SURVEYING AND MONITORING AMPHIBIANS USING AQUATIC FUNNEL TRAPS

MICHAEL J. ADAMS

University of Washington
College of Forest Resources, Box 352100
Seattle, Washington 98195-2100

KLAUS O. RICHTER

King County Natural Resources Division
506 Second Avenue
707 Smith Tower Building
Seattle, Washington 98104

AND

WILLIAM P. LEONARD

Department of Natural Resources
Washington Natural Heritage Program
P.O. Box 47016
Olympia, Washington 98504-7016

INTRODUCTION

In many situations, aquatic funnel trapping is the simplest and most repeatable method for surveying and monitoring pond-breeding amphibians. Its use only requires that a worker visit a site on two subsequent days and have received basic instruction in trap placement protocol and amphibian identification. Perhaps its greatest advantage is that, unlike various methods involving hand capture or direct observation, the skill and experience of the worker will have little impact on capture success. Thus, trapping protocols can be consistently repeated over time or among sites.

Funnel trapping is suitable for almost any survey or monitoring situation but is especially recommended for sites where dense vegetation or woody debris limit visibility and inhibit dip-netting. This is the typical situation in lowland areas but also occurs at higher elevations (e.g. montane lakes with large amounts of rock or tree structure). Traps are also useful because they can be left overnight when many amphibians appear most active.

Our experience suggests that funnel traps will reliably determine the presence of most pond-breeding amphibians in the Pacific Northwest and are capable of detecting small populations of cryptic larvae that might go unnoticed with other techniques (Adams et al.

in prep). Trap rates can sometimes be used as a rough index of relative abundance, but the assumption of equal capture probability over time is likely to be violated in monitoring programs (untested). Moreover, detected/not-detected may be a better response variable for monitoring than relative abundance in many situations (e.g. most broad spatial-scale monitoring programs or when the population of interest is regulated in the adult stage)(see Fellers, this vol.).

A disadvantage of funnel trapping is that workers must be able to identify larval amphibians since few adults are captured. Most larvae in the Pacific Northwest are identifiable to species, but some species can be difficult to distinguish during some stages of development. At low elevations west of the Cascades, it is best to trap at least once in mid-winter to detect long-toed salamander (*Ambystoma macrodactylum*) adults in the breeding season. Their larvae will be present through mid-summer, but can be difficult to distinguish from small larvae of northwestern salamanders (*A. gracile*). A tadpole caught at some mid-elevation areas in the Cascades, might be one of three species of native ranid frogs. The tadpoles of these species cannot be reliably distinguished in the field while still early in development (but see Corkran and Thoms 1996).

Other disadvantages are the need for many traps, which can require a substantial time or monetary investment, and the requirement that traps be left overnight. Also, some trap mortality will occur and can be high if traps are misused.

Here, we describe three types of aquatic funnel trap and discuss their use in the Pacific Northwest.

FUNNEL TRAP DESIGNS

Aquatic funnel traps employ a holding chamber with one or two tapered mouths that channel organisms toward a small entrance to the trap interior. Once inside, organisms tend to aggregate around edges where they cannot find the hole through which to escape. They are most effective at capturing larval amphibians, but juveniles and adults are also captured with some frequency. These traps are also useful for capturing invertebrates that prey upon larval amphibians. There are many types of funnel trap, but two general designs are widely used for pond amphibians and a third holds promise (Fig. 1).

The first type of aquatic funnel trap is the commercially available minnow-trap. These are constructed from either 1/4-inch (6 mm) mesh plastic or galvanized-steel hardware-cloth (ca. $8 U.S.). The coarse mesh allows water to circulate through the trap and thereby reduces trap mortality compared to more sealed designs. Minnow traps have funnels at both ends that taper from a 19-cm diameter opening down to a 2.5-cm entrance-hole. Traps can be opened in the middle to remove captured organisms and the two halves can be nested within each other and within other traps for transport. The entrance hole can be enlarged to accommodate large bullfrog tadpoles (*Rana catesbeiana*) or salamander larvae. A disadvantage of minnow traps is that the mesh covering is too coarse to retain larvae in early stages of development and some small species cannot be captured until late stages (*Taricha granulosa* and *Bufo boreas* larvae may never be large enough for capture in this design).

