercial collecting or harvest by individuals or companies until the impact of these activities is assessed.* Wildlife officials in other states or provinces should be consulted to assist in the evaluation of possible impacts and in the formulation of realistic regulations.

Currently, the commercial harvest of turtles in Minnesota is licensed but not regulated (with the exception of minimum size restrictions on snapping turtles) by the Fisheries Section, Division of Fish and Wildlife, DNR. *We recommend that the commercial harvest of turtles in Minnesota be limited to two species only (snapping turtles and spiny softshell turtles) and that the harvest of snapping turtles be regulated.* Recommendations on the regulations applicable to spiny softshell turtles have not been formulated pending specific information on the harvest of this species. *Furthermore, we suggest that turtles (and other reptiles and amphibians) be considered "wildlife" rather than "fish"; hence, all the herpetofauna of the state, including turtles and frogs, should be under the control of the Wildlife Section.*

The taking or harvest of frogs has also been licensed and regulated by the fisheries section: presently, frogs may be taken only by holders of fishing licenses for use as bait or by scientific or special permit for study, for angling purposes, frogs (less than 6", tip of nose to toes) may be possessed, bought, sold, and transported in any numbers. *We recommend that the impact on frog populations of using frogs for bait be assessed and, if warranted, that appropriate regulations be adopted to curtail or limit such activities.*

In our judgement, certain species may be vulnerable to overcollecting to supply specimens for the pet or skin trades. In particular, some turtles and most of the large snakes occurring in Minnesota are valued at retail prices exceeding $25 per individual. Some species are relatively abundant in the state and may be easily collected in large numbers by experienced collectors for sale to animal or skin dealers. Many species in Minnesota use communal hibernacula where individuals congregate in the spring and fall, and thus are particularly susceptible to collectors. *We recommend that collecting for sale to animal or skin dealers or pet stores (including interstate sales) be assessed and thereafter be regulated on a species-by-species basis.*

Bounties on snakes, particularly rattlesnakes, are still on the books and in effect in some Minnesota counties, notably Houston, Winona, Wabasha, and Fillmore. In 1982, Houston County paid more than $3,000 for rattlesnakes at $1/snake. *We recommend that all bounties on any species of the Minnesota herpetofauna be abolished.*

Recently, there has been considerable interest in the use of larval salamanders for bait. "Waterdogs" are available for sale and shipment anywhere in the U.S. and Canada except Saskatchewan; this bait appears to be larval salamanders (*Ambystoma tigrinum*) originating from stocks in the southern U.S. Once such exotic forms are introduced and established, they pose a threat to native forms. *We recommend that the importation of amphibians and reptiles—especially nonnative species—in large numbers for commercial purposes (resale) other than the pet trade be prohibited.*

3. *Expand research on Minnesota herpetofauna.* Several major difficulties were encountered in determining the status of amphibians and reptiles in the state. First, the basic reference on the state's herpetofauna (Breckenridge, 1970) was published almost forty years ago; and even though distribution maps were updated in 1958, individual species accounts have not been revised or rewritten. Consequently, although Breckenridge's book has been an essential, invaluable reference, it is now out of date and requires extensive supplementation from existing literature. Second, recent surveys and inventories have been regional in scope and conducted by diverse agencies usually under the supervision of a nonherpetologist. Furthermore, the results of these studies have been scattered in government reports and publications and are not easily consolidated and integrated on a statewide basis. *Clearly, an up-to-date account of the distribution and abundance and natural history of the herpetofauna in Minnesota is needed; we recommend that such a project be undertaken.* It would be an essential resource in conducting surveys and inventories, in making regional management decisions, in evaluating the effects of environmental changes, and in increasing public awareness and appreciation of amphibians and reptiles in Minnesota.

