Resources for Monitoring Pond-breeding Amphibians in the Northcentral USA

by

Melinda G. Knutson, James E. Lyon, and Jeffrey R. Parmelee

Farm Ponds as Critical Habitats for Native Amphibians: Final Report

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U.S. Geological Survey
Upper Midwest Environmental Sciences Center
2630 Fanta Reed Road
La Crosse, Wisconsin 54603

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Melinda G. Knutson and James E. Lyon

U.S. Geological Survey
Upper Midwest Environmental Sciences Center
2630 Fanta Reed Road
La Crosse, Wisconsin 54603

and

Jeffrey R. Parmelee

Simpson College
Department of Biology
701 North C Street
Indianola, Iowa 50125

Abstract
Public and private land managers are interested in monitoring amphibian populations to evaluate the risk of population declines. In this report, we describe monitoring methods and resources useful for biologists undertaking monitoring of amphibians breeding in pond environments in the northcentral USA. We include states in the U.S. Geological Survey Amphibian Research and Monitoring Initiative, Upper Mississippi Region (Illinois, Indiana, Iowa, Kansas, Kentucky, Michigan, Minnesota, Missouri, Nebraska, North Dakota, Ohio, South Dakota, and Wisconsin). The monitoring resources are derived from the literature and our experiences with a study of amphibians breeding in small farm ponds in southeastern Minnesota (Driftless Area Ecoregion) conducted from 2000 to 2001. We provide an overview of methods and list resources for conducting anuran calling surveys, egg mass surveys, larval surveys, and amphibian deformity assessments, and we list precautions to prevent the spread of diseases. We also present one method of collecting habitat information associated with a breeding site. The appendixes list equipment and resources useful for conducting amphibian surveys. Examples of data sheets are provided, along with a list of amphibians present in the northcentral USA.

Key words: amphibian, midwestern USA, monitoring, northcentral USA, pond, resources

Introduction

Declines in amphibian populations around the world, including some in the northcentral USA (Hay 1998; Lannoo 1998; Bury 1999; Alford et al. 2001) and high rates of deformed frogs in some locations (Helgen et al. 1998) have stimulated interest in amphibians as bioindicators of the health of ecosystems. Public and private land managers are interested in monitoring amphibian populations to evaluate the risk of population declines (Mossman et al. 1998).

We describe monitoring methods and resources useful for biologists undertaking monitoring of amphibians breeding in pond environments in the northcentral USA. We included states in the U.S. Geological Survey (USGS) Amphibian Research and Monitoring Initiative (ARMI), Upper Mississippi Region (Illinois, Indiana, Iowa, Kansas, Kentucky, Michigan, Minnesota, Missouri, Nebraska, North Dakota, Ohio, South Dakota, and Wisconsin). The monitoring resources are derived from the literature and our experiences in a study of amphibians breeding in small farm ponds in southeastern Minnesota (Driftless Area Ecoregion) conducted from 2000 to 2001 (Knutson et al. 2002).

As concern about amphibians increases, more agencies and herpetologists are engaged in monitoring activities. Amphibian monitoring methods are rapidly evolving because new research is focusing on improving monitoring methods. The USGS ARMI is monitoring amphibians across the USA and is a resource for monitoring methods (http://www.mp2-pwrc.usgs.gov/armi/index.cfm). The USGS Science Centers with active research on amphibians in the northcentral USA include Upper Midwest Environmental Sciences Center (La Crosse, Wisconsin), Northern Prairie Wildlife Research Center (Jamestown, North Dakota), National Wildlife Health Center (Madison, Wisconsin), and Columbia Environmental Research Center (Columbia, Missouri) (http://biology.usgs.gov/pub_aff/centers.html).

General Considerations

Anyone undertaking amphibian survey work has a responsibility to avoid harming the amphibians or their habitats. Persons planning to sample amphibians should work in cooperation with state or federal wildlife professionals. Lack of knowledge about sensitive habitats or populations could result in the spread of diseases, damage to breeding habitats, or local reproductive failure of amphibian populations. State and federal laws protect amphibians from exploitation. Collection permits are required from the appropriate state and/or federal authorities before collecting or handling amphibians. Consult your state wildlife management agency for guidance. Permission for sampling should also be obtained from the landowner.

Qualifications and Training

Biologists undertaking amphibian surveys should be familiar with the amphibian species in their area. A number of field guides and general herpetology references are available to assist biologists who are unfamiliar with amphibians (Wright and Wright 1949; Conant and Collins 1991; Stebbins and Cohen 1995; Harding 1997; Petranka 1998; Moriarty and Bauer 2000). Surveyors should be able to identify anurans by call and identify amphibian adults, eggs, and larvae in the field by sight or through the use of keys (Altig et al. 1998; Parmelee et al. 2002). In addition, skills in the identification of aquatic vegetation are useful. Training with a professional is strongly encouraged. Some universities offer herpetology courses as part of their academic program and some offer short summer courses at biological field stations. For biologists new to amphibian surveys, we recommend consulting herpetologists in your state to assist you.

Collecting and Handling

While performing amphibian surveys, it may be necessary to handle amphibian eggs, larvae, and adults. The following procedures will minimize the risk of injury to amphibians during collecting and handling (Fellers et al. 1994; Lips et al. 2001). Before handling amphibian eggs, larvae, or adults, wash your hands so they are free of soap, insect repellent, sunscreen lotion, and any other potential toxins. Hands should be moistened with water before handling any amphibians.

Handling of amphibian eggs should be minimized. When possible, identify eggs in place. Larvae should be handled with a dip net and not removed from the water for more than 2 min. During larval surveys, larvae can be held in buckets filled with pond water and placed in a cool place out of direct sunlight. Larvae should be released as soon as they are identified.

