Chapter 7
Wetland Wildlife Monitoring and Assessment

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Abstract Monitoring wetland wildlife is complex and requires use of various techniques to obtain robust population estimates. Herpetofauna, birds and mammals frequently inhabit wetlands and adjacent uplands. Sampling herpetofauna can include passive techniques such as visual encounter and breeding call surveys, and capture techniques that use nets and traps. Common bird monitoring techniques include scan surveys, point counts, nest searches, and aerial surveys. Some mammals, such as bats, can be sampled with audio devices, whereas mark-recapture techniques are most effective for other taxa. For all groups, the techniques used depend on the monitoring objective and target species. This chapter describes various techniques for monitoring populations of wetland wildlife. If these techniques are incorporated into a robust sampling design, they can be used to document changes in species occurrence, relative abundance, and survival.

7.1 Introduction

Wetland wildlife (e.g., freshwater turtles, amphibians) are some of the most imperiled taxa in the world. Many species that use wetlands (e.g., waterbirds) have great economic and recreational importance. Thus, monitoring wildlife populations in wetlands is a fundamental component of management and conservation. Monitoring data can be used to document species distribution, estimate relative abundance, and track population change over time. Monitoring data also are useful in evaluating wildlife responses to management and conservation strategies. If monitoring data are collected using an

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unbiased sampling design, they can be used for making science-based, adaptive management decisions.

Monitoring wetland wildlife requires a combination of techniques to sample effectively the species that are present. Four wildlife groups that commonly use wetlands for portions of their life cycle include amphibians, reptiles, birds, and mammals. This chapter outlines standard procedures for sampling these animals, which includes passive methods, such as visual or auditory surveys, and techniques where animals are captured. Below, we describe some considerations for monitoring wildlife in wetlands.

7.2 Monitoring Considerations

The first step in selecting a monitoring technique is to identify the management, conservation, or research question (Witmer 2005). If determining species presence is the goal, techniques that target animal detection (e.g., call surveys, animal tracks) can be used. Techniques that produce count data (e.g., visual encounter surveys, scan sampling) can be used if estimating relative abundance is an objective. Lastly, if the goal is to develop a model that predicts population trends, a combination of techniques will be needed that estimate survival, reproduction, and relative abundance (e.g., mark-recapture). Sampling duration and costs typically increase from determining species presence to developing a predictive population model. Therefore, matching sampling techniques with the monitoring objective is key to ensuring appropriate data are collected considering the available resources (Witmer 2005).

Regardless of the technique used, rarely can 100 % detection of individuals present be ensured when sampling wetlands. Wetlands contain dense vegetation that aids in concealment, and many wetland wildlife are secretive or cryptically colored which reduces detectability. Detection also can be affected by observer experience, weather conditions, and time of day or year. Given that species detection is imperfect within a wetland, occupancy or relative abundance estimates can be biased depending on the sampling conditions or target species. Analytical techniques are available to correct for imperfect detection (see MacKenzie et al. 2006), and typically involve repeated sampling in a designated area. For very rare species, detection can be near zero, thus alternative sampling approaches (e.g., adaptive cluster sampling, Thompson and Seber 1996) may be needed. Sampling intensity and duration also should be considered, and correspond with monitoring objectives and animal life history. For example, breeding bird surveys should occur during late spring and early summer, whereas sampling for resident wildlife (e.g., rodents) could occur throughout the year. The goal of this chapter is not to discuss possible study designs (e.g., random, stratified, systematic, cluster) or analytical methods (e.g., occupancy modeling, calculating detection probabilities, Jolly-Seber population estimation) associated with monitoring wildlife populations, but to present techniques used to sample wetland wildlife. We recommend readers consult a statistician or biometrician to assist with designing an unbiased sampling approach and analyzing monitoring data. Chapter 1 (Vol. 1) covered several of these basic sampling principles, and various classic texts exist for guidance (Williams et al. 2002; Montgomery 2005; McComb et al. 2010; Zar 2010). Additionally, we define statistical terms and concepts that are used in this chapter (Box 7.1).

Box 7.1

The accuracy of a given population estimator can be defined as how close the actual estimate is to the true population value. The precision of the estimator is defined as how much variability there is in the estimate, based on repeated sampling. Ideally, a monitoring method should produce accurate and precise estimates. The accuracy of a given method can vary based on the species being monitored and the local setting under which monitoring is occurring. The precision of the method is a function of the inherent variance associated with the technique used and sample size. Consideration also must be given to the area and time period being sampled to ensure that sampling is representative of the population parameter being estimated. A census technique is defined as any method in which the goal is to count all individuals in the population. An index of relative abundance is any method in which a parameter estimate is counted that is related to total population size. All population estimation techniques can be affected by bias, a measure of the difference between the expected value of a given population parameter and the true value. Bias can result from many potential sources, including effects related to the behavior of the species being targeted and the ability and experience of the observer. For most methods, there are three sources of variability for population estimates: spatial variability, temporal variability, and detectability. Spatial variability results because not all sites occupied by a given species have the same size, habitat configuration, and density. A sampling framework is needed (e.g., stratified approach), as a result, to ensure that the samples are representative of the areas being occupied by the target species. Temporal variability results because populations are dynamic and change by time of day, stage of the life cycle, and year. Sampling needs to account for the sources of temporal variability. Detectability can be defined as the probability a given individual will be detected by the observer given that it is present to be detected. Detectability can vary based on species, observer, weather, time of day, season, and habitat conditions. Wherever possible, estimates of detectability should be used to adjust indices of relative abundance to reduce bias.

Additional considerations during sampling include animal welfare and personal safety. In most cases, wildlife collection permits need to be acquired prior to sampling if animal capture is part of the study design. Further, capture, handling, and marking techniques may require approval by an institutional animal care and use committee. Some techniques described below (e.g., capturing snakes and bats) can be dangerous and require training by an expert or immunizations. We recommend that novice biologists consult experts for initial hands-on training if dangerous wildlife will be handled. Many wetland wildlife species harbor zoonotic pathogens so standard biosafety precautions, such as wearing disposable gloves and disinfecting equipment, should be practiced. Biologists and researchers can contribute to pathogen pollution (i.e., introduction of novel pathogens into a population, Cunningham et al. 2003) while sampling wetlands by unintentionally translocating pathogens among populations on fomites. Thus, all sampling gear and footwear should be disinfected before moving among wetlands. A solution of 10 % bleach or 2 % Nolvasan® (chlorhexidine diacetate) with a contact time of 10 min will inactivate most pathogens.