In the interim, we recommend that surveys be targeted to specific regions within the state where more information on the distribution and abundance of the herpetofauna is required. The occurrence of many species is not well-documented in numerous counties. For example, on a short-term project to study one species, Lang (1982) documented 15 new county records in a four-county area. Regional surveys would be particularly desirable in the following areas: southeast Minnesota (DNR region 5), southwest Minnesota (region 4S), and east central Minnesota (region 3E).

A clearinghouse for herpetological information would facilitate recordkeeping and greatly simplify mapping distributions, preparing accounts, etc. Furthermore, one location for handling specimens to be identified and storing new distribution records would be convenient. The most extensive collection of herpetofauna in the state is presently at the James Ford Bell Museum of Natural History (= Minnesota Museum of Natural History MMNH), University of Minnesota. This collection contains nearly all of the specimens cited in Breckenridge (1970), and it is curated by a professional herpetologist.

There is a need to compile existing information on the local herpetofauna from the various state parks, State Scientific and Natural Areas, nature preserves, private tracts, etc., and also to solicit sightings, records, and observations from field professionals. The Uncommon Wildlife Reports filed by Department of Natural Resources personnel were generally useful in supplementing distributions based on specimens. Much more information on amphibians and reptiles could be gathered if specific efforts were made to alert various field personnel to the need for additional specimens and observations. For example, a visit with Bob Chance at Blue Mounds State Park resulted in documentation of the occurrence of the lined snake in Minnesota; a specimen accidentally killed later in the summer was saved and sent to the Bell Museum. Many naturalist groups could also be encouraged to contribute to such efforts.

Education programs that focus on the identification, observation, and preservation of amphibians and reptiles would greatly enhance such efforts. In this regard, the slide set with audio tape on the frogs and toads of Minnesota that was prepared for the Nongame Program of the Minnesota DNR is an excellent start for such a program. *We recommend that the DNR try to facilitate exchanges of information on amphibians and reptiles in order to upgrade current data on the distributions and abundances of Minnesota herpetofauna.*

The importance of establishing and maintaining baseline ecological studies on common species in typical habitats is often overshadowed by an emphasis on rare and uncommon species with peripheral distributions. A number of species which are rare or occasional in occurrence in states to the south are abundant and, hence, more easily studied in Minnesota. Some examples are Blanding's turtle, painted turtles, redbelly snakes, plains and common garter snakes, tiger salamanders, and wood frogs. In north temperate areas in particular, the herpetofauna constitutes as much biomass as other vertebrate groups; energy flow through a few abundant species may constitute a significant fraction of the total energy flow through the system (20% for salamanders in a New Hampshire forest; Pough, 1983). Certain species may be important indicators of habitat quality, e.g., northern cricket frogs, pickerel frogs (Vogt, 1981). Assemblages of amphibians and reptiles, and in particular their interspecific interactions, are not well studied or well understood; abundant species inhabiting typical Minnesota habitats provide excellent research opportunities. Finally, understanding the population dynamics of the common Minnesota herpetofauna is a necessary first step in any conservation program. We need to know more about seasonal activitiy patterns, reproductive behaviors, densities, and abundances of these species; and for each species, how these and other parameters respond to specific environmental changes. *We recommend that ecological studies be conducted on abundant species (listed in Minnesota herpetofauna: priorities) in typical Minnesota habitats.* Such studies should be coordinated on a regional basis with other states and provinces.

4. *Establish public education programs.* The biological and economic importance of Minnesota's amphibians and reptiles is generally not acknowledged or appreciated. In fact, most residents are probably not able to identify more than a few of the 48 species in the state; even people living in rural areas often are not able to distinguish between major groups, e.g., lizards and salamanders. Clearly, there is a need to increase public awareness of and appreciation for Minnesota's herpetofauna. Information should be made available on how to identify the various species, salient features about their life histories, and the functional signficance and ecological roles of these forms in the diverse environments they inhabit. *We recommend that public education programs be developed in consultation with professional herpetologists and then be widely distributed throughout the state.*