Preventing the Spread of Diseases

Disposable gloves should be used for handling animals when disease is suspected. To prevent the spread of potential pathogens or the introduction of novel species to new sites, animals should not be transported among sites. Any animals that are removed from the site for captive rearing or other purposes should not be released back into the environment. They should be euthanized and either preserved as voucher specimens, or disposed of properly (Green 2001).

If sampling will include contact between field gear (footwear, clothing, and equipment) and aquatic habitats, preventing contamination among sites is important. To prevent the spread of diseases from one amphibian population to another, all field gear should be cleaned and sanitized among study sites. The USGS National Wildlife Health Center (Madison, Wisconsin) has developed standard operating procedures for handling amphibians and disinfecting equipment (Green 2001). These guidelines also cover biosecurity precautions and reporting procedures if you suspect amphibian disease at a site. The Fieldwork Code of Practice developed by the Declining Amphibian Populations Task Force (http://www.npwrc.usgs.gov/narcam/techinfo/daptf.htm) also describes accepted safety precautions to take to prevent the spread of disease.

Sampling Design

Sampling design (where and how frequently to sample) may be the most important consideration in a monitoring study and determines what information can be derived from the data. Careful planning is especially important if you have specific management objectives for conducting the survey. If you are unsure about whether your planned design will meet your management objectives, consult references (Thompson et al. 1998; Yoccoz et al. 2001), a statistician, or a research biologist. The USGS Florida Caribbean Science Center (Gainesville, Florida) has investigated statistical design and analysis with respect to amphibian surveys. They describe issues related to sampling design on their Web site (http://www.fcsc.usgs.gov/armi/Framework/framework.html). Before you start, consider the types of habitats you want to include in your project or study, their size and distribution, and what maps are available showing these habitats. Stratified random sampling, aided by computer software, is often used to randomly select sample points from different habitat types.

Sampling and Recording Data

Standard survey techniques for amphibians include anuran calling surveys, egg mass surveys, larval surveys, and visual searches for adults (Heyer et al. 1994; Olsen et al. 1997). For those unfamiliar with amphibians, locating, collecting, and identifying amphibians (adults, eggs, larvae) can be challenging. We present resources for conducting amphibian surveys, including a list of field equipment (Appendix A), examples of field data sheets (Appendix B), resources for amphibian identification (Appendix C), amphibian species found in the northcentral USA (Appendix D), and species of management concern (Appendix E). Species names are based on Crother (2001).

Careful recording of the data collected during sampling is important for the effort to have any long-term value. The examples of data sheets (Appendix B) list the essential information to record. In the past, recording of sampling sites generally involved mapping on USGS quad sheets. Today, global positioning system (GPS) equipment makes it easy to record the spatial coordinates of sampling sites. We recommend recording location information at each site to accurately link your data with digital maps.

Anuran Calling Surveys

Anuran calling surveys are used to identify locations where adult frogs and toads are attempting to breed. Some states have been collecting anuran calling data over the last decade (Hemesath 1998; Mossman et al. 1998). Amphibian habitat associations have been derived from calling survey data (Knutson et al. 1999; Knutson et al. 2000), as well as population trend estimates (Mossman et al. 1998).

Anuran calling surveys are easier to perform than egg or larval surveys and are frequently conducted by volunteers. However, calling surveys do not provide evidence that breeding is successful. Eggs, larvae, and metamorphs are needed to confirm successful reproduction for anurans. Calling surveys are not used to survey salamanders because salamanders do not call. However, salamanders often breed in the same locations as anurans and may be detected by visual search or larval sampling.

Calling anurans can be heard in wetland habitats from early spring through midsummer. Frogs and toads (*Rana* and *Bufo* spp.) often conceal themselves in vegetation—including emergent vegetation, flooded grass, and shrubs—while calling. Treefrogs (*Hyla* spp.) also call from trees adjacent to breeding ponds. Most anuran calling surveys are conducted after dark. Headlamps are useful for keeping your hands free and for walking to breeding sites in the dark. Many anurans will also chorus during the day, especially at the peak of breeding activity.

Anurans make a variety of calls. Release calls are given by males of many species attempting to avoid accidental amplexus with other males. These calls are typically quieter than mating calls. The American Bullfrog and Northern Leopard Frog will sound alarm calls when approached or disturbed. Variations on mating calls are given by males trying to defend their calling territory. Most anuran call recordings will point out these differences. During daylight hours, bird songs may sound like amphibians. Later in the summer, a variety of insect calls must be distinguished from anuran calls.

Protocols for anuran calling surveys have been developed by the USGS North American Amphibian Monitoring Program (NAAMP 2002). Several states have state anuran calling programs that cooperate with North American Amphibian Monitoring Program. We recommend using protocols adopted by your state wildlife management agency so that your data are compatible with other, similar data collected in your state. Numerous resources, including sound recordings, are available to help you learn the calls for frogs in your area (Appendix C). Times and minimum air temperature guidelines are available to plan the timing of calling surveys in each state (NAAMP 2002).

Visual Encounter Surveys

Visual encounter surveys identify amphibian adults and possibly metamorphs at a site. The details of conducting visual searches have been described in several references (Crump and Scott 1994; Olsen et al. 1997).

Egg Mass Surveys

Egg mass surveys provide evidence that mating occurred. The number of egg masses is also an indication of the number of adults that bred at that location (Crouch and Paton 2000). Some amphibian species are most effectively surveyed by egg mass surveys because their egg masses are large and easily found (Crouch and Paton 2000). Searching for egg masses while attempting to locate calling individuals allows one to observe the relation among calling adult anurans, their eggs, and their choice of egg-laying sites. Polarized sunglasses help reduce glare when searching for eggs during the day.