7.3 Monitoring Herpetofaunal Populations

Herpetofauna include animals in the Classes *Amphibia* and *Reptilia*. Various species of amphibians and reptiles use wetlands and their adjacent terrestrial habitats. To obtain the most representative estimates of species occurrence, relative abundance, or other demographic indices, a combination of aquatic and terrestrial sampling techniques are typically required. Below, we summarize aquatic and terrestrial sampling techniques for amphibians and reptiles; some methods (e.g., funnel and pitfall traps) can be used for both groups. We also summarize techniques for marking herpetofauna for mark-recapture studies.

7.3.1 Amphibians

Most amphibian species in the temperate regions of the world have a complex life cycle that involves development in aquatic systems as larvae and in terrestrial systems as juveniles and adults (Wells 2007). Thus, sampling amphibian populations associated with wetlands typically involves a combination of techniques that target both ecosystems (Dodd 2009). Information collected in the wetland zone of aquatic systems typically addresses questions related to reproductive effort, larval production, and possible recruitment; whereas, data collected in the terrestrial environment provide information on survival and recruitment of juveniles, adult survival and population size, and dispersal. Techniques involve passive counts, capture methods, and marking individuals for survival and dispersal estimates. Below are some approaches for sampling amphibians in aquatic and terrestrial systems; we encourage readers to consult Dodd (2009) for additional details.

Fig. 7.1 Amphibian sampling techniques in the aquatic environment. Egg mass surveys for anurans (**a**) and ambystomatid salamanders (**b**), dip-net sampling (**c**), frog-call recorders (**d**), seine sampling (**e**), larval enclosure sampling (**f**), and aquatic minnow traps (**g**) (Published with kind permission of © Matthew Gray, William Sutton, and David Steen 2014. All Rights Reserved)

7.3.1.1 Aquatic Sampling

Many amphibian species oviposit floating egg masses or attach eggs to emergent or submersed vegetation in wetlands (Fig. 7.1; Wells 2007). Egg mass counts can be used as an index of adult population size and reproductive effort (Paton and Harris 2010). For some amphibian species (e.g., *Ambystoma maculatum*, Petranka 1998:80) females can lay multiple clutches, thus egg mass counts need to be adjusted if inferences are made on per capita reproduction or adult population size. Wells (2007:501) summarized information on average number of egg masses and clutch size for several anuran species, while Petranka (1998) provides information on oviposition strategies of salamanders. Egg mass identification can be done reliably to genus; species-level identification requires more experience and typically knowledge of breeding species and their phenology at a site. Most amphibian identification guides (e.g., Dorcas and Gibbons 2008; Niemiller and Reynolds 2011) and websites include photos of egg masses. Most egg masses are deposited in the littoral zone of wetlands in water that is <60 cm (Wells 2007), thus sampling should focus in areas close to the water's edge. Some species prefer to lay egg masses amongst vegetation (e.g., *Pseudacris* spp.), whereas other species prefer more open water (e.g., *Lithobates catesbeianus*). Counts should be performed at least twice per year (i.e., spring and summer) to incorporate breeding phenology (Paton and Harris 2010). Amphibian movements and breeding tend to be associated with precipitation (Wells 2007), hence targeting sampling within 48 h of a rain event likely increases egg mass detection. If a site is sampled multiple times, egg mass counts can be adjusted for changes in detection associated with rainfall. Counts are typically performed along a transect or within a designated area, and search time and number of observers is recorded to standardize relative abundance estimates (Paton and Harris 2010). To estimate relative abundance per species, divide number of egg masses counted per species by the collective minutes searched for all observers then divide this quotient by the number of observers. This estimate can be compared among years and sites if egg mass detectability is similar.

Amphibian larvae in the temperate regions include frog tadpoles and salamander larvae. Tadpoles of various species can be found in wetlands; the most commonly encountered salamanders in North American wetlands belong to the Ambystomatidae and Salamandridae families (Wells 2007). The most common capture techniques include nets, traps and enclosure sampling (Skelly and Richardson 2009). Schmutzer et al. (2008) used a combination of seine nets in deeper water and dip nets in shallow water to sample larval amphibians. Typically, seine nets (0.48-cm mesh) are pulled over a specified distance for relative density estimates, and are most effective if emergent vegetation is absent (Fig. 7.1). Dip netting can be done at sampling points along transects that traverse the elevational gradient of the wetland (Schmutzer et al. 2008) or in random locations within the emergent vegetation zone (Fig. 7.1). For truly random sampling, a 1-m^2 grid should be overlaid on a geo-referenced image of the wetland in a GIS and cells randomly generated for sampling locations. Similar to egg masses, sampling can be standardized by recording the number of dips taken over a specified duration and the number of individuals that participate (Skelly and Richardson 2009). There are a variety of dip nets that can be used; however, we recommend one with a large opening (e.g., 40×40 cm) and deep net (>50 cm) with fine mesh (<0.25 cm). Dip nets should be plunged down into the water including the leaf litter and quickly scooped upward. Alternatively, nets can be dragged through the water for a specified distance. Net contents, which may include litter and substrate, should be carefully sorted to detect larvae. Dip nets can be destructive to habitat and cause injury to larvae if the net frame strikes them; thus, dipnetting may not be ideal for threatened species or frequent, long-term monitoring.

The most common type of trap used to capture amphibian larvae is a minnow trap, which contains two opposing funnels that taper inward (Fig. 7.1; Skelly and Richardson 2009). Larvae are naturally directed into the tunnel, and after passing through a small opening are unable to find the opening again. Minnow traps should be placed in shallow water with at least 10 % of it exposed to provide air if adult salamanders are captured and should be checked every 12–24 h (Skelly and

Richardson 2009). If minnow traps are placed in deepwater zones of a wetland, they should be tethered to a permanent structure (e.g., tree or stake at the edge of the wetland) to prevent the traps from sinking or floating away, and to facilitate relocation. Drechsler et al. (2010) describe the design for a modified funnel trap that has greater capture efficiency than traditional traps.