   Programs such as the slide presentation on frogs and toads of Minnesota prepared for the Nongame Program are excellent for this purpose; additional programs should be developed, including presentations on amphibians and reptiles, information brochures, identification keys, and other educational tools. DNR personnel in the field, park naturalists, and others professionally engaged in interpretive programs within the state should receive training aimed at increasing public awareness of and appreciation for Minnesota's herpetofauna. The status determinations and recommendations of this committee (Amphibian and Reptile Group, Endangered Species Technical Advisory Committee) should be incorporated into these programs and publicized. In some states, there is an individual within the state conservation unit who initiates and coordinates programs and policies dealing with amphibians and reptiles (for example, State Herpetologist, Missouri Department of Conservation; Reptile and Amphibian Specialist, New York State Department of Environmental Conservation). *We recommend that such a position be established in Minnesota.*

APPENDIX III
Annotated Bibliography

Professional and amateur herpetologists collect books about herpetology with the same enthusiasm as they collect specimens. The following list of references is by no means comprehensive, but is intended as a starting point. A basic herpetological library for Minnesota should include: a field guide (I prefer Conant), *Natural History of Amphibians and Reptiles of Wisconsin* (Vogt, 1982), *Reptiles and Amphibians of Minnesota* (Breckenridge, 1970), a general reference on amphibian and reptile biology (see below), and this booklet.

Advanced General Texts

These books are the only college-level texts devoted to herpetology. General zoology and vertebrate comparative anatomy texts will also provide background information.

Duellman, W.E. and L. Trueb. 1985. *Amphibian Biology*. New York: McGraw-Hill.

Goin, C. J., O.B. Goin, and G.R. Zug. 1978. *Introduction to Herpetology*. 3rd ed. San Francisco: W.H. Freeman.

Porter, K.R. 1972. *Herpetology*. Philadelphia: W.B. Saunders.

General Books

These books vary in their level of presentation, but most are written for the general public and are profusely illustrated.

Bellairs, A. 1969. *The Life of Reptiles*. Vol. I & II. New York: Universe Books.

Carr, A. 1963. *The Amphibians*. New York: Life Nature Library, Time Inc.

Cochran, D.M. 1961. *Living Amphibians of the World*. New York: Doubleday.

Frazer, J.F.D. 1973. *Amphibians*. London: Wykeham.

Gans, C. 1975. *Reptiles of the World*. Bantam Nature Guide, Knowledge Through Color Series. New York: Bantam Books.

Klauber, L.M. 1972. *Rattlesnakes: Their Habits, Life Histories, and Influence on Mankind*. Vols. 1&2. Los Angeles: University of California Press.

Minton, S.A., Jr. and M.R. Minton. 1969. *Venemous Reptiles*. New York: Charles Scribner's Sons.

______________ 1973. *Giant Reptiles*. New York: Charles Scribner's Sons.

Neill, W.T. 1971. *Last of the Ruling Reptiles*. New York: Columbia University Press.

Oliver, J. 1955. *The Natural History of North American Amphibians and Reptiles*. Princeton, NJ: D. Van Nostrand Co.

Parker, H.W. and A.G.C. Grandison. 1977. *Snakes—A Natural History*. Ithaca, NY: Cornell University Press.

Pope, C.H. 1961. *The Giant Snakes*. London: Routledge and Kegan Paul.

________ 1974. *The Reptile World: A Natural History of the Snakes, Lizards, Turtles, and Crocodilians*. New York: Alfred A. Knopf.

Pritchard, P.C.H. 1967. *Living Turtles of the World*. Neptune City, NJ: TFH Publications.

Schmidt, K.P. and R.F. Inger. 1957. *Living Reptiles of the World*. New York: Doubleday.

Identification

All field guides have advantages and disadvantages. I like Conant's because of the detail of his distribution maps. The Golden Guides offer a wealth of well-illustrated background information. Browse through them and see what you like.

