APPENDIX H. TWRA Sampling Protocols for Select Faunal Groups

Sampling Protocols for Surveying the Shorebirds, Small Mammals and Bats, Reptiles, and Amphibians of Tennessee

Wildlife Diversity and Endangered Species Programs

2005

Shorebird Monitoring Protocol for the Interior Regions of Tennessee

Introduction

Widespread degradation and loss of habitat are causing rapid and profound changes to bird populations throughout North America. As a consequence, information on how bird populations are changing and becoming increasingly important to wildlife managers. Early detection of population changes is essential to a wildlife planning strategy. Measuring population size or change is also crucial for evaluating the effectiveness of population management programs.

There are 32 species of shorebirds that migrate through the state of Tennessee. Many species of shorebirds have been in decline for some time. In most cases, the causes of shorebird population declines are poorly known. Some possible causes are deterioration of nesting, migration, and wintering grounds, or just simple natural population fluctuations. Determining which of these actions, if any, are warranted will be possible only after implementing a shorebird monitoring plan.

Goals

I. Monitor long-term population trends for individual shorebird species.
II. Derive more precise population estimates of species population size.
III. Monitor regional use of habitat and population responses to habitat changes.
IV. Ensure that this information is incorporated into the overall Shorebird Monitoring and Management Plan.

Objectives

I. Design, test, and implement a long-term survey for tracking population trends for all shorebirds species of Tennessee.
II. Promote an ongoing research program for evaluation and improvement of population estimates and monitoring protocol.
III. Design a long-term program for monitoring regional patterns of habitat use as a tool to identify quality habitat and shifts in distribution patterns.
IV. Designate a center or location for maintaining shorebird population databases.
V. Establish a practical mechanism for assuring that the monitoring programs are designed to carry out sound evaluations of the effectiveness of habitat management programs.
VI. Improve National and International coordination and implementation of monitoring programs.

Shorebird Survey Design and Methodology

Design of population sampling and choice of counting methods require careful thought before beginning any monitoring program. The method requires an understanding of the spatial and temporal dynamics of the population, the advantages and disadvantages of counting techniques, and statistical issues relevant to one's ability to infer about population sizes or trends.

For non-breeding surveys, the annual summary statistic is the mean count per survey. For breeding surveys, the summary statistic is the mean number of birds per unit of effort (e.g., birds per route).

Historically, the TWRA has relied upon shorebird enthusiasts for information on shorebird occurrences and population estimates. The TWRA has also participated in the International Shorebird Survey (ISS). However, shorebird migration inventories and qualified abundance information is lacking.

The TWRA will monitor two areas per administrative unit for shorebird surveys during the months of July, August, and September. A minimum of three days will be surveyed at each site during each month. There will be a minimum of five-day intervals between surveys.

In general terms, first occurrence (I.E. first sightings), peak migration, and pass through dates will be noted.

Shorebird monitoring can be conducted in many ways. Occurrence and counts of shorebirds will be made similar to the ISS methodology. Counts conducted will be estimates of abundance.

The first method for estimating populations will be **total counts** of shorebirds using ground based counting methods. This method will be conducted in the following way:

1. Transects should be mapped out for each sampling location. Transect census points should be a minimum of 500 meters apart. Transects should be separated by at least one drainage system to minimize duplicate counts of individuals. Total counts are an estimation of all the shorebirds in the sampling locations (2 per administrative unit), so total counts should cover the entire sampling area if possible.

2. At each census point, a single observer records the total number of shorebirds present within a 10 minute period. Exact geographic locations should be recorded using GPS (if available). Habitat type and approximate size should also be recorded.

3. Species occurrence and species abundance estimates will also recorded during this allotted time.

The second method used will be **extrapolated counts**. This method will be conducted in the following way:
When the number of birds is greater than a few hundred birds, estimation procedures have to be used. The birds in a large flock may be estimated by counting a block of 50 or 100 birds and then estimating how many similar-sized groups make up the entire flock. Photographs can be used to estimate the average density of flocks and then measure the area covered by the flocks using terrestrial markers. Then the estimates may be multiplied area by density to estimate the total number of birds within the flock.

In a large mixed-species flock, observers estimate the proportion of the various species making up the flock and then the total number of birds in the flock is divided up into individual species counts according to these estimated proportions. In some cases, where the observers are unable to determine the proportions of each species, only the species composition of the flock is noted. The total count of these unallocated birds is then divided up into individual species counts assuming that the proportions of the unallocated birds is similar to the proportions of identified species in the rest of the site.

a) In small roosts and feeding flocks, the number of birds can be counted directly.

b) For small flocks of even density, the birds can be counted individually (1, 2, 3, 4, 5 etc.) to produce an accurate total. If a suitable landmark is present it can be used to help count the birds.

c) In unevenly distributed flocks with small groups of varying size, each group of birds should be rapidly counted and added together.

d) For larger numbers of birds in evenly distributed flocks the birds should be counted in multiples e.g. 2, 4, 6, 8 or 3, 6, 9, 12, etc. Again if landmarks are present they can be used to help divide the flocks in order to count them more accurately.

e) For densely packed flocks in flight or at a roost, the birds should be counted in estimated blocks. The size of the blocks used (10, 100, 1000 etc.) varies according to the size of the block. The largest flocks of 10,000 birds or more present the biggest counting problems with even the block method giving a rough estimate of numbers.

f) Flying flocks often bunch in the centre. In this case it is important that the blocks are closer together in the centre of the flock than towards the edges, but in practice this may be difficult to achieve.

The third and final method used will be **guesstimates**. This method is primarily when you don't have time to conduct a total count or extrapolated count method or when the birds fly away before the recorder has adequate time to conduct the proper counting method. This method will be conducted in the following way:

1. Use the same methods as above with two exceptions. First, the amount of time required for species occurrence and population estimates is not required and the guesstimate should be based on sound reasonable population estimates. Secondly, the species occurrence inventory may be made based on known characteristics of shorebirds and not on time consuming exact identification of these birds.

*Attached is the shorebird questionnaire (attachment 1) that is to be filled out at each new site location along with a survey data form (attachment 2) that is to be filled out each time a person goes to the field to monitor shorebirds.

*Attachment 1

Site Questionnaire

1. Is this site an upland area (field, prairie, etc)? Yes No

2. Is the site natural? or man made?

3. Are water levels managed? unmanaged? unknown?

4. Is this site a feeding area? resting area? or both?

5. Is the census area public? private? or both?

6. Is the land managed for wildlife? Yes No

7. What is the approximate size of the area in acres? _______

8. Are the major disturbances in this area related to natural factors? human activities?

9. How many times a day would you say the resting area for shorebirds is disturbed (approximate)? ________

10. What are the major causes of the disturbance? People?

Vehicles? Other animals? Boats? Other, what? ____________

11. What are the major shorebird habitats of your census area? (Circle One)

Permanent lake or pond	Temporary lake or pond
Sewage Treatment area	Fresh water marsh
Fresh water river	Agricultural field

Small Mammal Inventory Protocols
All personnel conducting small mammal surveys will be inoculated with the appropriate and available vaccines to prevent disease contraction.

Problem Two of the Wildlife Diversity chapter of TWRA's Strategic Plan identifies the lack of information concerning the status of nongame species populations, their ecological limiting factors and subsequent management implications. Strategy #1 calls for species inventories and/or assessments as well as habitat evaluations to determine community integrity, relative abundance of species, population densities and ecological requirements of nongame species. Very little information is available in regard to small mammals inhabiting TWRA managed lands. With these small mammal inventories TWRA will begin to collect data needed to determine the diversity, distribution and relative abundance of small mammals inhabiting its Wildlife Management Areas.