Enclosure samplers are either rectangular (box-type) or circular, and are designed to enclose a designated area for sampling (Mullins et al. 2004; Skelly and Richardson 2009). A very simple, circular enclosure can be created by cutting the bottom off of a 120-L garbage can (Fig. 7.1). Enclosures are placed into water with about 5 cm of the bottom sunk into the substrate and the contents netted. Nets should be small (20×13 cm) with fine-mesh and a sturdy handle; most aquarium nets are too flimsy. Nets are repeatedly dipped through the entire water column and surface area for a minimum of ten times (Werner et al. 2007). Dipping should cease after ten consecutive dips result in no captures (Werner et al. 2007). Similar to the other methods, enclosures can be randomly or systematically placed in the wetland. For all procedures, captured larvae can be placed in a holding container until they can be identified and enumerated. Dr. Ronn Altig has written several keys for the tadpoles of North America (Altig 1970, 1987), and collaborated in developing a U.S. Geological Survey website (http://www.pwrc.usgs.gov/tadpole/). Tadpole identification can be difficult and requires knowledge of unique combinations of the vent, spiracle, and eye positions on the body, oral disc morphology, and dentition. Petranka (1998) provides descriptions of most salamander larvae in North America.

7.3.1.2 Terrestrial Sampling

Postmetamorphic amphibians can use terrestrial habitat within a considerable distance from a breeding wetland. In a review by Semlitsch and Bodie (2003) of the core terrestrial habitat for 32 North American amphibians, they reported that amphibians used habitat within 159–290 m of their breeding site. Smith and Green (2005) also reported that 40 of 90 (44 %) amphibian species reviewed moved <400 m, with salamanders being less mobile in general compared to anurans. Most amphibians acquire food resources necessary for growth and survival, estivate and hibernate, and disperse between wetlands in the uplands (Wells 2007). Thus, sampling terrestrial systems around wetlands for amphibians is a fundamental component of population monitoring. As with larvae, it is recommended that multiple sampling methods are used to increase the likelihood of detecting all amphibian species (Ribeiro-Junior et al. 2008; Farmer et al. 2009).

One of the most common techniques used to document anuran species occurrence is advertisement call surveys. Advertisement calls are produced by adult males of most frog and toad species during breeding to attract females (Wells 2007). Anuran calls are unique among species, and most species can be reliably identified with practice. Several CDs are available with anuran calls from North America (e.g., Elliot et al. 2009). Calls can be recorded by observers or automated recording devices that are deployed overnight (Fig. 7.1; Dorcas et al. 2010). Procedures for performing call surveys vary, but the most widely used approach in North America follows protocols outlined by the North American Amphibian Monitoring Program (NAAMP, http://www.pwrc.usgs.gov/naamp/). The NAAMP is composed of routes randomly located throughout North America where volunteers listen for breeding frogs at ten stations per route. The NAAMP protocol specifies that call surveys are performed between 30 min following official sunset and 0100 h. Surveys are performed for 5 min only, and during that time, all frog species heard are recorded along with an index of relative abundance (Burton et al. 2007). Several studies suggest that 5 min is adequate to detect most breeding anurans (Shirose et al. 1997; Gooch et al. 2006; Burton et al. 2007). A call index = 1 when calls from different males do not overlap, = 2 when calls overlap but individual males can be distinguished, and = 3 when calls overlap and individual males are indistinguishable (Burton et al. 2007).

Call surveys are inherently biased for most anuran communities if detection is not corrected, because acoustical properties, including sound power and call frequency, differ among species (Dorcas et al. 2010). Additionally, ambient conditions can impact detection positively or negatively (e.g., during rain events or windy nights, respectively; Dorcas et al. 2010). Observers also differ in their ability to detect species and record similar abundance (Burton et al. 2007). Thus, most experts recommend that call surveys should be used to document species occurrence only (Dorcas et al. 2010). Surveys should be performed at least once monthly from early spring through summer to encompass most of the anuran breeding season (Wells 2007). Performing surveys within 48 h following a rain event may increase the likelihood of species detection, because call frequency is correlated with precipitation in many species (Wells 2007).

More detailed information on adult population size and processes (e.g., survival, dispersal) can be measured using capture-recapture techniques. The most common capture techniques used in the terrestrial environment include: drift fences with pitfalls, artificial cover objects, funnel traps, and visual encounter surveys (Willson and Gibbons 2010). Drift fences can completely or partially enclose a wetland, or be constructed as single segments or an array (Fig. 7.2; Willson and Gibbons 2010). Drift fences can be made of various materials, but plastic erosion fencing with 60-cm wooden stakes tends to be least expensive and is easy to erect. Although more expensive than erosion fencing, metal flashing or hardware cloth is more durable – usually, erosion fencing is usable for 1–2 years only. The bottom of the fence should be buried to prevent amphibians from crawling underneath; soil from holes dug for pitfalls (discussed below) can be used to bury the fence. If the goal is to estimate adult breeding population size or number of metamorphosed juveniles produced, drift fences should be placed near the wetland. Gray et al. (2004) standardized drift fence placement at 10 m above the expected high waterline and parallel to the wetland. Single drift fence segments or arrays can be erected between wetlands or along terrestrial contours to identify movement corridors and estimate dispersal rates.

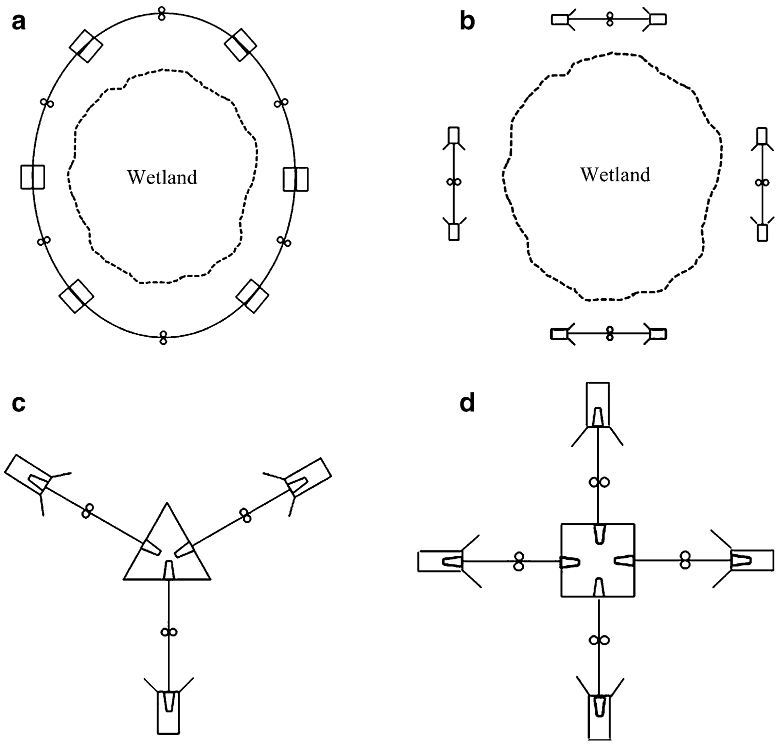

Fig. 7.2 Drift-fence arrays for sampling herpetofauna in the terrestrial environment. Wetlands fully (**a**) and partially (**b**) encircled with drift fence and pitfalls. Three- (**c**) and four-fence (**d**) arrays, which include a large box trap at the center (Figure 7.2c published from Sutton et al. (2010) with kind permission of Current Zoology 2010. Figure 7.2d modified from Burgdorf et al. (2005) and published with kind permission of the Society for the Study of Amphibians and Reptiles 2005