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

Conant, R. 1975. *A Field Guide to the Reptiles and Amphibians of Eastern and Central North America.* 2nd ed. Boston: Houghton Mifflin. Peterson Field Guide Series.

Smith, H.M. 1978. *A Guide to Field Identification—Amphibians of North America.* New York: Golden Press. 1978.

Smith, H.M. and E.D. Brodie, Jr. 1982. *A Guide to Field Identification—Reptiles of North America.* New York: Golden Press.

Stebbins, R.C. 1966. *A Field Guide to Western Reptiles and Amphibians.* Boston: Houghton Mifflin. Peterson Field Guide Series.

Here are two useful papers on amphibian embryonic development and tadpole identification.

Altig, R. 1970. A key to the tadpoles of the continental United States and Canada. Herpetologica 26(2):180-270.

Gosner, K.L. 1960. A simplified table for staging anuran embryos and larvae with notes on identification. Herpetologica 16:183-190.

Herpetological Memoirs

Several herpetologists have written books about their experiences. These books are fun to read and provide insight into the daily lives of working scientists. Gibbons' book contains much information on field techniques.

Gibbons, W. 1983. *Their Blood Runs Cold—Adventures with Amphibians and Reptiles.* Unversity, AL: The University of Alabama Press.

Taylor, E.H., A.B. Leonard, H.M. Smith, and G.R. Pisani. 1975. *Edward H. Taylor: Recollections of an Herpetologist.* Monograph of the Museum of Natural History, The University of Kansas, No. 4.

Twitty, V.C. 1966. *Of Scientists and Salamanders.* San Francisco: W.H. Freeman.

Regional Herpetology

Unfortunately, there is no recent account of the herpetofauna of Minnesota. Breckenridge is in its third printing but has not been revised, except for some bibliographic updating, since its original publication in 1944. For this reason, some of the taxonomy is out of date and can be confusing. It remains a valuable source of information on natural history. Herp distributional data for Minnesota has been recently updated by Lang and Moriarty. Vogt's book covers many of the species found in Minnesota and, in my opinion, is one of the most beautiful and useful regional herp accounts.

Breckenridge, W.J. 1970. *Reptiles and Amphibians of Minnesota.* Minneapolis: University of Minnesota Press (3rd printing).

Fishbeck, D.W. and J.C. Underhill. 1959. A Check List of the Amphibians and Reptiles of South Dakota. Proceedings of the South Dakota Academy of Sciences. 1959:107-113.

Henderson, C. 1979. A preliminary review of the occurrence, distribution, legal status, and utilization of reptiles and amphibians in Minnesota. Minnesota Department of Natural Resources.

Lang, J. 1984. The Reptiles and Amphibians of Minnesota: Distribution Maps, Habitat Preferences, Selected References. Unpublished final report to the Nongame Program, Minnesota Department of Natural Resources.

Moriarty, J. 1985. Distribution Maps for Reptiles and Amphibians of Minnesota. Minnesota Herpetological Society and Nongame Program, Minnesota Department of Natural Resources.

Preston, W.B. 1982. *Amphibians and Reptiles of Manitoba.* Winnipeg: Manitoba Museum of Man and Nature.

Vogt, R.C. 1981. *Natural History of Amphibians and Reptiles of Wisconsin.* Milwaukee: Milwaukee Public Museum.

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

Handbooks

These books are part of a larger series, *Handbooks of American Natural History.* Although the taxonomy is dated, they remain an invaluable source of basic natural history information.

Bishop, S.C. 1947. *Handbook of Salamanders.* Ithaca, NY: Comstock.

Carr, A. 1952. *Handbook of Turtles.* Ithaca, NY: Cornell University Press.

Smith, H.M. 1946. *Handbook of Lizards.* Ithaca, NY: Comstock.

Wright, A.H. and A.A. Wright. 1949. *Handbook of Frogs and Toads of the United States and Canada.* 3rd ed. Ithaca, NY: Comstock.