Bats
Bats will be surveyed from May through September to determine species occurrence and foraging site locations. Before working with bats, investigators should be able to properly identify bats to species. Investigators should also have basic knowledge of roosting habits, foraging strategies, and seasonal movements of bats. Furthermore, investigators should have knowledge of environmental effects on bats (i.e. temperature, humidity, light intensity, and topography). Sixteen species of bats may occur in Tennessee (Table 1). A complete list of materials needed to capture bats is provided (Table 2). Bats will be surveyed using the following procedures:

Roost Searches
Counting individuals in a roost is an effective way of collecting occurrence and abundance data. Roost searches will be carried out by individuals where bats are known to roost. This may include caves, trees, bridges, old home places, barns, chimneys, etc. If available, bat activity and species presence may be determined at each sampling station using the Anabat II ultrasonic detector attached to a laptop computer. There are three methods that may be used when conducting roost searches (direct roost counts, nightly dispersal counts, and nightly emergence counts)[1].

Direct roost counts
Direct roost counts may be conducted wherever bats roost. Depending on the species, roost counts may be conducted day or night. Many studies have been conducted on bridges as roosting sites for bats. Johnson et al. conducted night roost surveys on concrete and steel I-beam type bridges for gray bats and found that bridges suitable for gray bat night roosting have exposed porous concrete ceilings with small cracks and holes that provide places for them to roost and rest.[2] Studies have shown that bridge crevices have been used as day roosts by big brown bats, southeastern myotis, gray bats,

[1] Wilson, Don E., F. Russell Cole, James D. Nichols, Rasanayagam Rudran, and Mercedes Foster. 1996. Measuring and monitoring biological diversity, standard methods for mammals. Smithsonian Institution Press, Washington and London. Pp. 105-114.
[2] Johnson, B. Joshua, Michael A. Menzel, John W. Edwards and W. Mark Ford. 2002. Gray bat night-roosting under bridges. Journal of the Tennessee Academy of Science 77(4):91-93.

little brown bats, evening bats and Mexican free-tailed bats in the Southeastern United States. BHE Environmental Inc. describes suitable roost bridges as, “bridges that provide cavern-like characteristics such as darkness, flowing water and/or protection from disturbance.” During a summer survey conducted by Britzkey and Morgan[3], bridge surveys were conducted both above (examining expansion joints of bridges) and below the road level during the day. BHE Environmental Inc. also searches the drain holes of bridges. Cave walls, old home places, barns and even trees may be searched for the presence of roosting bats.

Nightly dispersal counts
Nightly dispersal counts are used to count bats as they disperse from their diurnal roost sites. Observers will be at the designated roost site one hour before nightfall and position themselves where the sky (or some other backdrop such as an open body of water) can be used to silhouette the bats. Light-gathering binoculars will aid in seeing bats as light levels decrease. Observers should count each bat only once, as some bats may circle the roost site before actually dispersing.

Nightly emergence counts
Nightly emergence counts can be the most accurate of all roost search methods. Counts are made as the bats depart their traditional day roosts. Similar to the nightly dispersal counts, nightly emergence counts should be conducted using the sky to silhouette the bats as well as using light-gathering binoculars. The total number of bats that reenter the roost should be subtracted from the total number of emerging bats to gain a more accurate estimate. Video and photographic equipment should be used when necessary to augment the visual observations. At roost sites where large aggregations of bats are known to be, accurate visual estimates may not be possible. In these cases, photographs taken at fixed intervals will be used. Nightly emergence counts will take place over three consecutive nights to account for emergence variability.

Mist Netting
Mist Nets
Mist netting will be conducted for three consecutive nights during each month for the months of May, June, July, August, and September. One mist-net night is defined as a single net being open for at least one hour. Five nets will be set each night mist-netting is attempted. This will result in a total of 75 mist-net trap nights per year.

<u>Procedure for Mist Netting Bats</u> [4]
Mist nets should <u>not</u> be used in areas where large numbers (thousands) of emerging bats are known to be.

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[3] Britzke, E. R. and K. L. Morgan. 2003. Summer survey of the Upper Cumberland Plateau, Tennessee. Submission to the Nature Conservancy of Tennessee. Pp. 25.
[4] Wilson, Don E., F. Russell Cole, James D. Nichols, Rasanayagam Rudran, and Mercedes Foster. 1996. Measuring and monitoring biological diversity, standard methods for mammals. Smithsonian Institution Press, Washington and London. Pp. 105-114.

All nets should be in place before sunset; however, nets should remain closed until just before dark to minimize the chance of catching birds going to roost.

Nets will be set at potential commuting, foraging, or drinking sites (across the middle of roads, streams, small pools or the lip of one edge of larger pools). Nets may also be set at the entrance of roost sites.

Poles will be anchored by driving a rebar into the ground and sliding the pole over the top of the rebar. In areas where rebar cannot be used, the poles will be anchored using rope. One person can set the nets, but it is much easier using two people. Nets will be carefully unfolded and the loops at the end of each trammel placed on the poles in sequential order.

Two-ply, braided nylon nets with 36 mm mesh will be used. Two-ply nets will be used, as they are less likely to be detected by bats. Mist nets are normally available in 3, 6, 9, 12, 42, and 60 m lengths. Three-meter nets work well at portal openings, while 12 m nets are better in large open areas. The 6 m nets are convenient and most often used when capturing bats. The 6 m nets can be set in more enclosed areas or in open areas by placing two 6 m nets end to end.

The net should be spread taunt enough so that a bag droop (about 1 inch) occurs in each panel. Nets will be checked throughout the evening to ensure they are taunt and adjustments made when necessary.

Nets should be placed within reasonable traveling distance to each other. This will reduce the overall time bats spend tangled in the nets, as it takes time for data collectors to travel between the nets. Nets will be checked every 15 minutes to reduce stress on the bats and the chances of holes being chewed in the nets.

During the time between net checks and data collection, all headlamps and flashlights should be off and talking kept to a minimum to avoid scaring off bats.

After a bat is caught, it should be removed as quickly as possible to avoid further entanglement or possible injury. After determining which side of the net the bat entered from, immobilize the bat with a gloved hand and carefully remove the net strands from the bat's head, body, and wings. Special care should be taken with wing membranes and bones, as they are very fragile. All bats should be immediately placed into pre-weighed cloth bags unless they are being processed right then.

Typically bats will be identified to species, sexed, weighed, aged, and forearm length recorded. Though bats are rarely recaptured, marking may be done to ensure all bats are new catches. When marking bats, "White-out" on the back of the head or a black permanent marker ("Sharpie") used on the wing membrane are acceptable. Both are nontoxic and will wear off in a couple of days. All bats should be worked as quickly as possible and released near the area they were captured.

Other standard bat measurements (Menzel, et al. 2002) used in identification include (see illustrations) – total length (TL), tail length (TV), foot length (HF), ear length (E), forearm length (FA), and tragus length (TR). Other data to record includes species, sex, age (young or adult), reproductive condition, body mass (grams), forearm length, time of capture, and evidence of recent feeding (females – determined visually or by abdominal palpation). Using the pre-weighed cloth bag, weigh bats to the nearest 0.1 grams by taking the total weight (bat and bag) and subtracting the weight of the bag. For species less than 5 grams weigh to the nearest 0.01 grams.

Nets will be closed at midnight. Most bat activity will have significantly decreased by this time.