Each species lays its eggs in characteristic ways (Stebbins and Cohen 1995). Most ranids lay their eggs in large masses, either in floating sheets or spherical masses near the water's surface, sometimes attached to vegetation. Toads lay eggs in long strings, typically in shallow water. Treefrogs lay their eggs in small masses or individually, attached to vegetation. Pond-breeding salamanders usually lay their eggs in masses attached to vegetation, at or below the water surface. While not all amphibians attach their eggs to vegetation, vegetation (living and dead) is often used for support by amphibians during the egg-laying process. As a result, pond-breeding amphibian eggs are usually found in association with vegetation. All pond-breeding amphibians in our region have pigmented eggs (Parmelee et al. 2002). Eggs or egg masses that are white or translucent are likely snail eggs that can be quite large.

Larval Surveys

Performing larval surveys is another method of detecting the presence of pond-breeding amphibians. The presence of larvae is good evidence that breeding was successful and that site conditions support larval development. There are a number of methods used to survey amphibian larvae (Heyer et al. 1994; Olsen et al. 1997). We recommend defining a search area for larval surveys. If your pond is small, you may want to search the entire pond. If your pond is large, you can define a search area, such as a 20-m diameter circle. Most amphibian larvae prefer shallower (<1 m depth) water, so shorelines and shallow areas should be your focus.

Dip nets or seines can be used to collect larvae. In our surveys, we attempted to standardize our dip net effort by placing all larvae collected during a 20-min dip net effort in a bucket. We then identified larvae by species and recorded their abundances (Appendix B).

The ability to successfully collect larvae depends on the density of larvae and the habitat characteristics. Small, temporary ponds may have relatively high densities of larval amphibians that can be collected with little effort. Larger, interconnected, permanent wetlands tend to have more dispersed populations of larval amphibians that increases the effort required.

Most amphibian larvae can be found among aquatic vegetation or other sheltering objects, where they seek food and refuge from predators. Toad tadpoles can often be seen in large schools in shallow, open water. Collecting amphibian larvae with a dip net requires walking carefully and slowly through the water, sweeping the net through stands of aquatic vegetation. In shallow, turbid, sparsely vegetated areas, larvae can often be found resting on the bottom. To prevent the escape of larvae, work from deeper water towards shallower areas. Immediately place collected larvae in a bucket containing water from the site. Put 2 to 3 L of water in the bucket and place it out of direct sunlight to prevent the larvae from overheating.

Funnel traps are another tool for collecting larvae (Adams et al. 1997). Funnel traps are useful when it is logistically feasible to deploy and check them regularly and when dense vegetation impedes the use of dip nets or seines. Because of the logistical considerations of sampling many sites, we collected the same species with less time using dip nets.

Identifying larvae in the field can be difficult for novices. Training by a herpetologist in the field is the best way to learn to identify larvae. Keys to amphibian larvae and eggs (Watermolen 1995, 1996; Parmelee et al. 2002) are useful in identifying species or groups of species. Some species can only be differentiated during the larval stage by examination of larval tooth patterns with the aid of a microscope (Altig et al. 1998; McDiarmid and Altig 1999). We recommend this only if you have training in amphibian larval identification. If you are unsure of your identifications, options include consulting a herpetologist or raising the larvae in the laboratory and making an identification from a metamorph or juvenile amphibian.

Amphibian Deformity Assessment

Recent concerns about amphibian deformities (Helgen et al. 1998; Johnson et al. 1999; Souter 2000; Rosenberry 2001; Johnson et al. 2002) have led management agencies to conduct deformity assessments to assess risks on public lands. Deformity assessments are usually performed on metamorphs from mid-June through mid-August. Accurate descriptions of any malformations you find are important for identifying causes (Meteyer 2000). The USGS North American Reporting Center for Amphibian Malformations provides guidance on how to conduct surveys for malformations and report your findings (http://www.npwrc.usgs.gov/narcam/).

Amphibian Disease Assessment

Amphibian disease is an emerging concern among herpetologists. Amphibian declines and species extinctions may be linked to novel and catastrophic diseases (Hero and Gillespie 1997; Daszak et al. 1999; Carey 2000; Green and

Sherman 2001; Kiesecker et al. 2001; Young et al. 2001). If you encounter a die-off or disease outbreak of amphibians, you should act quickly to have the problem diagnosed. The USGS National Wildlife Health Center (Madison, Wisconsin) is experienced in identifying amphibian pathogens. The Center has guidelines on handling and shipping specimens for diagnosis (Green 2001). Contact them for assistance before sending specimens.

Collecting Voucher Specimens

To verify the identification of eggs and larvae encountered in the field you will initially need to collect and preserve voucher specimens (McDiarmid 1994); (McDiarmid and Altig 1999; Simmons 2002 (in press)). A set of voucher specimens can be sent to a specialist for positive identification. Once you are confident in your identification skills, collections will not be necessary. Most states require collection permits issued by the state Department of Natural Resources or similar agency. The permits must be carried in the field during sampling and must accompany any preserved specimens. Remember to observe all wildlife laws and only collect where it is legal and where the collection of a few individuals will not affect the population. Species that are classified as endangered, rare, threatened, or of special concern (Appendix E) should be collected only with special permission from appropriate authorities.

Preserving Eggs and Larvae

Larvae should be anesthetized according to procedures recommended by Green (2001). There is no perfect preservative, and the techniques for preserving specimens are still debated (McDiarmid 1994; McDiarmid and Altig 1999). We recommend preserving amphibian eggs and larvae by placing them in a small vial filled with a 10% formalin solution. Alcohol is more pleasant to work with and safer than formaldehyde, but tends to dehydrate specimens. Whatever preservative you use, read the relevant Material Safety Data Sheets to learn how to safely handle and store that chemical.