——————— 1957. *Handbook of the Snakes of the United States and Canada.* 3 vols. Ithaca, NY: Cornell University Press.

Collection and Preservation

Knudsen is an excellent all around guide for the collection and preservation of plants and animals. Pisani was the primary source of information for the chapter on preparation of specimens.

Anderson, R.M. 1960. *Methods of Collecting and Preserving Vertebrate Animals.* Bull. No. 69, Biology Series No. 18, National Museum Canada, Ottawa. 164 pp.

Consumer Product Safety Commission. 1981. Investigation of Exposure to Formaldehyde from Preserved Specimens.

For a copy, write to:
Freedom of Information Officer
Office of the Secretary
Consumer Product Safety Commission
Washington, DC 20207

Czajka, A.F. and M.A. Nickerson. 1974. *State Regulations for Collecting Reptiles and Amphibians.* Milwaukee Public Museum, Special Publications in Biology and Geology, No. 1.

Hall, E.R. (ed.). 1962. *Collecting and preparing study specimens of vertebrates.* Misc. Publ. No. 30, Univ. Kansas, Mus. Natl. Hist.

Knudsen, J.S. 1972. *Collecting and Preserving Animals and Plants.* New York, NY: Harper and Row.

Pisani, G.R. 1973. *A Guide to Preservation Techniques for Amphibians and Reptiles.* Herpetological Circular No. 1., Society for the Study of Amphibians and Reptiles. See SSAR below for ordering information.

Slevin, J.R. 1927. The making of a scientific collection of reptiles and amphibians. Proc. Calif. Acad. Sci., 16(9): 231-259.

Herps In Captivity

Frye, F.L. 1973. *Husbandry, Medicine, and Surgery in Captive Reptiles.* Bonner Springs, KS: VM Publishing Co.

Nace, G.W. et al. 1974. *Amphibians: Guidelines for the Breeding, Care, and Management of Laboratory Animals.* Washington D.C.: National Academy of Science. Available from: Printing and Publishing Office, National Academy of Sciences, 2101 Constitution Avenue NW, Washington 20418.

Murphy, J.B. and J.T. Collins (eds.). 1980. *Reproductive Biology and Diseases of Captive Reptiles.* Contributions to Herpetology No. 1, Society for the Study of Amphibians and Reptiles. See SSAR below for ordering information.

Ecology

This booklet covers a variety of techniques useful in general survey work. If you are interested in more detailed studies of the population and community ecology of amphibians and reptiles, or just want to broaden your ecological/evolutionary expertise, these general college-level texts will get you started. The best way to learn about the kinds of advanced research being done is to browse through the professional herp journals: *Copeia*, *Herpetologica*, and the *Journal of Herpetology.*

Krebs, C. J. 1985. *Ecology: The Experimental Analysis of Distribution and Abundance.* 3rd ed. New York: Harper and Row.

Scott, N.J., Jr. (ed.). 1982. *Herpetological Communities.* Wildlife Research Report 13; U.S. Dept. of the Interior, Fish and Wildlife Service.

Smith, R.L. 1980. *Ecology and Field Biology.* New York: Harper and Row.

Nature Photography

These two books come highly recommended by nature photographers. Angel's book is best for beginners. Blaker's book explains more advanced technical procedures.

Angel, H. 1980. *Nature Photography.* Gr. Britain: Fountain Press, Argus Books Limited.

Blaker, A. 1976. *Field Photography.* San Francisco: W.H. Freeman.

Frog and Toad Calls

Bogert C.M. *Sounds of North American Frogs: The Biological Significance of Voice in Frogs.* New York: Folkways Records. LP record; 92 calls of 50 species of frogs and toads accompanied by a profusely illustrated essay on the subject.

Voices of the Night. New edition. 1982. LP record. Recorded calls of 36 species of frogs and toads from eastern North America. Send $8.95 plus $2.00 shipping to Cornell University Laboratory of Ornithology, 157 Sapsucker Woods, Ithica, NY 14850.