Pitfalls are placed adjacent to the drift fence typically every 10 m and at the ends of fence sections to capture amphibians that intercept the fence (Fig. 7.3; Gray et al. 2004). Standard placement is two opposing pitfalls; one on each side of the fence (Willson and Gibbons 2010). However, digging holes near each other for pitfalls can result in the dirt collapsing between them. Thus, an alternative design is to place one pitfall on alternating sides of the fence every 5 m (Burton et al. 2009), which results in the same pitfall density. Pitfalls can be made of various materials but large (19-L) plastic buckets tend to capture the greatest number of species (Willson and Gibbons 2010). Pitfall captures for most amphibian communities will be biased, as many tree frog (Hylidae) species can climb out and large ranid frogs (e.g., *Lithobates catesbeianus*) can jump out (Willson and Gibbons 2010). Usually, 1-cm holes are drilled in the bottom of pitfalls to allow water to drain during rain events, and a moist sponge is placed in the pitfall to prevent desiccation of captured amphibians. However, in arid or hot regions, a sponge may be insufficient to keep amphibians moist, thus some water (e.g., 5 cm) can be put in pitfalls to reduce desiccation. If water is

Fig. 7.3 Pitfall traps (19-L plastic buckets) used to catch amphibians and reptiles. Pitfall traps must be installed flush with the ground and as close to the drift-fence to be as effective as possible (Published with kind permission of © Matthew Gray and William Sutton 2014. All Rights Reserved)

added, the sponge should remain in the pitfall to allow small mammals captured incidentally to climb out of the water. When pitfalls are not covered, they should be checked at least every 24 h, and we recommend they are opened at least 2 days per week. Typically, captured amphibians are measured, marked uniquely, and released on the opposite side of the fence that they were captured. Covering pitfalls with bucket lids for 24 h between capture events can prevent immediate recapture and biases in population estimates. Drift fences that completely encircle wetlands should be checked daily or partitions removed to allow unrestricted movement when sampling is not occurring. Similar to other techniques, opening pitfalls during rain events can facilitate amphibian captures due to greater movement.

Artificial cover objects can be used as a technique to supplement species detection; however in general, this method does not provide good estimates of population size because recapture rates tend to be low (Fig. 7.4; Bailey et al. 2004a, b). Given that amphibians desiccate easily (Wells 2007), cover objects can provide moist microhabitat during the day. Cover objects (e.g., 120 × 120 cm or smaller) are usually made of untreated plywood or corrugated tin (Willson and Gibbons 2010). The odds of catching amphibians are often greater under wood objects, whereas reptile captures tend to be greater under tin (Grant et al. 1992; Hampton 2007). Objects should be deployed for at least 1 month prior to sampling so that suitable microclimate conditions develop under the object. Funnel traps (discussed in the following section) are often used in combination with drift fences and cover objects to capture amphibians and reptiles (Willson and Gibbons 2010).

Amphibians are often found amongst leaf litter and under natural cover objects (e.g., logs, stones) in the terrestrial environment (Wells 2007). Thus, searching for amphibians under natural cover objects has become a standardized sampling method. Visual encounter surveys can be time- or area-based. For both survey types, natural cover objects are searched for amphibians. Searching during nights with rain can increase the likelihood of detecting individuals. Crump and Scott (1994) describe three levels of search intensity: Level 1 = counts of amphibians on the surface only, level 2 = level 1 and amphibians detected under natural cover objects, and level 3 = previous levels and intense searches through leaf litter and the interior of decaying logs. Level 2 is most commonly used because level 3 destroys