Larvae can be placed individually, or as a lot of 5 to 20 individuals in screw top vials. Do not place too many individuals in one container. Immediate labeling is a must; use pencil or indelible ink on all submerged tags. Field tags should be linked to corresponding field notes; labels with detailed information must be kept with the specimens. Do not rely on your memory as a record of locality, date, and habitat information. The minimum information includes as follows: date, locality (kilometers from a crossroad or other landmark or GPS coordinates), habitat description, and name of the collector. We recommend maintaining a numbered log that links to tags on the vials. Other important information includes notes on live coloration (specimens quickly lose color in preservative). Specimens should be deposited in a museum or university collection where they can be appropriately cataloged, maintained, and available for researchers worldwide.

Habitat Assessment

Decisions about what habitat data to collect should be made by clarifying the research questions. Measuring habitat variables can be time-consuming. We tried various methods and found that simple habitat assessments were best, unless you have a specific need to be more detailed. The habitat assessment area should correspond to the area sampled for amphibians. Several references describe methods of collecting habitat information (Heyer et al. 1994; Olsen et al. 1997).

We present one example of measuring biotic and abiotic habitat variables at a site (Appendix B). The method is relatively simple and is based primarily on visual estimates of cover. Habitat assessments should be done after surveys for amphibians to avoid disturbing amphibians before the survey. Familiarity with aquatic vegetation is helpful (Fassett 1957; Borman et al. 1997; Chadde 1998), although we present estimates of cover by vegetative growth habit, not species or genera.

Cover information can be collected on the various types of vegetation (Appendix B). Vegetation is broadly defined as determined by plant habit (i.e., submerged, emergent, terrestrial, etc.). Information on substrate characteristics (sediment particle size estimates) can also be collected.

Canopy Cover

Visual estimates can be made of tree cover directly overhead, including overhanging canopy from trees with trunks located outside of the survey area. Canopy cover is estimated for woody vegetation >3 m in height. Because forest canopies often consist of multiple layers, we estimate total canopy cover and canopy cover above a height of 5 m (upper canopy). The estimate of upper canopy coverage may equal, but should not exceed the total canopy coverage.

Aquatic Habitat Cover

We estimated the total amount of aquatic habitat (habitat currently covered with water) contained within the sampling area.

Vegetation Cover

We also estimated vegetation cover for the entire sampling area, including submerged, floating–leaved (both rooted and nonrooted), emergent, woody/shrub (<3 m tall), and terrestrial vegetation (nonwoody vegetation including grasses and forbs). Because water levels may vary and aquatic plants may be found on dry substrates, plant categories can be determined according to growth preferences and not on hydrologic conditions present at the time of the assessment. The coverage of dormant woody vegetation can also be recorded.

Litter, Log, and Rock Cover

We estimated the coverage of dead leaf and plant litter, downed log, and rock cover for the entire sampling area of both aquatic and terrestrial portions of the site combined.

Water Depth

Because water depth usually varies across a sampling area, we suggest estimating water depth at five points randomly placed within the survey area. A measuring pole can be constructed from a PVC pipe. When measuring water depth, avoid resting the bottom of the measuring pole on submerged vegetation or large woody debris. If the water depth is greater than can be measured, record "Greater than" the maximum measurable depth.

Substrate Characterization

Underwater substrates can be characterized by particle size and organic content. Substrate type can be examined by sight and feel at the same five points used to determine average water depth. Only a small quantity (~ 2 cm^3) of substrate is needed for characterization and should be taken to a substrate depth of about 2 cm (Yin et al. 2000).

Landscape Context

The quality of the landscape surrounding your study site (context) is important to the persistence of amphibian populations. Persistence may be less likely if potential breeding sites are isolated or the surrounding landscape is potentially hostile to amphibians (row crops, major roads, industrial zones). If you record your survey site accurately with a GPS receiver, you will be able to evaluate the quality of the landscape surrounding your site using digital land cover maps and GIS software.

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Appendix A. Equipment List

Dip nets: 14 inches x 16 ½ inches aluminum frame with 24 inches aluminum handle. Net bag: 1/16 inches mesh, 18 inches deep. (Duraframe Dipnet, Viola, Wisconsin; 'intermediate wide teardrop')

Thermometer: Pocket alcohol thermometer with protective case, -10 to -110? C. (Fisher Scientific, Cat. No.15-021-5B)

Headlamp (Petzl "Duo").

Global Positioning System (GPS) receiver (Garmin GPS III, Garmin International, Olathe, Kansas).

PVC measuring pole: 2-m PVC pipe marked with centimeter gradations and fitted with 7.6-cm (3 inches) PVC pipe flange to prevent the measuring pole from sinking into soft sediments.

Plastic buckets: 3-5 gallon capacity.

10% buffered formalin (Fisher Scientific)
Directions for preparing:
http://www.jcu.edu.au/school/phtm/PHTM/frogs/pmfrog.htm - S4.

Glass specimen vials with plastic caps (Fisher Scientific).

Meter tape (25 m).

Watch or stop watch.

Sprayer for disinfectant (general duty 12-L capacity sprayer).

Hip and /or chest waders.
Small kayak: May be useful for surveying certain habitat types.

Amphibian call recordings (Appendix C).

Regional amphibian and reptile guides (Appendix C).

Covered clipboard.

Rite-in-the-rain paper.

Data sheets.

Collection permits.