Minnesota Frogs and Toads. A tape cassette and slide show unit (96 slides) showing the frogs and toads of Minnesota, their habitats, and distribution. The tape narration includes the vocalizations of the various species and background information on amphibian ecology. This is an informative introduction to the frogs and toads of Minnesota. Contact the Nongame Program of the Minnesota DNR for information on this set.

Professional Herpetological Organizations

There are three major herpetological societies in America. Each one publishes a scientific research journal that is available by subscription. The best way to obtain information on subscription rates and membership information and privileges is to write to the business offices of the societies. These can be obtained from recent issues of the journals (addresses change as elected officers of the societies change). The journals are available at the library of the Bell Museum of Natural History.

American Society of Ichthyologists and Herpetologists.
Publishes *Copeia* quarterly.

Herpetologists' League, Inc.
Publishes *Herpetologica* quarterly.

Society for the Study of Amphibians and Reptiles.
Publishes *Journal of Herpetology* quarterly.

In addition to its quarterly journal, the Society for the Study of Amphibians and Reptiles issues a number of other valuable publications on a less regular schedule:

Facsimile Reprints in Herpetology are photolithographic reproductions of out-of-print, but still useful herpetological books and papers.

Contributions to Herpetology is a series of book-length contributions which include proceedings of symposia and scientific monographs.

Herpetological Review contains information of general herpetological interest, such as news of institutions, upcoming programs and events, geographic distribution information, husbandry tips, life history notes, techniques, and herpetological product advertisements.

Herpetological Circulars publishes miscellaneous manuscripts which will be of value to the general herpetological community and segments of the public-at-large. *Guide to Preservation Techniques for Amphibians and Reptiles*, *Standard Names for North American Amphibians and Reptiles*, and *Outline of Suggested Treatments for Diseases of Captive Reptiles* are a few of the available titles.

Catalogue of American Amphibians and Reptiles is a series of over 300 accounts dealing with particular genera or species of herps. Accounts include a description, distribution information, and literature survey.

APPENDIX IV
Snakebite

Minnesota is a good place to live if you dread being bitten by a poisonous snake. We have only two poisonous species (one is uncommon), and they are found only in the southeastern corner of the state. It is a safe bet that the water moccasins, copperheads, and rattlers that your neighbor sees sunning on the porch of his lake cabin in northern Minnesota are really something else.

The timber rattler (*Crotalus horridus*) occurs in the Mississippi River valley in southeastern Minnesota. It does not appear to extend north of the confluence of the Mississippi and St. Croix Rivers in either Minnesota or Wisconsin. The species has been reported from southern Washington, Dakota, Goodhue, Wabasha, Olmsted, Winona, Filmore, and Houston counties (Lang, 1984).

Because the timber rattler can be locally abundant, you may find yourself in rattlesnake country even in Minnesota. Timber rattlers exhibit a conspicuous seasonal movement pattern. In the spring and fall, they can be found on bluffs, ledges, and rocky outcrops near overwintering dens. In the summer they move down from these sites to bottomlands, croplands, and forests.

The massasauga (*Sistrurus cantenatus*) is not common in Minnesota. It has been reported only from river bottomlands in Wabasha and Houston counties. Based on available evidence, Lang (1984) suggests that there are no breeding populations in Minnesota and the massasaugas reported are individuals brought by floods from Wisconsin.

What should you do if bitten by a poisonous snake in Minnesota? As noted earlier, if you are alert and cautious in poisonous snake country, the chances of being bit are very small. It is not, however, impossible; a young girl died in 1983 after a bite from a timber rattler in Wisconsin.

Both our poisonous snakes are pit vipers with haemotoxic venom—modified saliva that digests muscle tissue, destroys blood cells, and causes hemorrhaging and swelling. Rattlers use their venom to kill their prey, but a timber rattler is not trying to eat you. The snake is striking defensively. Various studies indicate that in a surprisingly high proportion of venomous snakebites—as much as one-half—little or no venom is injected (Minton, 1969).