A smaller but less expensive trap can be constructed from 2-liter pop bottles (Richter 1995). This design is capable of capturing the smallest amphibian larvae but limits water circulation and, thus, may increase trap mortality when water temperatures are warm or trap rates high. A pocket of air can be trapped inside so that adult amphibians, late-development larvae, and garter snakes can breath. Because of their light weight, pop-bottle traps can be attached to vegetation or wood-dowels to trap higher in the water column or near the surface; their orientation can be either horizontal or vertical. However, the mouths of pop-bottle traps are not wide enough for the largest bullfrog tadpoles, neotenic salamanders, and adult ranid frogs.

A third type of funnel trap has only recently come into use but appears the most useful for amphibian sampling. It is constructed of nylon webbing (3-mm mesh) wrapped around a spring-loaded wire frame. Because of the spring, the trap is collapsible but will hold its shape when expanded. It has square funnels on both ends with entrance holes of approximately 5-cm diameter. A zippered pouch is provided for baiting if desired. Traps are available from commercial net suppliers (ca. $8 U.S.). Advantages of this design are its light weight and fine mesh which will contain small larvae while still allowing water to circulate through the trap. Capture rates of the collapsible trap are equal to or better than wire minnow traps in lowland ponds and their light weight, collapsible design makes them suitable for backcountry work (R. B. Bury and C. Pearl, pers. comm.; W. Leonard and L. Hallock, unpubl. data). Bury and Major (this vol.) use this funnel trap type for amphibian inventory and monitoring in National Parks of the Pacific Northwest.

The funnel area of various trap designs differs and this effects the number of traps needed. For example, pop-bottle traps are smaller in diameter (10cm) than minnow traps and have only a single funneled entrance, so up to seven may be needed to equal the trap rate of a single minnow trap. This conversion is based solely on a comparison of the funnel areas (567 cm^2 for a minnow trap vs. 78 cm^2 for a pop-bottle trap) and assumes that trap rate is a linear function of this area. Capture differences between pop-bottle and minnow traps may decrease if there is a benefit of simply trapping at more points within a wetland when pop-bottle traps are used. Also, the use of bait may reduce this ratio since capture rates will be less dependent on funnel area. Thus, while two pop-bottle traps do not have as much funnel area as one minnow trap, they may catch the same number of organisms if there is an increased catch from trapping at two points rather than just one (holding funnel-area constant) or if bait draws the animals in (see discussion of bait below).

Regardless of the true catch ratio, it should be noted that trap types are not interchangeable in terms of capture efficiency. Because of these differences, we recommend using fewer minnow traps than pop-bottle traps for a given area (see Habitat Based Protocol below). Despite the increase in capture efficiency with collapsible traps, we do not feel that fewer can be used without compromising coverage.

TRAPPING PROTOCOLS

Basics

A trapping protocol determines when and where traps should be set and how many traps should be used. It should be designed based on the goal of trapping (i.e., inventory vs. monitoring), the species that could potentially be encountered, and the level of precision desired (probability of detecting all species that are present). A workable protocol will be specific to a general class of waters but will be flexible enough to accommodate all of the habitat types that may be encountered within that class (e.g., a protocol for high alpine lakes will be different from one for lowland marshes). Likewise, a protocol for conducting an inventory might differ from that for long-term monitoring although we recommend designing inventories with monitoring in mind whenever possible.

Target Species

The first steps in any survey are to identify the sites or region to be sampled, to identify the species that may occur there, and to determine the activity periods of those species at the survey site(s). These steps are described earlier (Thoms and Corkran, this vol.). Next, since the focus of funnel trapping is on larval amphibians, the time interval between hatching and metamorphosis must be estimated. Trapping will be most effective between these dates. If minnow traps are to be used, then trapping cannot begin until larvae are large enough to be retained by 6-mm mesh. Exceptions are *Rana catesbeiana* and *Ambystoma gracile* whose larvae are generally present and active all year. Also, some species can be surveyed by trapping breeding adults (e.g., *A. macrodactylum*).

Timing

We suggest surveying a site approximately three different nights over 4 - 9 months at lower elevations; 3 months in montane habitats. A lowland trapping protocol (west of Cascades) might include samples in late winter (breeding *A. macrodactylum*), mid-spring (*R. catesbeiana*, *A. gracile* larvae, *T. granulosa* adults), and early summer (native tadpoles). Many of these species can be caught during other seasons, but trapping during multiple seasons increases the total trapping effort and the probability of detecting all potential amphibians. Also, since weather conditions may affect trap rates (untested), trapping on multiple nights will help ensure that a species is not missed due to poor trapping conditions. With fewer trapping dates or shorter intervals between dates it becomes more important to accurately estimate the optimal trapping period for target species.