Appendix B. Examples of Field Data Sheets

Study description:
Survey site location: ______________________ UTM coordinates: _______________ E _______________ N
UTM error: ____________________________ Datum:________________ Spheroid:_____________________
Habitat type: _________ Date begin: _________ end: _____ Time (e.g., 1600) begin: _________ end: ________ .
Observer initials:__________Recorder's initials:________ Temperature: Air _____? C Water _____? C
Sky conditions:_______ Wind speed: _______ Water present (Yes/ No) _______ (For road/trail calling surveys)
Data entered in computer (date): _______________ Data proofed (date) : ______________ Point ID # : ______
Check the assessments made:

Frog chorus survey		Specimens collected: (list species, numbers, and purpose)
Egg mass survey		
Larval survey		
Water quality		
Vegetation		
Deformity assessment		(Collection requires appropriate state and/or federal permits)

Calling Survey (5 min)

Species code	Species	Call index[a]	Notes

[a] 0 = No frogs of a given species can be heard calling.
1 = Individuals of a species can be heard; calls not overlapping.
2 = Individual frogs can be heard calling; but some overlap, can estimate number of frogs present.
3 = Full chorus; numerous frogs can be heard; chorus is constant and overlapping.

Additional Observations: Fill out for observations of other herpetofauna and for egg mass and larval surveys

Taxa (reptile, amphibian)	Life stage[a]	Species code	Species	Number[b]	Abundance code[c]	Notes[d]

[a] Life stage : egg, larva, metamorph, adult.
[b] Number: Total number of individuals or egg masses encountered.
[c] Abundance code: Larval survey, 0 (0), 1 (1–10), 2 (11–100), 3 (>100) Do not enter species name or code if species ID is not positively known.
[d] Notes: Enter information on sex of individuals, if known (m/f), or any other pertinent data.

Field Data Sheet (Page 2)

Additional Observations (Continued): Fill out for observations of other herpetofauna and for egg mass and larval surveys

Taxa (reptile, amphibian)	Life stage[a]	Species code	Species	Number[b]	Abundance code[c]	Notes[d]

[a] Life stage : egg, larva, metamorph, adult.
[b] Number: Total number of individuals or egg masses encountered.
[c] Abundance code: Larval survey, 0 (0), 1 (1–10), 2 (11–100), 3 (>100) Do not enter species name or code if species ID is not positively known.
[d] Notes: Enter information on sex of individuals, if known (m/f), or any other pertinent data.

Habitat Assessment

Water depth (centimeters):

Depth 1	Depth 2	Depth 3	Depth 4	Depth 5	Avg. Depth

Substrate characterization (codes 1–7[a]):

Substrate 1	Substrate 2	Substrate 3	Substrate 4	Substrate 5

[a] Silt/clay = 1, mostly silt with sand = 2, mostly sand with silt = 3, hard clay = 4, gravel = 5, sand = 6, organic muck = 7.

Canopy, vegetation, and litter cover (assessed for entire survey area):

Cover type	Cover class[a] (1–5)	
Trees/shrubs canopy cover	Upper (>5 m)	
	Total (>3 m)	
Aquatic habitat		
Floating-leaved		
Submerged		
Emergent		
Woody/shrubs (Less than 3 m tall)		
Terrestrial (grasses and forbs)		
Leaf and plant litter		
Downed log		
Rock		

[a] Visual estimate of coverage 1 = 1–20%, 2 = 21–40%, 3 = 41–60%, 4 = 61–80%, 5 = 81–100%.

Field Data Sheet (Page 3)

Beaufort Scale for determining wind speed:

Code	Wind speed kph	Wind speed mph	Indicators
0	0–2	0–1	Calm, smoke rises vertically.
1	3–5	2–3	Light air movement, smoke drifts.
2	6–11	4–7	Slight breeze, wind felt on face; leaves rustle.
3	12–19	8–12	Gentle breeze, leaves and small twigs in constant motion.
4	20–30	13–18	Moderate breeze, small branches are moved, raises dust and loose paper.
5	31–39	19–24	Fresh breeze, small trees in leaf begin to sway; crested wavelets form.
6	40–50	25–31	Strong breeze, large branches in motion.

Sky conditions codes (codes 3 and 6 are not used).

Code	Sky condition
0	Few clouds
1	Partly cloudy (scattered) or variable sky
2	Cloudy or overcast
3	
4	Fog or smoke
5	Drizzle or light rain (not affecting hearing ability)
6	
7	Snow
8	Showers (affecting hearing ability)

Codes for estimating vegetative cover:

Cover class	Visual estimate of coverage (%)
1	1–20
2	20–40
3	40–60
4	60–80
5	80–100

Field Data Sheet (Page 4)

Growth habit of representative taxa:

Habit	Representative taxa
Submerged	*Elodea* (water weeds), *Ceratophyllum* (Coontail), *Potamogeton* (pond weeds), Algae
Floating–leaved	Rooted: *Nymphae* and *Nuphar* (water lilies) Nonrooted: *Lemna* and *Spirodela* (Duckweed), Algae
Emergent	*Typha* spp. (Cattail), *Sagittaria* spp. (Arrow heads)
Woody/shrub (<3 m tall)	May include moist soil species such as *Salix* (Willow) or upland species such as *Cornus* (Dogwood). Also includes seedlings of tree species (i.e., *Acer* spp.).
Terrestrial (grasses and forbs)	May include moist soil species such as *Leersia* (cut-grass) or more upland species.

Substrate types and codes:

Substrate code	Substrate type and physical description
1	Silt/clay: Fine particle size, feels smooth when rubbed between fingers.
2	Mostly silt with sand: Material appears fine grained, but has slight gritty feel when rubbed between fingers
3	Mostly sand with silt: Sandy appearance, with finer material present. Feels gritty to the touch
4	Hard clay: Fine material, without gritty feel. Substrate tends not to be flocculent because of cohesiveness.
5	Gravel: Coarse substrate with particles between 3 and 32 mm.
6	Sand: Sandy appearance, gritty feel, no finer material (silt/clay) evident.
7	Organic muck: Dark or black smooth substrate. May contain some identifiable, but darkly stained plant material

Appendix C. Resources for Amphibian Identification

Some of this information is adapted from Moriarty and Bauer (2000).