If you are bitten, try to catch the snake or identify it positively. If that is not possible, the bite marks alone will identify the snake as poisonous; the fangs of our pit vipers leave conspicious puncture marks. A nonvenomous snake, with its small, sharp teeth, leaves a series of small pricks in the shape of a snake's mouth.

Snakebite treatment has gone through a series of fads over the years. In the western states in the late 1800's whiskey therapy was used. Dr. W.F. Beattie, writing in the New York Medical Journal in 1873, prescribed one-half pint of bourbon every five minutes until a quart was taken for snakebite victims (Minton, 1969). More recent recommendations have included cutting the bite area and sucking out venom, tying tight tourniquets to slow down absorption and spread of venom, and applying cold treatments. Unfortunately, these treatments may do more harm than good.

Today, doctors advise that you remain calm and go to the nearest hospital for treatment. A constricting band (not a tourniquet) should be placed above the bite. A constricting band is loose enough so that a finger can be inserted underneath it. Release the band for ninety seconds every ten minutes; this allows surface circulation. A tourniquet is tied much tighter and cuts off deep circulation in the appendage. Do not use a tourniquet.

Antivenin may be administered at the hospital to counteract the venom. Antivenin is usually produced by injecting modified snake venom into a horse (or a goat) and then collecting the antibodies produced. The problem with antivenin is that you may have an allergic reaction to it, which can cause more damage than the actual snakebite. Sensitivity tests are available to help the physician decide if the antivenin is safe to use. If you are administered antivenin to which you are allergic, drugs are available to counteract the allergic response.

With this basic information you should be able to discuss snakebite intelligently with the doctor, who, because venomous snakebite is rare in Minnesota, may know less about it than you. If he or she decides to cut-and-suck, ask why.

APPENDIX V

Sample Data Sheet

Species ______________________________

Collector ____________________ Date ____________

Locality ______________________________

____

Weather/Temperature ______________________________

Habitat ______________________________

Microhabitat ______________________________

Notes (sex, measurements, behavior, color, other species observed)

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INFORMATION FOR CONTRIBUTORS

Original papers concerning Natural History, especially of the Upper Midwest, will be considered for publication in the Occasional Papers of the Bell Museum of Natural History. Review articles will not be accepted unless specifically solicited, or unless review leads to new and important generalizations.

All manuscripts are to be in English and must be typed (double spaced throughout, including tables) and submitted in triplicate. Xerox copies are best as editing will be done on the copy. The title page should include the title and the author's name and address. An abstract should follow the title page and should not constitute more than three percent of the entire manuscript. Only three ranks of subtitles are allowed in the text.

References to literature in the text are by author, date, and specific pages cited. A Literature Cited with entries arranged alphabetically by last names of authors must include complete references, inclusive pages for journal articles, and total pages for books or other separate publications. Footnotes are not acceptable. Style and punctuation of citations is as follows:

Shaw, A.B. 1956. Notes on *Modocia* and Middle Cambrian trilobites from Wyoming. Jour. Paleontology, Vol. 30: 141-145.

Frey, D.G. 1965. Other invertebrates—an essay in biogeography. P. 613-631 *in* Wright, H.E., and D.G. Frey, *Editors*, The quarternary of the United States . . . . Princeton Univ. Press, Princeton, New Jersey, 922 p.

A complete set of illustrations shall accompany each copy of the manuscript. Illustrations must be neat and photographs of a high quality to insure the best possible reproduction. Each illustration should be labeled on the back with a figure number and the name and address of the author, and must be accompanied by a typewritten legend on a separate page.

Maps must conform to good cartographic principles. Avoid upside-down lettering on streams, label hydrographic features with right-slanted letters, and label man-made features with vertical letters.