Baiting Traps

Traps are sometimes baited (fish eggs, clam strips, etc.) to attract amphibians. However, the effect of bait on capture rates is unknown and its effectiveness likely varies between taxa. Unbaited traps rely upon chance movements of animals within the water column to bring them into a funneled entrance. We have successfully trapped most species with both baited and unbaited funnel traps. Some rudimentary tests suggest no attraction to baited traps for *A. gracile* larvae, *A. macrodactylum* adults, *T. granulos*a adults, *R. catesbeiana* tadpoles, and Pacific treefrog (*Hyla regilla*) tadpoles (Table 1). However, fresh salmon eggs did significantly increase traps rates for tiger salamander (*Ambystoma tigrinum*) larvae in one experiment. Usefulness of bait for inventories needs further examination.

Bait effectiveness may vary with food availability at the site trapped, bait type, and water movements, which in turn can differ over time and among sites. Thus, we recommend that bait not be used in monitoring programs since it adds another variable that can cause bias.

Trap Placement

We recommend placing traps in water depths ranging from the minimum depth necessary to allow amphibians to swim into the opening of the trap (depends on type of trap used) to a maximum of 1 m. In our experience, traps placed in deeper water rarely catch additional taxa. Traps should be left out overnight when trap rates appear highest. The amount of time traps are left in the water should be roughly standardized and should range from 12 to 24 hours.

Aquatic funnel traps are potentially deadly to amphibians and other aquatic animals if traps are lost, forgotten or inadequately monitored. Several simple steps should be taken to ensure that traps are not lost. Trap locations should be marked by means of floats attached to the trap or strings that are tied to vegetation. Alternatively, colored flagging can be tied to nearby vegetation or pin flags can be used to mark trap locations. Care should be taken to secure traps when working in flowing water or on steep depth gradients so they are not carried off and lost. Traps should never be left for more than 24 hours without being checked and we suggest 12 hours is optimal.

When traps are checked regularly, mortality will generally be low, but in certain situations may become unacceptably high. The amount of mortality that is tolerable will depend on the importance of the site to the species that occur there (e.g., zero mortality should be a primary goal at the last known site for a species), landowner/manager preferences, and the personal beliefs of the surveyors.

In general, try to avoid the following situations. First, avoid using pop-bottle traps in warm water (mid-summer) or when many animals (especially fish) will be caught. A combination of these conditions in particular can lead to very high mortality due to asphyxiation. Second, avoid complete submersion of mesh traps during the breeding seasons of most species when many adults might be captured. Long-toed salamanders are an exception and can survive prolonged submersion; perhaps because of the cold water temperatures during their breeding season. Third, in some simple wetlands it may be possible to avoid complete submersion of mesh traps when the same habitat type is found in shallower water. We suggest that this approach is seldom justified. Amphibians are abundant out to ca. 75-cm depth and confining traps to shallower waters may cause a systematic bias in captures (untested). Moreover, some important habitat types do not occur in waters shallow enough to avoid submersion. Overlooking a population will often be more detrimental than sacrificing a few individuals and the trap-placement protocols we describe below do not follow the third suggestion.

Determining where to place traps within a wetland is one of the most important features of a trap-placement protocol. It must insure that all habitats are adequately surveyed and, for monitoring programs, that bias is minimized. The next two sections describe two protocols and discuss their strengths and weaknesses.

SHORELINE-BASED PROTOCOL

Shaffer et al. (1994) have described a monitoring protocol that determines the number of traps (or other sampling techniques) needed and where to place them in ponds and lakes. It includes a randomization scheme that appears suitable for replication of effort over time or among sites without bias. Their technique can also be adapted for inventories when there is a well-defined shoreline and depth gradient.

With their technique, a 100-m segment of shoreline is selected and divided into five 20-m segments (each divided into four depth zones). A trap is randomly placed within each depth zone of each 20-m segment. The 100-m segment that is selected may be of special interest or it may be considered representative of the site.

Selecting a single 100-m segment of a wetland to monitor cannot provide statistical inference for the entire wetland and is inadequate for an inventory. Instead, we suggest randomly placing the 20-m segments throughout the wetland. Alternatively, it may be possible to sample the entire shoreline at smaller sites. The number of segments used will depend on the variability of the shoreline and the level of precision desired. Enough segments should be used so that all of the major habitat types are sampled. Ideally, there will be at least 3 segments in each major habitat type (segment size can be shortened to accomplish this). To ensure that segments are distributed over the entire site, it may be useful to stratify sampling according to aspect or habitat type.