National/Regional

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Appendix D. List of Amphibian Species Found in the Northcentral USA

Order	Family	Taxonomic order	IT IS number	Letter codes	Common name[a]	Scientific name
Caudata	Sirenidae	1000.9	173736	SIINTE	Lesser Siren	*Siren intermedia*
Caudata	Amphiumidae	1002.0	173612	AMTRID	Three-toed Amphiuma	*Amphiuma tridactylum*
Caudata	Proteidae	1004.0	208249	NEMACU	Mudpuppy	*Necturus maculosus*
Caudata	Cryptobranchidae	1006.0	208176	CRALLE	Hellbender	*Cryptobranchus alleganensis*
Caudata	Salamandridae	1008.0	888117	NOVIRI	Eastern Newt	*Notophthalmus viridescens*
Caudata	Ambystomatidae	1009.0	173594	ABANNU	Ringed Salamander	*Ambystoma annulatum*
Caudata	Ambystomatidae	1010.0	208204	ABBARB	Streamside Salamander	*Ambystoma barbouri*
Caudata	Ambystomatidae	1011.0	173598	ABJEFF	Jefferson Salamander	*Ambystoma jeffersonianum*
Caudata	Ambystomatidae	1012.0	173599	AMLATE	Blue-spotted Salamander	*Ambystoma laterale*
Caudata	Ambystomatidae	1013.0	173590	ABMACU	Spotted Salamander	*Ambystoma maculatum*
Caudata	Ambystomatidae	1014.0	173591	AMOPAC	Marbled Salamander	*Ambystoma opacum*
Caudata	Ambystomatidae	1016.0	173604	AMTALP	Mole Salamander	*Ambystoma talpoideum*
Caudata	Ambystomatidae	1017.0	173605	AMTEXA	Small-mouthed Salamander	*Ambystoma texanum*
Caudata	Ambystomatidae	1018.0	173593	AMTIGR	Tiger Salamander	*Ambystoma tigrinum*
Caudata	Plethodontidae	1021.0	173699	ANAENE	Green Salamander	*Aneides aeneus*
Caudata	Plethodontidae	1022.0	999104	DECONA	Spotted Dusky Salamander	*Desmognathus conanti*
Caudata	Plethodontidae	1023.0	173633	DEFUSC	Northern Dusky Salamander	*Desmognathus fuscus*
Caudata	Plethodontidae	1024.0	173641	DEOCHR	Allegheny Mountain Dusky Salamander	*Desmognathus ochrophaeus*
Caudata	Plethodontidae	1024.1	173634	DEWELT	Black Mountain Salamander	*Desmognathus welteri*
Caudata	Plethodontidae	1024.2	173640	DEMONT	Seal Salamander	*Desmognathus monticola*
Caudata	Plethodontidae	1025.0	173685	EUBISL	Northern Two-lined Salamander	*Eurycea bislineata*
Caudata	Plethodontidae	1026.0	550246	EUCIRR	Southern Two-lined Salamander	*Eurycea cirrigera*
Caudata	Plethodontidae	1027.0	173687	EULONG	Long-tailed Salamander	*Eurycea longicauda*
Caudata	Plethodontidae	1028.1	173687	EUGUTT	Three-lined Salamander	*Eurycea guttolineata*
Caudata	Plethodontidae	1029.0	208311	EULUCI	Cave Salamander	*Eurycea lucifuga*
Caudata	Plethodontidae	1030.0	208314	EUMULT	Many-ribbed salamander	*Eurycea multiplicata*