Illustrations are classed as figures and plates. A figure is a black and white line illustration that can be printed within page size. Plates are (1) line drawings that must be printed larger than page size, (2) illustrations that require color, and (3) all photographs.

Fifty reprints will be furnished gratuitously to the author. In event of multiple authorship, the fifty will be divided equally among the authors. Additional reprints will be provided each author at cost.

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Minneapolis, Minnesota 55455

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No. 1. 1916 .... Out of Print
Roberts, T.S. The winter bird-life of Minnesota. Being an annotated list of birds that have been found within the state of Minnesota during the winter months. 20 p.

No. 2. 1926 .... Out of Print
Roberts, T.S. The winter bird-life of Minnesota. P. 5-10.
Roberts, T.S. The migration of Minnesota birds. P. 11-16
Roberts, T.S. March and April bird-lore in Minnesota. P. 17-22.
Roberts, T.S. May bird-lore in Minnesota. P. 23-28.
Four radio talks broadcasted in the University of Minnesota program over Station WCCO in Minneapolis, 1926.

No. 3. 1930 .... $.50
Roberts, T.S. Some changes in the distribution of certain Minnesota birds in the last fifty years. P. 9-14.
Kilgore, William. Breeding of the Connecticut Warbler *(Oporornis agilis)*, with special reference to Minnesota. P. 15-28.
Breckenridge, W.J. Breeding of Nelson's Sparrow *(Ammospiza nelsonii)* with special reference to Minnesota. P. 29-38.
Breckenridge, W.J. A hybrid Passerina *(Passerina cyanea + Passerina amoena)*. P. 39-40.

No. 4. 1950 .... Out of Print
Gunderson, H.L. A study of some small mammal populations at Cedar Creek Forest, Anoka County, Minnesota. 49 p.

No. 5. 1952 .... Out of Print
Olson, S.T., and W.H. Marshall. The Common Loon in Minnesota. 77 p.

No. 6. 1953 .... Out of Print
Gunderson, H.L., and J.R. Beer. The mammals of Minnesota. 190 p.

No. 7. 1957 .... Out of Print
Underhill, J.C. The distribution of Minnesota minnows and darters in relation to Pleistocene glaciation. 45 p.

No. 8. 1961 .... Out of Print
Tester, J.R., and W.H. Marshall. A study of certain plant and animal interrelations on a native prairie in Northwestern Minnesota. 51 p.

No. 9. 1963 .... $.75
Dickerman, R.W. The song sparrows of the Mexican Plateau. 79 p.

No. 10. 1971 .... $2.00
Phillips, G.L., and J.C. Underhill. Distribution and variation of the Catostomidae of Minnesota. 45 p.

No. 11. 1972 .... $1.40
Eddy, S., R.C. Tasker, and J.C. Underhill. Fishes of the Red River, Rainy River, and Lake of the Woods, Minnesota, with comments of the distribution of species in the Nelson River Drainage. 24 p.

No. 12. 1973 .... $1.50
Moore, J.W. A catalog of the flora of Cedar Creek Natural History Area, Anoka and Isanti counties, Minnesota. 28 p.

No. 13. 1974 .... $1.25
Birney, E.C., J.B. Bowles, R.M. Timm, and S.L. Williams. Mammalian distributional records in Yucatán and Quintana Roo, with comments on reproduction, structure, and status of peninsular populations. 25 p.

No. 14. 1975 .... $3.00
Timm, R.M. Distribution, natural history, and parasites of mammals of Cook County, Minnesota. 56 p.

No. 15. 1977 .... $1.50
Merrell, D.J. Life history of the leopard frog, *Rana pipiens*, in Minnesota. 23 p.

No. 16. 1980 .... $3.25
Weaver, M.G., and D.J. McLaughlin. Mushroom Flora of Minnesota, a contribution. 89 p.

No. 17. 1985 .... $3.00
Braun Hill, S. and D.H. Clayton. Wildlife after dark: A review of nocturnal observation techniques. 24 p.