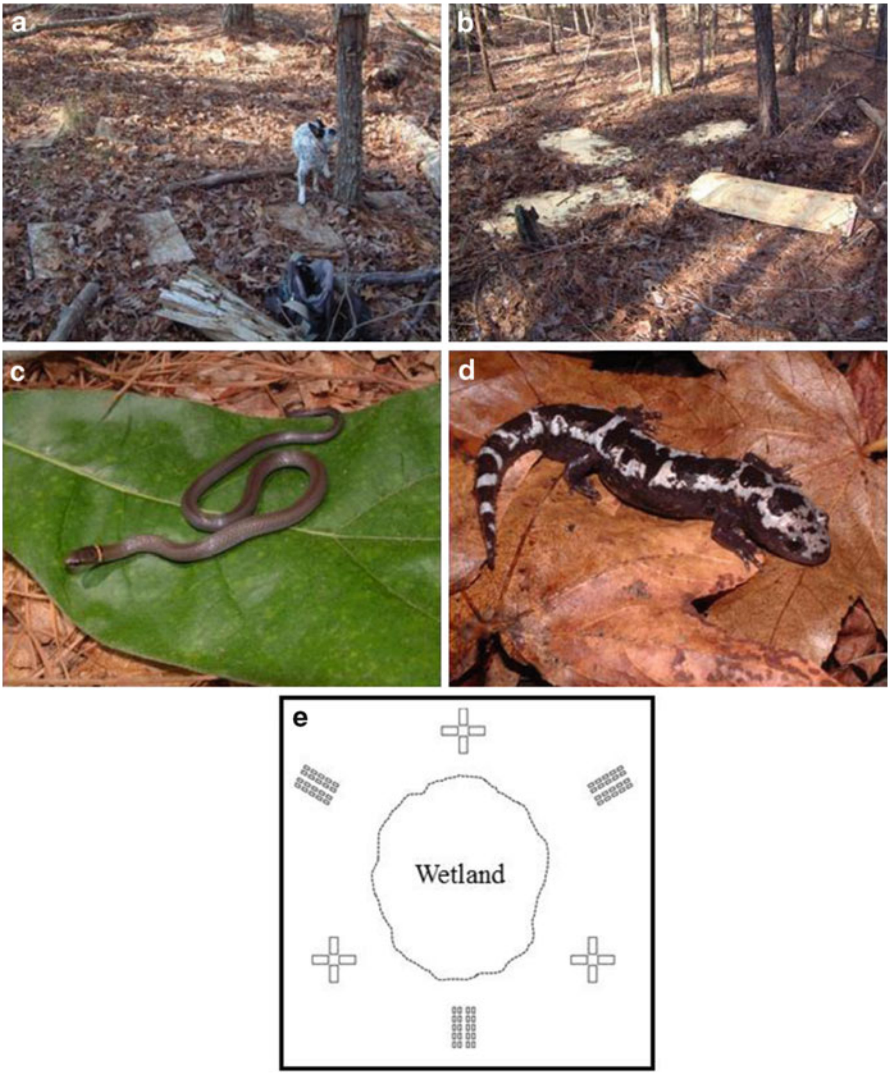

Fig. 7.4 Cover objects such a small (**a**) and large (**b**) plywood boards can placed directly on the ground to survey amphibians and reptiles. Fossorial species such as ring-necked snakes (*Diadophis punctatus*; **c**) and marbled salamanders (*Ambystoma opacum*; **d**) can be sampled using this technique. A variety of coverboard sizes can be used to obtain a more complete sample of the herpetofaunal community and can be distributed at increasing distances from the wetland area (**e**) (Published with kind permission of © William Sutton 2014. All Rights Reserved)

amphibian habitat, and detection is low for level 1 except during rain events. Similar to egg and larval sampling, time-based surveys involve searching for a designated duration (e.g., 30–60 min) and adjusting the number of detected amphibians for the amount of time searched and number of searchers used.

Area-based searches can be in plots or belt transects. Plots are typically 10 × 10 m or 25 × 25 m, with larger plot sizes used when amphibian densities are low (Crump and Scott 1994). The most common transect dimensions are 50 or 100 m in length and 1 or 2 m in width (Marsh and Haywood 2009). Plots or transects can be randomly or systematically placed in a sampling area; adaptive cluster sampling is recommended for species that are uncommon or have a clustered distribution (Marsh and Haywood 2009).

7.3.2 Reptiles

Worldwide, reptiles are found in a wide variety of habitats. Specifically, wetlands provide habitat for many reptile species, with some species endemic to these ecosystems. Reptiles are an important component of wetland ecosystems, as their species diversity is equal to or higher than amphibians in some regions of the United States (Russell et al. 2002). Most reptiles associated with wetlands use both terrestrial and aquatic habitats. Therefore, it is necessary to use multiple sampling techniques to monitor reptile communities effectively. In this section, we discuss standardized reptile sampling techniques including drift-fences, artificial cover objects, aquatic trapping techniques, and visual encounter surveys.

7.3.2.1 Aquatic Sampling

Similar to amphibians, visual encounter surveys can be used to sample aquatic reptiles in wetland ecosystems. Visual encounter surveys are commonly used to detect basking turtles (e.g., map turtles [*Graptemys geographica*]), semi-aquatic snakes (e.g., northern water snakes [*Nerodia sipedon*]), and crocodilians (Fig. 7.5). Sampling should be performed at a distance (20 m or greater) to prevent disturbance. We recommend establishing viewing stations systematically around a wetland. Blinds can be erected to aid with entry and departure without disturbing basking individuals. Kayaks or similar watercrafts can be used to quietly traverse transects on larger bodies of water or rivers. Binoculars or a spotting scope should be used to aid in detection and identification of species and individuals. Basking reptiles are frequently detected on emergent structures (e.g., logs and stumps) and along the banks of wetlands or rivers that are devoid of vegetation. We recommend performing basking surveys for turtles and snakes late morning through mid-day (10:00–15:00 h when water temperatures are between 15 and 25 °C (Coleman and Gutberlet 2008). Nocturnal surveys from watercrafts with a bright spotlight (≥200,000 candlepower) are often performed for detecting crocodilians (Fujisaki et al. 2011). Visual encounter surveys can be used to estimate species occurrence or relative abundance. Mark-resight techniques with highly visible marks (discussed later) are generally necessary to estimate relative abundance. However, certain occupancy-based analyses permit simultaneous estimation of

Fig. 7.5 Common aquatic reptile sampling techniques. Visual encounter surveys can be used to spot basking turtles (**a**) and semi-aquatic snakes (**b**). Baited hoop traps are commonly used to sample aquatic turtles (**c**), whereas crayfish traps (**d**) permit sampling in deeper water and may increase detectability of secretive semi-aquatic snakes such as *Farancia* spp. (Published with kind permission of © Sean Sterrett, William Sutton, and David Steen 2014. All Rights Reserved)

occupancy, abundance, and detection probability based on presence-absence or count data (Fujisaki et al. 2011; Royle and Nichols 2003; Royle et al. 2005).

Baited hoop traps and basking traps are the primary method used to sample aquatic turtles. Hoop traps are a series of large hoops wrapped with netting material with an elliptical funnel-like entrance on one side of the trap (Fig. 7.5). Hoop traps are typically baited with canned fish to attract turtles. To prevent captured turtles from drowning, traps should be placed in shallow water with a portion of the top exposed. If sampling in deeper water is necessary, one or more 2-L plastic bottles should be placed inside the trap as buoys to create a breathing space, or traps can be tethered to a tree on the edge of the river or wetland. Turtles are known to escape from hoop traps, thus they should be checked a minimum of two times per day to reduce trap escapes (Frazer et al. 1990; Brown et al. 2011). Hoop traps can be modified to include fyke nets or leads that function as an underwater drift-fence to direct turtles into a central trap (Vogt 1980; Glorioso et al. 2010). We recommend using fyke traps in isolated portions of a wetland, such as a narrow cove to intercept swimming turtles (Vogt 1980). Lastly, a basking trap is a square floating structure with an open center and a net or wire basket attached underneath (Brown and Hecnar 2005; Gamble 2006). Turtles bask along the frame of the trap and are captured when they fall into the center, which typically occurs as the trap is approached by the researcher. Basking traps should be placed in areas where turtles are likely to bask, such as shallow wetlands with abundant woody structure.

To increase the likelihood of species detection, we recommend using at least two trap types and placing them in a variety of depths and wetland types (Glorioso et al. 2010). Regardless of trapping method, all traps must be checked daily to minimize capture mortality. Trapping effort can be calculated by multiplying the number of traps set by the total number of sampling events. To standardize capture success at a particular site, divide the total captures by the overall sampling effort.