Table 1. List and status of possible bat species in Tennessee (Harvey and Britzke 2002).

Common Name	**Genus**	**Species**	**Status***	**Data Code**
Little brown bat	*Myotis*	*lucifugus*	C	MYLU
Southeastern bat	*Myotis*	*austroriparius*	S	MYAU
Gray bat	*Myotis*	*grisescens*	E	MYGR
Northern long-eared bat	*Myotis*	*septentrionalis*	C	MYSE
Indiana bat	*Myotis*	*sodalist*	E	MYSO
Eastern small-footed bat	*Myotis*	*leibii*	S	MYLE
Eastern pipistrelle	*Pippistrellus*	*subflavus*	C	PISU
Big brown bat	*Eptesicus*	*fuscus*	C	EPFU
Rafinesque's big-eared bat	*Corynorhinus*	*rafinesquii*	S	CORA
Townsend's big-eared bat	*Corynorhinus*	*townsendii*	E	COTO
Eastern red bat	*Lasiurus*	*borealis*	C	LABO
Seminole bat	*Lasiurus*	*seminolus*	U	LASE
Hoary bat	*Lasiurus*	*cinereus*	C	LACI
Silver-haired bat	*Lasionycteris*	*noctivagans*	U	LANO
Evening bat	*Nycticeius*	*humeralis*	C	NYHU

Brazilian Free-tailed bat	*Tadarida*	*brasiliensis*	U	TABR

*** C- Common U- Uncommon S- Special concern E- Endangered**

Table 2. List of materials needed to catch bats.

List of Materials	
Mist nets	3-m, 6-m, and 12 m (or multiple 6 m)
Poles	Two 1.5-m (5-ft) segments joined by a sleeve will be needed at each end of the net. These should be light-weight.
Anchor cord	In areas where the ground is too rocky, poles may need to be tied instead
Stakes	Rebar [one for each pole, at least (76.2cm) 30 in long]
Mallet	For driving rebar into the ground
Caliper	Measuring (a metric ruler will work, but a caliper is better)
Scales	At least 30 gm (have an extra one for back up)
Cloth bags	For holding and weighing bats after removal from the net. Bags should be able to tie at the open end. Old sheets sewn into quart-size bags work well.
Knife	For cutting rope, etc…
Headlamp	For freeing both hands
Extra batteries	For headlamps/flashlights
Leather gloves	For handling the bats (lightweight gloves work best)
Watch	For noting the time of capture/release
Clipboard	For holding data forms
Data forms	For filling in data (weather-proof paper is recommended)
Pencils	To complete data forms (Do not use regular ink pens-they smear when wet)

White-out/Sharpie	If marking bats is a consideration
Camera/film	For voucher pictures
Waders/boots	Some areas will be wet and muddy
Two-way radios	For communicating with others in your group
Cell phone/radio	Especially if you are alone
First Aid kit	Be sure you have disinfectant in case you are bitten
Food/Water/Matches	In case you end up spending the night there (people have been known to get lost at night)
Folding Chair	To sit in while completing data forms

Small Mammals
Small mammal occurrence data and abundance data expressed in terms of catch per unit effort (individuals/trap night) will be determined with the use of live capture box traps placed along straight-line transects.

Commercial mouse and rat-sized live-traps [Small Sherman Aluminum Folding Live Traps (7.62x8.89x22.86cm) and Tomahawk Folding Wire-mesh Live Traps (15.24x15.24x48.26cm)] will be set three consecutive nights the months of October through March with at least one week between each sampling period. A transect of 100 meters (unless unique habitat prevents this transect length) per habitat type will be established.

Procedures:
Investigators should wear protective clothing, including long pants and long-sleeved shirt, socks and heavy shoes. Each investigator should carry a waterproof marker and data sheet. Wear leather gloves to handle small mammals.

Small Sherman live traps will be used along trap-lines.

Mark the beginning and end of transects with flagging tape.

On the first day of the sampling period arrive at transect in time to set all traps before dark. Leave traps along transect for the remainder of the sampling period.

Carrying the traps required for the transect in a sack or shoulder bag, walk the trap line, placing each trap as level as possible, with the mouth of the trap flush with the ground. The ground may be cleared and leveled by scraping the soil with the foot. Place two traps at every 10-15 meters along the transect line. Traps should be placed within 2 meters of the transect line. Place traps at cover features such as alongside or inside rotting logs, rock piles, burrows, etc. In hot seasons or climates, avoid placing traps in areas that will be exposed to direct sun. If this is impossible, traps may be covered with a board or with canvas cloth. If freezing temperatures are likely, add two cotton balls to each trap to provide bedding material during the night.

Mark each trap site with flagging tape to avoid loss of traps.

Check the trigger mechanism of each trap for proper adjustment and sensitivity as it is placed. This can be checked prior to going to the field to save time.

Bait each trap with a mixture of peanut butter and rolled oats.

Traps should be checked as early in the morning as possible, especially in hot weather and when traps are exposed to direct sun.

Check traps, if trap door is sprung, place a plastic baggie over one end of trap. Turn trap upside down, opening trapdoor and emptying specimen into baggie for identification and measurements. Close baggie and take necessary weights and measurements.

Make the following measurements on <u>voucher specimens</u> only and record on datasheet:
Weigh each mammal in grams.
Measure in millimeters tail length (base of tail to tip).
Measure total length (Head + Body + Tail).
Measure length of hind foot and ear length.
For all other specimens weigh, age (adult, sub-adult, juvenile) and sex (cannot sex shrews).

On first two nights, mark specimens with Non-toxic, White Correction Pen to note recaptures.

The number of individuals trapped per number of trap nights will be used as an index of abundance of small mammal species per transect. Richness or diversity will be determined by the number of different species captured per transect.

Tomahawk live-traps will be set opportunistically (not along transects) for the capture of wood rats. For opportunistic sampling, place 2-3 traps near brush piles, fallen logs, burrows, abandoned car bodies, or other items that provide shelter. When in or near buildings, place traps parallel to and against walls or other vertical surfaces, in cabinets, behind appliances and furniture, and on shelves above the floor, being especially attentive to areas where evidence of rodent activity is visible.

Before the next sampling period traps should be decontaminated with a 5% Lysol® (1 quart to about 4 ½ gallons of water in a 5 gallon plastic bucket).

Straight-line Drift Fences:
While utilizing this method of capture for reptiles and amphibians, it is probable that small mammals will also be taken. If so, take the same measurements and mark as above. This incidental data will be noted and used in determining the richness of the area. It will not be factored into the individuals/trap night abundance equations.

Artificial Cover Surveys:
Small mammals, primarily voles and shrews may be encountered while checking cover boards. If so, take the same measurements and mark as above. This incidental data will be noted and used in determining the richness of the area. It will not be factored into the individuals/trap night abundance equations.

Voucher Specimens:
Photographs and/or a preserved specimen documenting each species occurrence will be taken. New species occurrences (i.e. range extensions) will be documented by preserving a specimen a depositing the voucher in the mammalian collection at the University of Memphis collection. Collected specimens should be euthanized with appropriate chemicals with properties similar to chloroform. Investigators should not breathe chemical fumes or use any chemicals near an open flame.