HABITAT-BASED PROTOCOL

The trapping technique described by Shaffer et al. (1994) is not always suited for sampling in our area and other protocols are needed. This is particularly true for lowland marshes that often are characterized by highly irregular shorelines and broad areas of shallow water with emergent vegetation and no perceivable depth gradient. Their technique has the advantage of an easily followed randomization scheme but, our experience suggests that it leads to an inefficient allocation of trapping effort in many

Pacific Northwest wetlands. If wetlands with complex shorelines and habitats will be encountered, it may be best to develop a trap-placement protocol based on the habitat structure of each site.

The first step in a habitat-based protocol is to identify the major habitat units at each site. Trapping will occur in each habitat unit. Wetland classes described by Cowardin et al. (1979) can serve as a guide, but are not essential for this technique. Habitat units should include, but may not be limited to, all of the main vegetation associations. These will often correspond to physical features such as aspect, substrate and water depth, that also are important to amphibians. However, if you encounter a wetland that appears homogenous, we suggest at least dividing it into north and south habitat units. Usually there will be several large patches of differing vegetation. Each of these patches (habitat units) should be sampled. A large wetland (>1 ha) should be divided into at least four habitat units (north, south, east, west), even if it lacks noticeable changes in vegetative characteristics. Habitats deeper than 1 m need not be sampled.

Habitat units may occur as large patches or as dispersed mosaics of microhabitats (e.g., small clumps of palustrine broad-leaved scrub-shrub habitat dispersed throughout a wetland but embedded in a matrix of other habitat types). Another example is when large trees occasionally shade small portions of a marsh. While they never occur as a large aggregated unit, they may be an important habitat unit if they occur regularly. There is a danger of overlooking such features when subjectively identifying habitat types. Be alert to this weakness. Randomly placing traps within defined habitats helps minimize this problem.

The number of traps needed will depend on the number and size of the habitat units, the type of trap used, and the level of precision required. No known technique can guarantee 100% detection and at some point the payoff for increased effort cannot justify the expense. We attempt to provide guidelines based on our collective experience with funnel trapping in the Pacific Northwest that will be likely to detect healthy populations of amphibians. The actual probability of detecting a population will depend on its size and habitat (see also Bury and Major, this vol.). We emphasize that the numbers given below are simply our opinion and await verification. Moreover, populations may experience extreme fluctuations in numbers over time (Pechmann et al. 1991) and species could be missed during off-years. When in doubt, use more traps.

If minnow or collapsible traps are used, we recommend two traps for a 25 m^2 habitat unit and then adding one trap each time the area of the habitat unit doubles. A 50 m^2 habitat unit should receive three minnow-traps, a 100 m^2 patch four, and so on. Thus, a ca. 0.5 ha. habitat unit should receive 9-10 traps. If pop-bottle traps are used, we suggest at least three times as many. If a total of ten traps are needed for a habitat unit, they can all be set out at once or some fraction can be set out over successive nights so that a total of ten trap-nights is achieved. However, trap-nights should not be split over the three different trapping intervals we recommended. Our experience suggests that these numbers are moderate to high. They should be used when it is important that failure to detect a species is rare. If only one (or a few) very important sites will be surveyed, more traps are recommended.

For a basic inventory, it is adequate to spread the traps around each habitat type haphazardly. Approximately one third of the traps should be placed along or near habitat edges. Ideally trap-placement should be randomized within habitat types in monitoring programs. This will help prevent systematic bias caused by some workers placing traps in better spots than others. The simplest way of randomizing placement is probably to sketch each habitat type and overlay it with a rough grid of approximately $25m^2$ cells. This doesn't need to be very accurate. Number each grid-cell and then randomly choose the cells where the traps will be placed. After walking to the approximate location of a $25\text{-}m^2$ cell, the exact placement (to the nearest $1m^2$) should be randomized by choosing two random numbers from 1-5 to indicate the coordinates where the trap should be placed within the cell. It is helpful to generate a list of random coordinates in advance.