Aquatic funnel traps (i.e., minnow traps), as used for sampling larval amphibians, are an effective method to sample aquatic and semi-aquatic snakes (Willson et al. 2008). We recommend using different aquatic minnow traps with varying funnel sizes and mesh openings to increase the number of snake species and size classes captured (Willson et al. 2008). Using commercial crayfish traps or modified trash can funnel traps will permit sampling at greater water depths and may target larger species missed with traditional funnel traps (Fig. 7.5; Johnson and Barichivich 2004; Luhring and Jennison 2008). Funnel traps can be placed singly throughout a wetland or as part of an aquatic drift-fence array. If drift-fences are used in tandem with aquatic funnel traps, we recommend the rectangular style minnow traps because they fit flush against the side of the fence, which will increase capture probability (Willson and Dorcas 2004). To reduce the number of escapes and trap-induced mortality of turtles, all traps should be checked daily (Willson et al. 2005, 2008).

7.3.2.2 Terrestrial Sampling

Terrestrial visual encounter surveys for reptiles are commonly used to sample reptile populations. Techniques similar to those discussed in the amphibian section (e.g., searching forest litter, turning cover objects) are most effective for detecting reptiles. To make data comparable among wetlands, it is essential to implement a unbiased sample design and standardize effort among individuals involved in the survey.

Reptiles commonly seek shelter under cover objects, such as large logs, for protection from predators, thermoregulation sites, and nesting habitat. Researchers can exploit these behaviors by using artificial cover objects to sample reptiles (Fig. 7.4; Russell and Hanlin 1999). Many species of reptiles use corrugated tin at a greater frequency compared to wooden cover objects (Lamb et al. 1998), but certain species (e.g., litter dwelling snakes) will frequently use wooden cover objects (Felix et al. 2010). Black plastic sheeting also has been used to sample snakes and lizards (Adams et al. 1999; Kjoss and Litvaitis 2001). Because cover objects can be used to sample amphibians and reptiles, we recommend using a combination of wood and tin cover objects of various sizes (small [48 cm × 30.5 cm], medium [121.9 × 60.0 cm], and large [243.8 cm × 121.9 cm]) if sampling reptiles and amphibians is an objective. Placing cover objects at increasing distances from the edge of the wetland into the terrestrial environment will also permit sampling of both semi-aquatic and terrestrial reptile species. As with amphibians, cover objects should be deployed at least 1 month before sampling begins.

To avoid disturbing the microhabitat and negatively affecting occupancy rates, cover objects should be sampled only 1–2 times per week (Dodd 2003). Each cover object can be numbered to assess use patterns and trends (Fellers and Drost 1994). We recommend that researchers record environmental covariates (e.g., air temperature, relative humidity, and % cloud cover) to account for possible abiotic factors influencing detections (Joppa et al. 2009). Once individuals are captured, they should be processed according to study objectives (e.g., mass and body length measurements, genetic samples), and assigned an individual- or plot-specific mark to account for recaptures.

Certain reptile species may be difficult to detect due to large yearly dispersal patterns, cryptic coloration, or secretive life history patterns. Drift-fences with pitfall and funnel traps are commonly used to capture reptiles (Fig. 7.6). We recommend using aluminum flashing (60–90 cm in height) instead of silt fence to sample reptiles, because the metal surface deters climbing and trespass of individuals. Drift fences set for reptiles are often erected as X- or Y-shaped arrays with pitfalls and rectangular double-entrance funnel traps placed at various locations along the fence. Single-entrance funnel traps also can be placed at the terminus of each fence, with a 1-m section of fence angled at 45° from each trap corner to direct reptiles into the traps (Sutton et al. 2010). Arrays can include a large, central box trap for capturing larger snake species; schematics for these traps have been detailed elsewhere (Fig. 7.6; Burgdorf et al. 2005; Sutton et al. 2010). Small doors can be installed on the sides of box traps to assist with safe removal of venomous snakes (Steen et al. 2010). A water source should be added to traps to prevent dehydration of captured individuals. To increase capture efficiency, the drift-fence should be shaped to extend 15–20 cm into the funnel entrance and fit flush with the dimensions of the trap funnel. It is important that funnel and pitfall traps are installed flush along the vertical surface of the fence to prevent reptiles from circumventing the trap (Jenkins et al. 2003). Design and placement of drift fence arrays should be planned prior to study implementation and should correspond with study objectives (Fig. 7.2; Todd et al. 2007), as discussed in the amphibian section.

7.3.3 *Methods for Marking Herpetofauna*

Studies that seek estimates of population size, survival, or dispersal require the recognition of previously captured individuals (Williams et al. 2002). A myriad of

Fig. 7.6 Drift-fence arrays with large box traps used to sample the reptile community, especially large snakes such as timber rattlesnakes (*Crotalus horridus*) and black racers (*Coluber constrictor*) (Published with kind permission of © William Sutton and David Steen 2014. All Rights Reserved)

marking techniques have been developed for a variety of wildlife species (Silvy et al. 2005), with some more successful than others. For a marking technique to be effective and result in unbiased parameter estimates, it cannot affect survivorship or behavior of the individual and must provide a permanent and easily detectable mark (Ferner 2010). Additionally, application of the mark should not cause undue stress or pain. Typically, anesthesia is unnecessary but topical analgesics (e.g., Orajel®) can be applied to reduce pain. We recommend that researchers consult a wildlife veterinarian for correct dosage if an analgesic is used, because some analgesics contain chemicals (e.g., benzocaine), which can function as a euthanizing agent.

The methods below have been approved previously by U.S. Institutional Animal Use and Care Committees (Ferner 2010). The most common method for marking lizards, anurans, and salamanders is removing a toe(s) from the hind or front foot (i.e., toe-clipping) that corresponds to a pre-determined numerical scheme (Woodbury 1956). Sharp scissors that are disinfected in 2 % chlorhexidine diacetate or 95 % EtOH can be used to remove digits. Excisions should be made at the lowest joint to reduce bleeding and regeneration. For frogs with webbed feet, the webbing should be cut prior to excising the toe. Silver nitrate sticks can be used to stop bleeding and a topical antibiotic applied to the excision site to reduce risk of infection. Multiple pairs of scissors should be used because each pair should soak in disinfectant for at least 1 min between animals.