Small Non-Volant Mammals of Tennessee

COMMON NAME	SCIENTIFIC NAME	STATUS	CODE
CHIPMUNK, EASTERN	(TAMIAS STRIATUS)	Common	TAST
COTTONTAIL, NEW ENGLAND	(SYLVILAGUS TRANSITIONALIS)	Rare	SYTR
LEMMING, SOUTHERN BOG	(SYNAPTOMYS COOPERI)	Common	SYCO
MINK	(MUSTELA VISON)	Common	MUVI
MOLE, EASTERN	(SCALOPUS AQUATICUS)	Common	SCAQ
MOLE, HAIRY-TAILED	(PARASCALOPS BREWERI)	Rare - D	PABR
MOLE, STAR-NOSED	(CONDYLURA CRISTATA)	Very Rare - D	COCR
MOUSE, COTTON	(PEROMYSCUS GOSSYPINUS)	Common	PEGO
MOUSE, DEER	(PEROMYSCUS MANICULATUS)	Common	PEMA
MOUSE, EASTERN HARVEST	(REITHRODONTOMYS HUMULIS)	Common	REHU
MOUSE, GOLDEN	(OCHROTOMYS NUTTALLI)	Common	OCNU
MOUSE, HOUSE	(MUS MUSCULUS)	Common	MUMU
MOUSE, MEADOW JUMPING	(ZAPUS HUDSONIUS)	Species of Concern - D	ZAHU
MOUSE, OLDFIELD	(PEROMYSCUS POLIONOTUS)	Common	PEPO
MOUSE, WHITE-FOOTED	(PEROMYSCUS LEUCOPUS)	Common	PELE
MOUSE, WOODLAND JUMPING	(NAPAEOZAPUS INSIGNIS)	Common	NAIN
RAT, BLACK	(RATTUS RATTUS)	Exotic	RARA
RAT, HISPID COTTON	(SIGMODON HISPIDUS)	Common	SIHI
RAT, MARSH RICE	(ORYZOMYS PALUSTRIS)	Common	ORPA
RAT, NORWAY	(RATTUS NORVEGICUS)	Exotic/Common	RANO
SHREW, LEAST	(CRYPTOTIS PARVA)	Common	CRPA
SHREW, LONG-TAILED	(SOREX DISPAR)	Rare - D	SODI
SHREW, MASKED	(SOREX CINEREUS)	Species of Concern - D	SOCI
SHREW, PYGMY	(SOREX HOYI WINNEMANA)	Very Rare	SOHO
SHREW, NORTHERN SHORT-TAILED	(BLARINA BREVICAUDA)	Common	BLBR
SHREW, SMOKY	(SOREX FUMEUS)	Species of Concern - D	SOFU
SHREW, SOUTHEASTERN	(SOREX LONGIROSTRIS)	Species of Concern - D	SOLO
SHREW, SOUTHEASTERN WATER	(SOREX PALUSTRIS)	Very Rare - D	SOPA
SQUIRREL, NORTHERN FLYING	(GLAUCOMYS SABRINUS)	Extremely Rare - E	GLSA
SQUIRREL, RED	(TAMIASCIURUS HUDSONICUS)	Common	TAHU

D = Deemed in Need of Management E = Endangered

Amphibian and Reptiles

Introduction

Tennessee has 66 species of amphibians and 55 species of reptiles. There are 21 species of frogs and 45 species of salamanders (Redmond and Scott 1996), 15 species of turtles, 9 species of lizards, and 31 species of snakes (TWRA Strategic Plan, 2000 – 2006). The Tennessee Wildlife Resources Agency is beginning an inventory to determine the herpetofauna on Wildlife Management Areas (WMA). A multiple gear approach will be used yielding data that is both qualitative (e.g. Species Richness)) and quantitative (i.e. catch per unit effort). After the first year of sampling, these protocols should be reviewed. Some methodology changes may be warranted. Scheduling should be evaluated as well. Not only does the timing of our sampling methods overlap, small mammals and shorebirds are also inventory targets. The gears used and frequency of sampling may need to be tailored for each area being inventoried.

Methods

Bringing together available resources is the first step when starting any project. Prior to beginning a field inventory, it is mandatory to collate existing information, which may have relevance to the current project. Information may come in various forms, maps, reports and articles, spoken word, and raw data. It may be that no herpetological survey has been conducted on a given management area but a WMA manager may know of occurrences of key species and the location of unique habitats.

Sufficient reconnaissance to the study areas should be conducted. The major habitat types on the area should be identified and all unique habitat features (Table 1) should be located and mapped (e.g. gps coordinates). These habitats should be described in detail sufficient as to allow the identifications of similar habitat at other sites or in other studies.

Table 1. Initial list of Habitat Descriptions

Aquatic Habitats	Terrestrial Habitats	Anthropogenic (Tiebout 2003)
Swamp	Forest	Barns
Marsh	Woodland	Roads
Oxbow	Savannah	Railroad Bed
Bottomland Hardwoods	Shrubland	
Seep	NWSG Field	
Pond	Hay Field	
Temporal Pond	Agricultural Field	
Mucky Area	Old Field	
River (TNC Size Class)	Rock Face	
Headwater (0.2 – 2m)	Cave	
Creek (2.2 – 20m)		
Small River (20.3 – 201m)		
Medium River (201.4 – 503m)		
Large River (>503m)		

Animal Measurements

All measurements will be made in metric units. Frogs and toads will be measured Snout to Vent Length (SVL), salamanders, lizards, and snakes will be measured SVL and total length (TL), carapace length will be measured on turtles. If possible, the sex of a specimen should be determined and recorded. Petranka (1998) has a section sex determination in salamanders. Snake handling protocols are described in *Standard for Components of British Columbia's Biodiversity No. 38*. Any mutations should be noted using *Field Guide to Malformations of Frogs and Toads*, by Carol Meteyer.

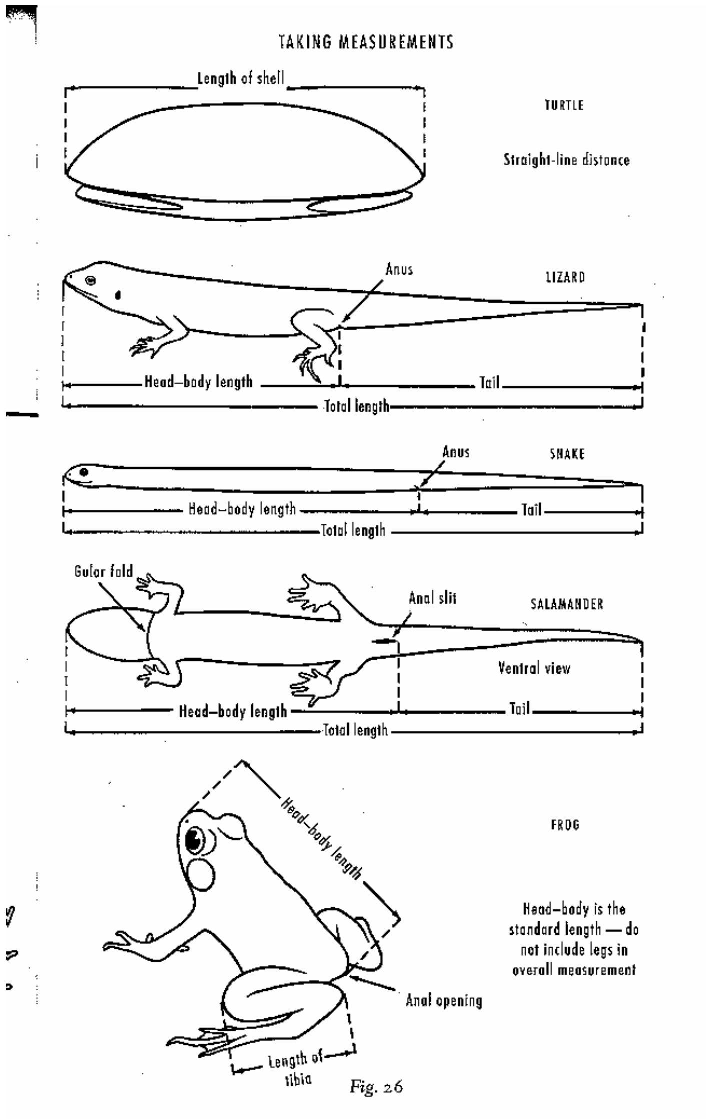

Figure 1. Illustration of reptile and amphibian measurements. From Conant and Collins (1998).