Order	Family	Taxonomic order	IT IS number	Letter codes	Common name[a]	Scientific name
Caudata	Plethodontidae	1031.0	173697	EUTYNE	Oklahoma Salamander	*Eurycea tynerensis*
Caudata	Plethodontidae	1032.0	208353	GYPORD	Spring Salamander	*Gyrinophilus porphyriticus*
Caudata	Plethodontidae	1034.0	173678	HESCUT	Four-toed Salamander	*Hemidactylium scutatum*
Caudata	Plethodontidae	1035.0	208278	PLALBA	Western Slimy Salamander	*Plethodon albagula*
Caudata	Plethodontidae	1036.0	173649	PLCINE	Eastern Red-backed Salamander	*Plethodon cinereus*
Caudata	Plethodontidae	1037.0	999112	PLDORS	Northern Zigzag Salamander	*Plethodon dorsalis*
Caudata	Plethodontidae	1039.0	173650	PLGLUT	Northern Slimy Salamander	*Plethodon glutinosus*
Caudata	Plethodontidae	1039.1	173661	PLKENT	Cumberland Plateau Salamander	*Plethodon kentucki*
Caudata	Plethodontidae	1039.2	208289	PLMISS	Mississippi Slimy Salamander	*Plethodon mississippi*
Caudata	Plethodontidae	1040.0	173667	PLRICH	Southern Ravine Salamander	*Plethodon richmondi*
Caudata	Plethodontidae	1041.0	173668	PLSERR	Southern Red-backed Salamander	*Plethodon serratus*
Caudata	Plethodontidae	1042.0	173634	PLWEHR	Wehrle's Salamander	*Plethodon wehrlei*
Caudata	Plethodontidae	1043.0	208302	PSMOND	Mud Salamander	*Pseudotriton montanus*
Caudata	Plethodontidae	1044.0	173681	PSRUBE	Red Salamander	*Pseudotriton ruber*
Caudata	Plethodontidae	1045.0	173730	TYSPEL	Grotto Salamander	*Typhlotriton spelaeus*
Anura	Pelobatidae	1046.0	173426	SCHOLB	Eastern Spadefoot	*Scaphiopus holbrookii*
Anura	Pelobatidae	1047.0	206989	SPBOMB	Plains Spadefoot	*Spea bombifrons*
Anura	Microhylidae	1048.0	173467	GACARO	Eastern Narrow-mouthed Toad	*Gastrophryne carolinensis*
Anura	Microhylidae	1049.0	173468	GAOLIV	Great Plains Narrow-mouthed Toad	*Gastrophryne olivacea*
Anura	Bufonidae	1050.0	173473	BUAMER	American Toad	*Bufo americanus*
Anura	Bufonidae	1052.0	173484	BUCOGN	Great Plains Toad	*Bufo cognatus*
Anura	Bufonidae	1053.0	173487	BUHEMI	Canadian Toad	*Bufo hemiophrys*
Anura	Bufonidae	1054.0	173478	BUFOWL	Fowler's Toad	*Bufo fowleri*
Anura	Bufonidae	1055.0	173476	BUWOOD	Woodhouse's toad	*Bufo woodhousii*
Anura	Hylidae	1056.0	173522	ACCREP	Northern Cricket Frog	*Acris crepitans*
Anura	Hylidae	1057.0	173511	HYAVIV	Bird-voiced Treefrog	*Hyla avivoca*
Anura	Hylidae	1058.0	173502	HYCHRY	Cope's Gray Treefrog	*Hyla chrysoscelis*
Anura	Hylidae	1059.0	173505	HYCINE	Green Treefrog	*Hyla cinerea*
Anura	Hylidae	1060.0	173503	HYVERS	Gray Treefrog	*Hyla versicolor*

Order	Family	Taxonomic order	IT IS number	Letter codes	Common name[a]	Scientific name
Anura	Hylidae	1060.1	173508	HYGRAT	Barking Treefrog	*Hyla gratiosa*
Anura	Hylidae	1061.0	173528	PSBRAC	Mountain Chorus Frog	*Pseudacris brachyphona*
Anura	Hylidae	1062.0	207304	PSCRUC	Spring Peeper	*Pseudacris crucifer*
Anura	Hylidae	1063.0	207301	PSSTRE	Strecker's Chorus Frog	*Pseudacris streckeri*
Anura	Hylidae	1064.0	207310	PSFERI	Southeastern Chorus Frog	*Pseudacris feriarum*
Anura	Hylidae	1065.0	207312	PSMACU	Boreal Chorus Frog	*Pseudacris maculata*
Anura	Hylidae	1066.0	173525	PSTRIS	Western Chorus Frog	*Pseudacris triseriata*
Anura	Ranidae	1067.0	207006	RAAREA	Crawfish Frog	*Rana areolata*
Anura	Ranidae	1068.0	173448	RABLAI	Plains Leopard Frog	*Rana blairi*
Anura	Ranidae	1069.0	173441	RACATE	American Bullfrog	*Rana catesbeiana*
Anura	Ranidae	1070.0	207002	RACLAM	Green Frog	*Rana clamitans*
Anura	Ranidae	1072.0	173435	RAPALU	Pickerel Frog	*Rana palustris*
Anura	Ranidae	1073.0	173443	RAPIPI	Northern Leopard Frog	*Rana pipiens*
Anura	Ranidae	1074.0	173460	RASEPT	Mink Frog	*Rana septentrionalis*
Anura	Ranidae	1075.0	173436	RASPHE	Southern Leopard Frog	*Rana sphenocephala*
Anura	Ranidae	1076.0	173440	RASYLV	Wood Frog	*Rana sylvatica*

[a]Adapted from Lannoo (1998), Crother (2000), and the Integrated Taxonomic Information System (ITIS).
All amphibians found in the northcentral USA are included, not only pond-breeders.
Names follow Crother (2000).
States include Iowa, Illinois, Indiana, Kansas, Kentucky, Michigan, Minnesota, Missouri, Nebraska, North Dakota, Ohio, South Dakota, and Wisconsin (U.S. Geological Survey Amphibian Research and Monitoring Initiative, Upper Mississippi Region).
The list may not be comprehensive for every state and is subject to revision.

Appendix E. State Conservation Status of Amphibian Species Found in the Northcentral USA

Common name	Scientific name	Status by state[a]												
		MO	IA	IL	IN	OH	KS	KY	MN	WI	MI	ND	SD	NE
Lesser Siren	*Siren intermedia*	P		P	P			P			X			
Three-toed Amphiuma	*Amphiuma tridactylum*	R												
Mudpuppy	*Necturus maculosus*	P	E	P	SPC	P	P	P	P	P	P	P	P	P
Hellbender	*Cryptobranchus alleganensis*	R		E	E	E		P						
Eastern Newt	*Notophthalmus viridescens*	P	E	P	P	P	T	P	P	P	P			
Ringed Salamander	*Ambystoma annulatum*	R												
Streamside Salamander	*Ambystoma barbouri*				P	P		P						
Jefferson Salamander	*Ambystoma jeffersonianum*			P	P	P		P			P			
Blue-spotted Salamander	*Ambystoma laterale*		E	P	SPC	E			P	P	P			
Spotted Salamander	*Ambystoma maculatum*	P		P	P	P		P		P	P			
Marbled Salamander	*Ambystoma opacum*	P		P	P	P		P			T			
Mole Salamander	*Ambystoma talpoideum*	R		P				P						
Small-mouthed Salamander	*Ambystoma texanum*	P	P	P	P	P	P	P			E			
Tiger Salamander	*Ambystoma tigrinum*	P	P	P	P	P		P	P	P	P	P	P	P
Green Salamander	*Aneides aeneus*				E	E								
Spotted Dusky Salamander	*Desmognathus conanti*			E				P						
Northern Dusky Salamander	*Desmognathus fuscus*				P	P		P						
Allegheny Mountain Dusky Salamander	*Desmognathus ochrophaeus*					P		P						
Black Mountain Salamander	*Desmognathus welteri*							P						
Seal Salamander	*Desmognathus monticola*							P						
Northern Two-lined Salamander	*Eurycea bislineata*					P		P						
Southern Two-lined Salamander	*Eurycea cirrigera*			P	P	P		P						
Long-tailed Salamander	*Eurycea longicauda*	P		P	P	P	T	P						
Three-lined Salamander	*Eurycea guttolineata*							P						
Cave Salamander	*Eurycea lucifuga*	P		P	P	E		P						
Many-ribbed salamander	*Eurycea multiplicata*	P					E							