Marking schemes have been developed for amphibians (Donnelly et al. 1994; Ferner 2010) and lizards (Enge 1997), and most schemes can account for many unique individuals. For lizards, the longest (fourth) toe of the hind foot should not be clipped, and removal of >1 toe per foot should be avoided. In general, toe clipping arboreal frogs or lizards is not recommended (Ferner 2010). Thumbs on the front feet of male anurans should never be clipped due to their importance for amplexus (Ferner 2010). Toe-clipping has been shown to have both negligible (Paulissen and Meyer 2000; Dodd 1993) and negative impacts (Bloch and Irschick 2005; Schmidt and Schwarzkopf 2010) on climbing and running behaviors in lizards. Amphibian responses to toe-clipping are similarly disparate. For example, McCarthy et al. (2009) found that in salamanders, the likelihood of recapture decreased with the number of removed toes, whereas other research has found limited or no impacts of toe-clipping on normal amphibian behaviors (Ott and Scott 1999; Liner et al. 2007; Phillott et al. 2007). If toe-clipping is not an option, additional marking options include branding (Clark 1971; Ferner 2010), paint marking (Jones and Ferguson 1980; Simon and Bissinger 1983), and injectable colored elastomers (Schmidt and Schwarzkopf 2010).

Similar marking schemes exist for turtles and snakes; however, marks must be applied using different methods. Turtles can be individually marked by notching within the first and last four marginal scales on either side of the carapace using a sharp-edged metal file or rotary tool (Cagle 1939; Honegger 1979; Enge 1997). Tools should be disinfected between individuals. Other inexpensive marking options include branding the plastron (Clark 1971) or painting identifying features on the carapace using permanent paints. Snakes can be marked by using sterilized scissors or fingernail clippers to remove ventral (Brown and Parker 1976; Spellerberg 1977) or subcaudal (Blanchard and Finster 1933) scales, according to a pre-determined numbering pattern (Enge 1997). Care must be taken not to cut the scales too deeply, as infection may result (Honegger 1979). Other related marking methods include using medical cautery units (Winne et al. 2006) or colored injectable elastomers (Hutchens et al. 2008) to apply a semi-permanent mark.

A more expensive but highly effective method to individually mark herpetofauna is using passive integrative transponder (PIT) tags. A PIT tag is a small microchip encased in a glass container that transmits a signal, which is interpreted with an electronic reader as a unique serial number (Fig. 7.7; Gibbons and Andrews 2004). PIT tags can be implanted either subcutaneously or intraperitoneally in snakes (Keck 1994), in the abdominal skin midway between the limb and the plastron in turtles (Buhlmann and Tuberville 1998; Rowe and Kelly 2005), and in the abdominal cavity of anurans and salamanders (Ferner 2010). PIT tags are implanted using a large syringe, so care must be taken not to damage internal organs. To reduce the chance of infection, the PIT tag along with the syringe needle must be disinfected before injection into the organism. PIT tags appear to have few

Fig. 7.7 Subcutaneous injection of a Passive Integrative Transponder (PIT) tag into *Anaxyrus cognatus* (**a**), PIT tag scanner (**b**), and PIT tag under skin of *Spea multiplicata* (**c**) (Published with kind permission of © Matthew Gray and Sumio Okada 2014. All Rights Reserved)

negative impacts on survival and growth rates of turtles (Rowe and Kelly 2005) and snakes (Keck 1994; Jemison et al. 1995). PIT tags have been used to mark anurans and salamanders (Hamed et al. 2008; Ferner 2010); however, use of this technique should be limited to larger species. This technique results in a permanent mark that is easy to differentiate; however, newly marked individuals are at risk of losing tags through the PIT tag injection site. Vetbond® and other veterinary grade skin adhesives can be used to close the injection site (Ferner 2010). We recommend a secondary mark (e.g., scute/scale mark or toe clip) in addition to the PIT tag to ensure that recaptured individuals are not overlooked.

7.4 Monitoring Bird Populations

Monitoring bird populations in wetlands is challenging because many species are migratory, and use of a given wetland may vary during different seasons. For the purposes of this chapter, we divide the seasons into functional life-cycle stages (breeding, migration, and wintering), and discuss different methods for the following species groups: waterfowl, wading birds, shorebirds, secretive marsh birds, songbirds and raptors (Table 7.1). Species within these groups comprise most avifauna associated with wetlands in North America. For more detail on monitoring bird populations, please see Bibby et al. (2000) or Ralph et al. (1993).

7.4.1 Waterfowl

There are 70 species of waterfowl (Order Anseriformes: Ducks, Geese and Swans) in North America. Most of these species nest in Canada and the northern latitudes of the United States and migrate south during winter. Waterfowl are monitored and

Table 7.1 Avian monitoring methods by waterbird group

	Waterfowl			Shorebirds			Wading birds			Secretive marshbirds			Songbirds			Raptors		
Monitoring method	B[a]	M	W	B	M	W	B	M	W	B	M	W	B	M	W	B	M	W
Population status and trends																		
Aerial counts	x	x	x				x	x	x							x	x	x
Point counts													x		x	x		x
Mist-netting													x	x	x		x	
Transect counts	x			x			x			x			x		x	x		
Territory mapping													x					
Call-back surveys										x		x	x			x		
Migration counts		x			x			x									x	
Reproduction																		
Nest monitoring	x			x			x			x			x			x		
Brood counts	x																	
Survival																		
Band recoveries		x	x														x	
Radio telemetry	x	x	x	x	x	x	x	x	x	x			x			x	x	x
Activity																		
Radio telemetry	x	x	x	x	x	x	x	x	x	x			x		x	x	x	x
Other methods																		
Stable isotopes	x		x	x		x	x		x	x		x	x		x	x		x
Genetic markers	x		x	x		x	x		x	x		x	x		x	x		x

[a]Breeding, Migration, Winter

managed in North America under the guidance of the North American Waterfowl Management Plan (NAWMP). The NAWMP is a formal agreement among the United States, Canada and Mexico to set population and habitat goals for continental waterfowl populations (NAWMP Planning Committee 2004). A primary objective of NAWMP is to restore and maintain continental waterfowl populations at approximately 62 million breeding ducks (NAWMP Planning Committee 2004). Several monitoring programs exist to ensure accurate and precise estimation of waterfowl population sizes each year. By using estimates of breeding pairs, brood production, and overwinter survival, harvest regulations can be set to ensure that populations are maintained at desired levels. Below are some monitoring programs and techniques used to estimate waterfowl populations.