Amphibian Sampling
Active and Passive Sampling will be employed to inventory Amphibians. Active sampling methods include Call Surveys and Visual Encounter Surveys (VES). Passive sampling methods include coverboards and drift fences with pitfalls and or funnel traps.

Call Surveys
The TWRA participates in the North American Amphibian Monitoring Program (NAAMP). Tennessee's Amphibian Monitoring Program (TAMP) utilizes volunteers to conduct sampling routes. To compliment this effort and to generate data for TWRA lands specifically, sampling routes will be conducted on a minimum of 4 tracts of TWRA owned lands (1 TWRA per region). On a small or roadless WMA with limited wetland habitat, fixed stations may be established and the protocols associated with the tamp routes may be followed.

Routes will have 10 stops with a minimum of 0.5 miles between stops. Stops are ground-truthed as to suitable amphibian habitat within 200 meters of the stop. Four runs per year will be conducted as according to prescribed calling periods (Table 1). Run nights will be based on appropriate air temperature, sky code and Beaufort wind codes (Tables 3, 4 and 5).

Table 2. Start and end dates in which sampling runs can be conducted for amphibian call counts

	Run 1*	**Run 2***	**Run 3***	**Run 4***
Start Date	January 10	March 10	May 10	July 1
End Date	February 20	April 15	June 15	August 9

Table 3. Minimum air temperature needed to conduct amphibian call counts.

4 Run System	**Minimum Temperature**
Run 1	5.6° C (42° F)
Run 2	5.6° C (42° F)
Run 3	10° C (50° F)
Run 4	12.8° C (55° F)

Table 4. Sky codes and conditions needed to conduct amphibian call counts.

Sky Codes (note 3 and 6 are not valid code numbers)	
0	Few Clouds
1	Partly cloudy (scattered) or variable sky
2	Cloudy or overcast
4	Fog or smoke
5	Drizzle or light rain (not affecting hearing ability)
7	Snow
8	Showers (is affecting hearing ability). Do not conduct survey

Table 5. Beaufort Wind codes. Routes will not be conducted under codes 4 or 5.

0	Calm (<1 mph) smoke rises vertically
1	Light Air (1-3 mph) smoke drifts, weather vane inactive
2	Light Breeze (4-7 mph) leaves rustle, can feel wind on face
3	Gentle Breeze (8-12 mph) leaves and twigs move around, small flag extends
4*	Moderate Breeze (13-18 mph) moves thin branches, raises loose paper
5*	Fresh Breeze (19-24 mph) trees sway

Routes will be run 30 minutes after sunset. At each stop an “effective” listening period of 5 minutes will be made. If running a fixed station, the listening period will be 30 minutes. All identified species will be noted and abundance will be estimated by the criteria in Table 6.

Table 6. Criteria used to determine abundance of species heard during chorus call counts.

1	**Individuals can be counted; there is space between calls**
2	Call of individuals can be distinguished, but there is some overlapping calls
3	Full chorus, calls are constant, continuous and overlapping

Visual Encounter Survey (VES)
The VES has been used primarily to rapidly evaluate large forested areas, as well for targeting specific species. VES is one in which field personnel walk through an area or habitat for a prescribed period of time or distance systematically searching for animals. Catch per unit effort (CPUE) is expressed as animals observed per hour of searching or area covered. This technique is appropriate for species inventory and relative abundance studies.

Three basic sampling designs are used for the VES: randomized walk, quadrant, and transect (Heyer et al. 1994). We will use quadrant and transect methods. A series of transects or quadrants (1-5) will be established in major and/or unique habitat patches. Though transect distances will be dependent on topography and habitat patch, for consistency lengths will be based on 25 meter increments (i.e. 25, 50, 75, 100 m). Quadrants should be 10m X 10m or 25m X 25m. In a unique area such as a small high elevation wetland, one transect of approximately 25m or one 25m x 25m quadrant would be sufficient. In major habitat types, such as a 40 hectare tract of contiguous mixed hardwoods, up to 5 transects of varying lengths and elevations would be established. VES will be conducted in forested and field habitats. Transects can also be conducted along streams and around ponds (Dodd 2003).

Transects or areas may be established as fixed sample sites to be sampled monthly through one yearly cycle. This repeated sampling should reduce seasonal bias associated with observing some amphibians. If the area being inventoried is large with complex habitat it may be desirable to sample at different sites each month or alternate sites so that more area may be covered. Transects will be searched at an intermediate level of intensity (Heyer et al. 1994). This means in addition to counting already exposed animals, the observer turns over surface objects such as rocks and logs within 1m of the transect and counts the animals uncovered. The cover objects must be returned to their original positions to minimize habitat disturbance. This type of search generally yields higher return per unit time than a low-intensity search because many amphibians hide under cover objects when conditions are not suitable for surface activity.

The VES may be used at ponds and in streams. Ponds can be sampled with dip nets or seines. Dodd (2003) describes taking sweep samples (at 15m intervals) with a dip net around an entire pond. Sweep surveys provide information on: 1) larval species presence at the time of sampling; (2) life history information, such as when eggs are deposited and tadpole developmental stage; (3) habitat information, such as microhabitat preferences and distribution of various larvae; and, (4) in some cases, an estimate of density (number of animals ÷ the amount of area sampled in reference to available habitat). Sampling effort is easily quantified. Sampling in a stream can involve the use of a dip net or seine but can also include walking a transect turning over rocks and searching hiding places (beginning downstream and working upstream). Amphibians are the targeted group of animals, but reptiles may be collected with this technique as well.

Drift Fences at Breeding Sites
Drift fences at breeding sites can be employed in species inventories studies for species

with defined breeding periods. For some species of salamanders, breeding season offers the best opportunity to document their presence.

Where feasible, a temporary or permanent pond will be identified for sampling. If small enough the entire pond will be encircled with the drift fence. If encircling the pond is uneconomical only a portion of the pond will be fenced. If the entire pond is not encircled, it will be assumed that only a subset of the species using the pond is observed. The drift fence will be constructed to prevent the animals from crawling under or over the fence. Pitfalls (will be placed at intervals along both sides of the fence. The pitfalls will be of sufficient size to capture amphibians, though the actual size of the pitfall may be dependent on substrate. Drainage holes in the bottom of the pitfall will allow excess water to drain. A small piece of sponge may be placed in the pitfall to prevent drowning of non-aquatic species.   Where needed shading of the pitfall should be provided. Traps will be checked daily during the sampling period. Once the animal has been captured and noted it will be placed on the opposite side of the fence. Appendix A provides breeding periods for the salamanders documented from Tennessee. Drift fences at breeding sites will be opened for a period (target of 3 days) each month, September – June.

Artificial Cover Surveys

Coverboard surveys can be used to determine species occurrence, as well as relative abundance as catch per unit effort. They are effective in sampling species that require the moist environments found under such natural objects as rotting logs or rocks.