This randomization scheme is somewhat cumbersome but assures maximum statistical defensibility. In practice, spreading traps haphazardly over well-defined habitat types may be as effective or preferable since it assures good coverage of the habitat type and its edges. If habitat types are difficult to define it may be more important to randomize so that some habitats are not systematically overlooked. In such situations, it also will be preferable to use more traps than is recommended above since there is habitat heterogeneity over which there is no control.

Conclusion

Funnel trapping is an effective technique for conducting amphibian inventories, but issues related to the number and allocation of traps need study. Funnel traps can also be used for monitoring detected/not detected trends, but relative abundance is difficult to monitor using any technique and the simplicity of obtaining such measures (e.g., trap rate) is deceptive.

The inherent variability of amphibian populations greatly reduces power to detect trends in relative abundance. Moreover, the relationship between an index of abundance and true abundance is likely to change over time and under different sampling conditions. For example, the invasion of a predator may lower the detectability of individual amphibians if the amphibians alter their behavior to avoid predation. Also, the effect of a predator entering a trap on capture probability is unknown. Since larvae often respond to the threat of predation by reducing activity or changing microhabitat (Lawler 1989; Skelly and Werner 1990; Feminella and Hawkins 1994; Manteifel 1995), capture probability may be lower when a predator is feeding in a trap.

A further consideration is that population regulation may occur in the larval and/or adult stages of pond-breeding amphibians (Wilbur 1980). If in the larval stage, monitoring relative abundance of larval populations may be informative. However, regulation in the adult stage may cause trends in breeding populations to go unnoticed by larval monitoring programs. Further study is required before funnel traps can be used to monitor relative abundance trends.

(Additional Literature Cited in this revision)

Feminella JW, and Hawkins CP. 1994. Tailed frog tadpoles differentially alter their feeding behavior in response to non-visual cues from four predators. J. N. Am. Benthol. Soc. 13(2):310-320.

Lawler SP. 1989. Behavioural responses to predators and predation risk in four speices of larval anurans. Anim. Behav. 38:1039-1047.

Manteifel Y. 1995. Chemically-mediated avoidance of predators by Rana temporariai tadpoles. J. Herpetol. 29(3):461-463.

Shaffer, H. B., R. A. Alford, B. D. Woodward, S. J. Richards, R. G. Altig, and C. Gascon. 1994. Quantitative sampling of amphibian larvae. Pp 130-141 In: W. R. Hyer, M. A. Donnelly, R. W. McDiarmid, L. C. Hayek, and M. S. Foster, eds. Measuring and Monitoring Biological Diversity: Standard Methods for Amphibians. Smithsonian Inst. Press, Washington, D. C.

Skelly DK, Werner EE. 1990. Behavioral and life-historical responses of larval American toads to an odonate predator. Ecology 71(6):2313-2322.

Wilbur, H. M. 1980. Complex Life Cycles. Ann. Rev. Ecol. Syst. 11:67-93.

Table 1. Tests of bait effectiveness in Washington. Differences between baited and control traps were tested with Mann-Whitney U tests. P-values indicate that only *Ambystoma tigrinum* was attracted to bait.

Test	Species	Stage	No. Caught Baited	No. Caught Control	p
*Pierce Co., March 1993. 17 baited with shrimp, 17 control minnow-traps.	*Rana catesbeiana*	tad	7	4	.780
	Ambystoma gracile	larv	2	6	.251
*Thurston Co., April 1993. 8 baited with bread, 8 control minnow-traps.	*A. gracile*	larv	2	2	1.0
	Taricha granulosa	adult	40	36	.911
Pierce Co., Jan/Feb 1995. 25 baited with fresh salmon eggs, 25 control pop-bottle traps.	*A. macrodactylum*	adult	204	272	.232
Pierce Co., May 1995. 20 baited with fresh salmon eggs, 20 control pop-bottle traps.	*A. macrodactylum*	adult	12	12	.936
	Hyla regilla	tad	106	68	.828
Lincoln Co., June 1995. 4 baited with fresh salmon eggs, 5 control pop-bottle traps.	*A. tigrinum*	larv	7	0	.007
Pierce Co., Aug 1996. 5 baited with fresh salmon eggs, 9 control. Minnow and collapsible traps.	*A. gracile*	larv	1	4	.378
	T. granulosa	adult	1	2	.925
Pierce Co. Oct 1996. 11 baited with fresh salmon eggs, 11 control. Minnow and collapsible traps.	*A. gracile*	larv	3	1	.28
	T. granulosa	adult	15	100	.114

*Data courtesy of R. B. Bury.