Common name	**Scientific name**	**Status by state[a]**												
		MO	**IA**	**IL**	**IN**	**OH**	**KS**	**KY**	**MN**	**WI**	**MI**	**ND**	**SD**	**NE**
Oklahoma Salamander	*Eurycea tynerensis*	P					P							
Spring Salamander	*Gyrinophilus porphyriticus*					P		P						
Four-toed Salamander	*Hemidactylium scutatum*	P		T	E	SPC		P	SPC	SPC	SPC			
Western Slimy Salamander	*Plethodon albagula*	P												
Eastern Red-backed Salamander	*Plethodon cinereus*			P	P	P		P	P	P	P			
Northern Zigzag Salamander	*Plethodon dorsalis*	P		P	P			P						
Northern Slimy Salamander	*Plethodon glutinosus*			P	P	P		P						
Cumberland Plateau Salamander	*Plethodon kentucki*							P						
Mississippi Slimy Salamander	*Plethodon mississippi*							P						
Southern Ravine Salamander	*Plethodon richmondi*				P	P		P						
Southern Red-backed Salamander	*Plethodon serratus*	P												
Wehrle's Salamander	*Plethodon wehrlei*					P		P						
Mud Salamander	*Pseudotriton montanus*					P								
Red Salamander	*Pseudotriton ruber*				E	P		P						
Grotto Salamander	*Typhlotriton spelaeus*	P												
Eastern Spadefoot	*Scaphiopus holbrookii*	R		P	SPC	E	P	P						
Plains Spadefoot	*Spea bombifrons*	P	P									P	P	P
Eastern Narrow-mouthed Toad	*Gastrophryne carolinensis*	P		P			P							
Great Plains Narrow-mouthed Toad	*Gastrophryne olivacea*	P					T							
American Toad	*Bufo americanus*	P	P	P	P	P	P	P	P	P	P	P	P	P
Great Plains Toad	*Bufo cognatus*	P	P						P			P	P	P
Canadian toad	*Bufo hemiophrys*						P		P			P	P	P
Fowler's Toad	*Bufo fowleri*	P	P	P	P	P	P	P			P			
Woodhouse's toad	*Bufo woodhousii*	P	P					P				P	P	P
Northern Cricket Frog	*Acris crepitans*	P	P	P	P	P	P	P	E	E	PRO		P	P
Bird-voiced Treefrog	*Hyla avivoca*			P				P						
Cope's Gray Treefrog	*Hyla chrysoscelis*	P	P	P	P	P	P	P	P	P	P		P	P
Green Treefrog	*Hyla cinerea*	P		P			P	P						

Common name	Scientific name	Status by state[a]												
		MO	IA	IL	IN	OH	KS	KY	MN	WI	MI	ND	SD	NE
Gray Treefrog	*Hyla versicolor*	P	P	P	P	P		P	P	P	P	P	P	P
Barking Treefrog	*Hyla gratiosa*							P						
Mountain Chorus Frog	*Pseudacris brachyphona*					P								
Spring Peeper	*Pseudacris crucifer*	P	P	P	P	P	P	P	P	P	P			
Strecker's Chorus Frog	*Pseudacris streckeri*	R		T										
Southeastern Chorus Frog	*Pseudacris feriarum*	P		P	P	?	P							
Boreal Chorus Frog	*Pseudacris maculata*		P						P	P	SPC	P	P	P
Western Chorus Frog	*Pseudacris triseriata*	P	P	P	P	P	P		P	P	P	P	P	P
Crawfish Frog	*Rana areolata*	R	E	P	E		T	P						
Plains Leopard Frog	*Rana blairi*	P	P	P	SPC		T				P		P	P
American Bullfrog	*Rana catesbeiana*	P	P	P	P	P		P	P	P	P		P	P
Green Frog	*Rana clamitans*	P	P	P	P	P	P	P	P	P	P			
Pickerel Frog	*Rana palustris*	P	P	P	P	P	P	P	P	P	P			
Northern Leopard Frog	*Rana pipiens*	R	P	P	SPC	P	P	P	P	P	P	P	P	P
Mink Frog	*Rana septentrionalis*						P		P	P	P			
Southern Leopard Frog	*Rana sphenocephala*	P	P	P	P	?	P	P						
Wood Frog	*Rana sylvatica*	R		P	P	P		P	P	P	P	P	P	

[a]Status: P = Present, E = Endangered, R = Rare, T = Threatened, PRO = Protected, SPC = Special concern, X = Presumed extirpated, ? = Status unknown.
The list is adapted from field guides and state Web sites and is subject to revision. All amphibians found in the northcentral USA are included, not only pond-breeders.