By setting out a standardized set of cover objects it is possible to determine the number of different species under a consistent uniform amount of cover.   Coverboards will be placed in an array or grid in a 5 x 5 pattern (Figure 1.). Coverboards will be of plywood or solid block of wood sized at 61cm x 61cm x 1.3cm (24” x 24” x .5”). Coverboards will be set no closer than 3m

□ □ □ □ □

□ □ □ □ □

□ □ □ □ □

□ □ □ □ □

□ □ □ □ □

Figure 1. Coverboard array.

to its nearest neighboring coverboard. In positioning the coverboard, the leaf litter will be removed and the boards will be placed in contact with the dirt substrate.   The GPS coordinates for each array will be taken at the center cover board.   An array must be in a location for at least a month prior to beginning a survey (Dodd 2002).

Coverboards will be checked three times monthly during the months of April and October, but may be checked periodically during the rest of the year when weather conditions are optimum for encountering amphibians. To reduce the frequency of disturbance, which reduces the efficiency of this method, coverboards will be run no more than once per 5-day period during these months. Amphibians are the target species, however, some reptiles and small mammals may also be found under coverboards. Any Animals encountered under the coverboard will be identified to species and enumerated. The coverboard will be re-positioned and the animal released at the edge of the board. Coverboard success generally increases after the boards have been in place for a period of time.

Reptiles

Visual Encounter Survey

The same VES methodology described in the amphibian section will be used to sample reptiles

Straight-Line Drift Fences with Traps and/or pitfalls

Drift fences can be used to determine species richness in an area and provide relative abundance data in the form of catch per unit effort. Straight-line drift fences are typically set in an intersecting array. A few examples are illustrated in Figure 2. A variety of configurations have been used in reptile inventories. A diagram of the array design used should be included in the reporting process. Cups or buckets may be used for pitfalls depending on the substrate. Silt fence material or aluminum flashing (50cm wide) may be used as the fence material. Fence lengths may be 5 to 20 meters long. The funnel traps can be double (Figure 3) or single ended and may be placed along the drift fence. Funnel traps should be shaded with pegboard or natural material.

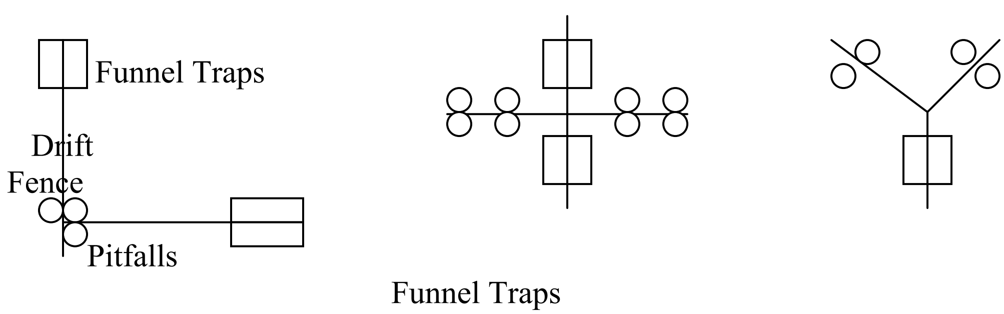

Figure 2. Perpendicular and Y straight-line drift fence using pitfalls and funnel traps. Configuration of traps and pitfalls can vary widely.

Figure 3. An example of a double-ended funnel trap. This trap was constructed of 3.2mm hardware cloth and fastened with zip ties. One funnel is removable and one is attached. The trap is 51 cm long.

Depending on topography, intersecting straight-line drift fences will be used to sample reptiles during the warm months of the year. During the period of September through June, drift fences will be opened for a period each month. A three-day trapping period should be targeted. Traps will be checked daily during the sampling period. As with any sampling method utilized, any animal occurrence will be noted.

Artificial Cover Surveys

Considerations for reptiles, such as increased body size and increased territory size, require coverboard surveys to be less structured than for amphibians. Coverboard material will vary in size and composition. For example, roofing tin (61cm X 122cm) or larger pieces of plywood provide excellent artificial cover for reptiles. Spacing for coverboards will be greater than for amphibians, but every effort will be made to utilize a grid pattern or material should be laid in a transect. Transects may be up to 150m in length with 10m between cover material. Coverboards targeting reptiles will be placed on top of the leaf litter in forested, as well as field, habitats. Arrays will be examined 3 times per month April - October. To reduce the frequency of disturbance, which reduces the efficiency of this method, coverboards will be run no more than once per 5-day period during these months.

Night Road Surveys
Road routes are an option on WMA's with road systems. These could be run in early spring (April/May) and early fall (Sept/Oct).
Heyer (1994) suggested a method where the investigator runs the route 3 times in a night with 15-minute intervals between runs. This method may be very effective at sampling snakes.

Aquatic Turtle Trapping
During the months of June through August, turtle traps will be placed in shallow water areas of streams and ponds within the sample area. Traps or hoop nets may be used. Traps will be set and checked daily for one week each month during the period. Turtles will be identified to species, enumerated and released.

Basking Turtle Surveys
Basking surveys provide species list and relative abundance data. Basking surveys may also help determine where future trapping may take place. Surveys may be conducted at Ponds or on Rivers. Surveys can be run between May and September. Binoculars and a spotting scope will be necessary for this technique.

Voucher Specimens
Photographs of specimens should be taken when possible. These photos can be used as vouchers for county records. With digital technology it should be possible to create a hard copy of the photo labeled with location data and stored in a notebook. New species occurrences (i.e. range extensions) will be documented by preserving a specimen then depositing the voucher in the herpetological collection at Austin Peay State University. Techniques for preparing amphibians as scientific specimens are described in Heyer et al. (1994, Appendix 4).

Mark and Recapture
At some point we will need to consider mark and recapture techniques. This aspect of our work should be carefully planned. The protocols for mark and recapture should be developed as an addition to this document further into our inventory and monitoring program.

Disinfecting Sampling Equipment and Other Sanitary Items
All equipment should be disinfected between sampling sites. A mild bleach solution or sanitary wipes may be used. Hands should be washed frequently and at a minimum between sampling sites. Only vinyl gloves should be used when handling amphibians because latex gloves are toxic to amphibians. Handling bags should not be reused. It is even suggested that seines be disinfected between sites (Dodd 2003).

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Species Codes

Species acronyms for use in recording occurrence data.

Symbol	Species
	Family Bufonidae - True Toads
AMTO	American Toad
FOTO	Fowler's Toad
	Family Hylidae – Treefrogs
NOCR	Northern Cricket Frog
SOCR	Coastal Plain (Southern) Cricket Frog
WBIV	Western Bird-voiced Treefrog
COGR	Cope's Gray Treefrog
GRET	Green Treefrog
BATR	Barking Treefrog
GRAT	Gray Treefrog
MOCH	Mountain Chorus Frog
NOSP	Northern Spring Peeper
UPCH	Upland Chorus Frog
	Family Microhylidae - Narrowmouth Frogs
ENAM	Eastern Narrow-mouthed Toad
	Family Pelobatidae – Spadefoots
EASP	Eastern Spadefoot
	Family Ranidae - True Frogs
NCRF	Northern Crawfish Frog
DUGO	Dusky Gopher Frog
AMBU	American Bullfrog
GRFR	Green Frog
PIFR	Pickerel Frog
SOLF	Southern Leopard Frog
WOFR	Wood Frog
	Ambystomatidae -Mole Salamanders
STSA	Streamside Salamander
SPSA	Spotted Salamander
MASA	Marbled Salamander
MOSA	Mole Salamander
SMSA	Small-mouthed Salamander
EATI	Eastern Tiger Salamander

	Amphiumidae -Amphiumas
THTO	Three-toed Amphiuma
	Cryptobranchidae -Hellbender
EAHE	Eastern Hellbender
	Plethodontidae -Lungless Salamanders
CMSA	Cumberland Mountain Salamander
CMDS	Carolina Mountain Dusky Salamander
SPDU	Spotted Dusky Salamander
NODU	Northern Dusky Salamander
IMSA	Imitator Salamander
SHSA	Shovel-nosed Salamander
SEAL	Seal Salamander
AMDS	Allegheny Mountain Dusky Salamander
OCSA	Ocoee Salamander
BRDS	Blue Ridge Dusky Salamander
BBSA	Black-bellied Salamander
SDSA	Santeetlah Dusky Salamander
BMSA	Black Mountain Salamander
PGSA	Pigmy Salamander
2LIN	Southern Two-lined Salamander
3LIN	Three-lined Salamander
JUSA	Junaluska Salamander
LTSA	Long-tailed Salamander
CASA	Cave Salamander
BR2L	Blue Ridge Two-lined Salamander
BECV	Berry Cave Salamander
TNCV	Tennessee Cave Salamander
SPRS	Spring Salamander
4TSA	Four-toed Salamander
MMSA	Midland Mud Salamander
RESA	Red Salamander
GRSA	Green Salamander
TESA	Tellico Salamander
CBSA	Cheoh Bald Salamander
ERBS	Eastern Red-backed Salamander
WSSS	White-spotted Slimy Salamander
NZSA	Northern Zigzag Salamander
NOSL	Northern Slimy Salamander

JOSA	Jordan’s Salamander
CUPL	Cumberland Plateau Salamander
MSSS	Mississippi Slimy Salamander
NGCS	Northern Gray-cheeked Salamander
SRSA	Southern Ravine Salamander
SRBS	Southern Red-backed Salamander
RELG	Red-legged Salamander
SASA	Southern Appalachian Salamander
SZZS	Southern Zigzag Salamander
WHSA	Wehrle’s Salamander
WESA	Weller’s Salamander
YOSA	Yonahlossee Salamander
	Proteidae -Waterdogs and Mudpuppy
MUDP	Common Mudpuppy
	Salamandridae -Newts
EANE	Eastern Newt
	Sirenidae -Sirens
WLSI	Western Lesser Siren
	Reptiles
	Anguidae - Glass Lizards and Alligator Lizards
EAGL	Eastern Slender Glass Lizard
	Iguanidae - Spiny Lizards
NFNL	Northern Fence Lizard
	Iguanidae -Anoles
GRAN	Green Anole
	Family Scincidae - Skinks
COSK	Coal Skink
5LSK	Common Five-lined Skink
SE5L	Southeastern Five-lined Skink
BRHD	Broad-headed Skink
FRSK	Ground Skink
	Teiidae - Whiptails & Racerunners
E6LI	Eastern Six-lined Racerunner
	Chelydridae - Snapping Turtles
EASN	Eastern Snapping Turtle
ALSN	Alligator Snapping Turtle

	Family Emydidae (Box and Water Turtles):
PATU	Painted Turtle
BOTU	Bog Turtle
NOMA	Northern Map Turtle
OUMA	Ouachita Map Turtle
FAMA	False Map Turtle
RICO	River Cooter
EABX	Eastern Box Turtle
POSL	Pond Slider
CUSL	Cumberland Slider
RESL	Red - Eared Slider
YBSL	Yellow-Bellied Slider
	Family Kinosternidae (Mud and Musk Turtles):
EAMU	Eastern Mud Turtle
COMU	Common Musk Turtle
SNMU	Stripe-necked Musk Turtle
	Family Trionychidae (Softshell Turtles):
MISS	Midland Smooth Softshell
SPSS	Spiny Softshell
	Viperidae - Pit Vipers
COHD	Copperhead
WECO	Western Cottonmouth
TIRA	Timber Rattlesnake
WPRA	Western Pigmy Rattlesnake
	Colubridae -
EAWO	Eastern Wormsnake
NOSC	Northern Scarlet Snake
EARA	Eastern Racer
RNSN	Ring-necked Snake
COSN	Cornsnake
RASN	Ratsnake
MUSN	Mud Snake
EHNS	Eastern Hog-nosed Snake
YEBE	Yellow-bellied Kingsnake
BLKI	Black Kingsnake
MISN	Milksnake
EACO	Eastern Coachwhip

MIGR	Mississippi Green Watersnake
PLBE	Plain-bellied Watersnake
BBWS	Broad-banded Watersnake
NDBW	Northern Diamond-backed Watersnake
NOWA	Northern Watersnake
NRGR	Northern Rough Greensnake
NOPI	Northern Pinesnake
QUSN	Queen Snake
DEBR	Dekay's Brownsnake
REBE	Red-bellied Snake
SOCR	Southeastern Crowned Snake
WRIB	Western Ribbonsnake
ERIB	Eastern Ribbonsnake
EGAR	Eastern Gartersnake
ROEA	Rough Earthsnake
SMEA	Smooth Earthsnake

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Conversion Factors

Convert From	Into	Multiply by
Acres	Hectare or square hetctometer	0.404687
Centemeter	Inches	0.3937
"	Millimeters	10
Feet	Meters	0.3048
Gallons	Liters	3.7854
"	Yards	0.333333
Grams	Ounces	0.03215
Hectares	Acres	2.471
"	Sqare meters	.00386
Inches	Centimeters	2.54
"	Millimeters	25.4
Kilometers	Miles (statute)	0.62137
Liter	Gallons	0.26417
Meters	Feet	3.28084
"	Yards	1.0936
Miles (statute)	Kilometers	1.609344
Ounce	Grams	31.1035
Quarts	Liters	0.94635
Square meters	Hectares	0.9991

Metric Equivalents for Various Units of Measure	
1 acre	= 0.404686 hectares
1,000 acres	= 404.686 hectares
1 cubic foot	= 0.028317 cubic meters
1,000 cubic feet	= 28.317 cubic meters
Breast Height	= 1.4 meters, or 4.5 feet above ground level

Species Breeding Information and Species Occurrence by Region

Salamander breeding periods.

	Spring			Summer			Fall			Winter		
Species	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec	Jan	Feb
Spotted Salamander										x	x	x
Marbled Salamander								x	x			
Mole Salamander										x	x	x
Small Mouth Salamander	x											x
Tiger Salamander									x			
Three Toed Amphiuma												x
Hellbender											x	
Mudpuppies										x	x	x
Green Salamander	x								x	x	x	x
Cumberland Mtn. Salamander	Spring						Autumn					
Dusky Salamander	Spring						Autumn					
Imitator Salamander	Spring						Autumn					
Seal Salamander		x					x	x				
Mountain Dusky Salamander	x					x		x				
Blackbelly Salamander							x					
Santeetlah Dusky Salamander												
Black Mountain Salamander												
Pygmy Salamander		x	x				x	x				
Southern Two-lined Salamander	x	x	x				x	x	x	x	x	x
Blue Ridge Two lined Salamander												
Junaluska Salamander	x							x				
Longtail Salamander									x	x	x	x
Cave Salamander					x	x	x	x				
Tennessee Cave Salamander												
Spring Salamander				x	x	x	x					
Four Toed Salamander												
Shovelnose Salamander												
Tellico Salamander												
Redback Salamander	x									x	x	x
N. ZigZag Salamander	x	x							x		x	x
Northern Slimy Salamander	x											
White spotted Slimy Salamander	x											
Mississippi Slimy Salamander	x											
Jordan's Salamander					x	x	x	x				
Cumb. Plat. Woodland Salamander					x	x	x	x				
Revine Salamander	x								x	x	x	x

Southern Redback Salamander	x											x
S. Appalachian Slimy Salamander												
Wehrle's Salamander	x	x										
Weller's Salamander								x				
Yonahlossee Salamander						x						
Mud Salamander							x					
Red Salamander							x					
Eastern Newt	x	x	x	x								
Lesser Siren	x									x	x	

Table 1. Salamander Species of the East Tennessee Region.		
Family Cryptobranchidae	*Giant Salamanders - Hellbenders*	
	Hellbender	**Cryptobranchus alleganiensis**
Family Proteidae	Giant Salamander- Mudpuppies and Waterdogs	
	Spotted Salamander	**Ambystoma maculatum**
	Marbled Salamander	*Ambystoma opacum*
	Mole Salamander	*Ambystoma talpoideum*
	Tiger Salamander	*Ambystoma tigrinum*
Family Salamandridae	Newts	
	Eastern Newt	**Notophthalmus viridescens**
Family Plethodontidae	*Lungless Salamanders*	
	Green Salamander	**Aneides aeneus**
	Seepage Salamander	*Desmognathus aeneus*
	Carolina Mountain Dusky Salamander	*Desmognathus carolinensis*
Contact zone for D. conanti & D. fuscus- hybrids likely	Spotted Dusky Salamander	*Desmognathus conanti*
	Northern Dusky Salamander	*Desmognathus fuscus*
	Imitator Salamander	*Desmognathus imitator*
	Shovel-nosed Salamander	*Desmognathus marmoratus*
	Seal Salamander	*Desmognathus monticola*
	Allegheny Mountain Dusky Salamander	*Desmognathus ochrophaeus*
	Ocoee Salamander	*Desmognathus ocoee*
	Blue Ridge Dusky Salamander	*Desmognathus orestes*
	Black-bellied Salamander	*Desmognathus quadramaculatus*
	Santeetlah Dusky Salamander	*Desmognathus santeetlah*
	Black Mountain Salamander	*Desmognathus welteri*
	Pigmy Salamander	*Desmognathus wrighti*
	Southern Two-lined Salamander	*Eurycea cirrigera*
	Three lined Salamander	*Eurycea guttolineata*
	Junaluska Salamander	*Eurycea junaluska*
	Long-tailed Salamander	*Eurycea longicauda*
	Cave Salamander	*Eurycea lucifuga*
	Blue Ridge Two-lined Salamander	*Eurycea wilderae*

*May soon be elevated to a separate species	Tennessee Cave Salamander (Berry Cave)	*Gyrinophilus palleucus guloineatus*
	Spring Salamander	*Gyrinophilus porphyriticus*
	Four-toed Salamander	*Hemidactylium scutatum*
	Tellico Salamander	*Plethodon aureolus*
	Eastern Red-backed Salamander	*Plethodon cinereus*
	White-spotted Slimy Salamander	**Plethodon cylindraceus**
	Northern Zigzag Salamander	*Plethodon dorsalis*
	Northern Slimy Salamander	*Plethodon glutinosus*
	Jordan's Salamander Plethodon jordani	
	Cumberland Plateau Salamander	*Plethodon kentucki*
*Likely to occur, but unconfirmed in Tennessee	Northern Gray-cheeked Salamander	*Plethodon montanus*
	Southern Ravine Salamander	*Plethodon richmondi*
	Southern Red-backed Salamander	*Plethodon serratus*
	Red-legged Salamander	*Plethodon shermani*
	Southern Appalachian Salamander	*Plethodon teyahalee*
	Southern Zigzag Salamander	*Plethodon ventralis*
	Wehrle's Salamander	*Plethodon wehrlei*
	Weller's Salamander	*Plethodon welleri*
	Yonahlossee Salamander	*Plethodon yonahlossee*
	Mud Salamander	*Pseudotriton montanus*
	Red Salamander	*Pseudotriton rubber*

Table 2. Salamander Species of the Middle Tennessee Region.		
Family Cryptobranchidae	*Giant Salamanders - Hellbenders*	
	Hellbender	**Cryptobranchus alleganiensis**
Family Sirenidae	*Sirens*	
	Lesser Siren	**Siren intermedia**
Family Proteidae	Giant Salamanders – Mudpuppies and Waterdogs	
	Mudpuppy	**Necturus maculosus**
Family Ambystomatidae	*Mole Salamanders*	
	Streamside Salamander	**Ambystoma barbouri**

	Spotted Salamander	*Ambystoma maculatum*
	Marbled Salamander	*Ambystoma opacum*
	Mole Salamander	*Ambystoma talpoideum*
	Small-mouthed Salamander	*Ambystoma texanum*
	Tiger Salamander	*Ambystoma tigrinum*
Family Salamandridae	Newts	
	Eastern Newt	**Notophthalmus viridescens**
Family Plethodontidae	**Lungless Salamanders**	
	Green Salamander	**Aneides aeneus**
	Northern Dusky Salamander	*Desmognathus fuscus*
	Southern Two-lined Salamander	*Eurycea cirrigera*
	Three-lined Salamander	**Eurycea guttolineata**
	Long-tailed Salamander	*Eurycea longicauda*
	Cave Salamander	*Eurycea lucifuga*
	Tennessee Cave Salamander	*Gyrinophilus palleucus*
* 1 record from Dekalb Co.	Spring Salamander	*Gyrinophilus porphyriticus*
	Four-toed Salamander	*Hemidactylium scutatum*
	Northern Zigzag Salamander	*Plethodon dorsalis*
	Northern Slimy Salamander	*Plethodon glutinosus*
	Mud Salamander	*Pseudotriton montanus*
	Red Salamander	*Pseudotriton ruber*

Table 3. Salamanders Species of the West Tennessee Region		
Family Cryptobranchidae	*Giant Salamanders- Hellbenders*	
* possible occurrence	Hellbender	**Cryptobranchus alleganiensis**
Family Amphiumidae	*Amphiumas*	
	Three-toed Amphiuma	**Amphiuma tridactylum**
Family Proteidae	*Giant Salamander – Mudpuppies and Waterdogs*	
	Mudpuppy	**Necturus maculosus**
Family Sirenidae	*Sirens*	
	Lesser Siren	**Siren intermedia**
Family Ambystomatidae	*Mole Salamanders*	
	Spotted Salamander	**Ambystoma maculatum**
	Marbled Salamander	**Ambystoma opacum**
	Mole Salamander	**Ambystoma talpoideum**
	Small-mouthed Salamander	**Ambystoma texanum**
	Tiger Salamander	**Ambystoma tigrinum**

Family Salamandridae	*Newts*	
	Eastern Newt	**Notophthalmus viridescens**
Family Plethodontidae	*Lungless Salamanders*	
	Spotted Dusky Salamander	**Desmognathus conanti**
	Southern Two-lined Salamander	**Eurycea guttolineata**
	Long-tailed Salamander	**Eurycea longicauda**
*Along the eastern edge of the West Tennessee Region only in karst areas.	***Cave Salamander***	**Eurycea lucifuga**
	Spring Salamander	**Gyrinophilus porphyriticus**
	Four-toed Salamander	**Hemidactylium scutatum**
	Northern Zigzag Salamander	**Plethodon dorsalis**
	Mississippi Slimy Salamander	**Plethodon Mississippi**
	Red Salamander	**Pseudotriton ruber**