7.4.1.1 National Programs

Waterfowl populations are monitored during the breeding season and winter. The U.S. Fish and Wildlife Service and the Canadian Wildlife Service, along with state and provincial agencies, collaborate to implement the North American Waterfowl Breeding Population and Habitat Survey, which has been estimating duck and goose populations annually on the major breeding grounds in North America since 1955. This program relies on aerial surveys of over five million square kilometers of wetlands from fixed-wing aircraft and helicopters. Surveys are conducted in May and early June in the principal waterfowl breeding grounds of North America, including the north-central United States and Canada (i.e., the Prairie Pothole Region), Alaska, and the eastern United States and Canada. Ground surveys are used in combination with aerial surveys to adjust estimates for visibility bias. Surveys are flown along fixed transects at low altitude (ca. 50 m above ground level), and waterfowl pairs are counted on individual wetlands. The sampling design allows the data from these transect surveys to be extrapolated to the entire population based on the area covered. A detailed description of this methodology and results from the annual survey are available (e.g., U.S. Fish and Wildlife Service 2011). To estimate the number of waterfowl that are expected during the fall migration (i.e., fall flight index), breeding population estimates are combined with estimates of habitat conditions, adult summer survival, and projected fall age ratio (young/adult, U.S. Fish and Wildlife Service 2011). To estimate wintering populations, the Mid-winter Waterfowl Population Survey has been conducted annually by state and federal wildlife agencies since 1935. This aerial survey covers the four migratory flyways in the United States and parts of Mexico. Results of the survey are reported annually for each of the four migratory flyways in North America, but are not comparable because of differences in survey methodology among flyways.

7.4.1.2 Monitoring Recruitment

Waterfowl recruitment was monitored annually by estimating number of broods on wetlands in the breeding grounds; however, this survey was discontinued in 2004 due to budget constraints. Individual studies, however, continue to report results on various species of interest. Recruitment can be monitored by searching for nests and counting broods. Nest searches can include systematically searching wetlands for diving duck nests or in adjacent uplands for many dabbling duck species. Once nests are located, they are monitored for activity every 3–5 days until the eggs hatch. Estimates of nest success are calculated based on the number of successful nests divided by the total number of nests monitored $\times$ 100 % (i.e., raw nest success), or by less biased methods involving calculation of the number of nest survival days using the Mayfield method (Mayfield 1975). Contemporary nest survival analyses, such as the nest analysis module in Program MARK and the logistic exposure model, also exist and are similar to predictions using the Mayfield method (Rotella et al. 2004). Brood counts can be conducted by aerial surveys, or if species-specific data are required, from the ground. These surveys are typically performed in July.

7.4.1.3 Band Recovery Programs

Analysis of band return data is the cornerstone of monitoring mortality rates for waterfowl and estimating population size. All banding data are administered

Fig. 7.8 U.S. Fish and Wildlife Service biologist affixing a leg band to a wood duck (*Aix sponsa*, *left*), and a hunter with a banded mallard (*Anas platyrhynchos*, *right*) (Published with kind permission of © Clayton Ferrell and Barry Pratt 2014. All Rights Reserved)

through the U.S. Geological Survey's Bird Banding Lab and Canada's Bird Banding Office of the Canadian Wildlife Service. The U.S. Fish and Wildlife Service, Canadian Wildlife Service, state and provincial agencies, and other non-governmental conservation organizations (e.g., Ducks Unlimited) cooperate in banding waterfowl during the summer (Fig. 7.8). Because waterfowl are hunted, bands are often reported by waterfowl hunters when a bird is harvested (Fig. 7.8). Band return data can be used to estimate the proportion of the population that is being harvested, track movements of birds, and estimate population size and over-winter survival. Because the banding data are age- and sex-specific, estimation of mortality rates can be for each sex and age class. In addition, hunter surveys are conducted annually by the U.S. Fish and Wildlife Service to estimate waterfowl harvest by species. Band return data and hunter harvest surveys are important in determining mortality rates and are used, along with breeding population estimates, for setting waterfowl hunting regulations annually.

7.4.1.4 Other Monitoring Methods

Several other methods are important for monitoring waterfowl populations. Radio telemetry has been used extensively to monitor survival, habitat use, activity, and movements during all stages of the annual cycle (i.e., breeding, migration, and wintering). The presence of stable isotopes of carbon, nitrogen, oxygen, and sulfur has been used as markers to determine the natal origin of individuals that were harvested on the wintering grounds (Hebert and Wassenaar 2005). Stable isotope analyses also are used to conduct trophic studies in wetlands and assess diets of waterfowl. Genetic markers have been used to identify waterfowl species and sub-species, where physical characteristics prevent differentiation. This approach has been used to discriminate among sub-species of Canada goose (*Branta canadensis*, Mylecraine et al. 2008).

7.4.2 Shorebirds

There are 49 species of shorebirds (Order *Charadriiformes*) that regularly breed in North America and warrant monitoring at local, regional or continental scales (Brown et al. 2001). Similar to waterfowl, shorebirds breed at northern latitudes in North America, but typically migrate farther distances. Many species of shorebirds that breed in northern Canada migrate to Central and South America, resulting in round-trip distances exceeding 20,000 km (Helmers 1992). Considering that flight is energetically demanding (Loesch et al. 2000), migrating shorebirds must land frequently to acquire high-energy food resources (Skagen and Knopf 1993). Mudflats and shallowly flooded wetlands are the primary foraging habitats used by migrating shorebirds (Helmers 1992). Widespread wetland loss in the continental United States has presumably led to less foraging and resting habitat for shorebirds than what was historically available (Brown et al. 2001). Over half of North American shorebird species are in decline, with most species representing long-distance migrants (Brown et al. 2001). Bart et al. (2007) estimated that 23 of 30 shorebird species in northeastern North America were experiencing declines. Obtaining accurate and precise estimates of shorebird population sizes is fundamental to ensuring conservation of this imperiled group of wetland fauna. A monitoring strategy for these species has been developed under the North American Shorebird Conservation Plan (Howe et al. 2000).

7.4.2.1 National Programs

Skagen et al. (2003) outlines the major components of the continental monitoring plan for shorebirds, which is the Program for Regional and International Shorebird Monitoring (PRISM). Shorebird populations are monitored by natural resource organizations and private partners in key breeding areas, during migration at key stopover areas, and on the wintering grounds. The continental survey is based on surveying 10–16 ha wetland plots, selected as a sample from a geographic information system of wetland areas. A rapid assessment approach is used to count shorebirds in these plots, based on point counts, area searches, or transect counts. More intensive methods are used on a subsample of plots to develop a correction factor for the rapid assessment density estimates. In temperate areas where roads are available, the North American Breeding Bird Survey can produce reliable results for common species. However, rare or imperiled species require a focused sampling approach. At stopover sites in the continental United States, ground counts can be conducted during the 6–8 week period when most shorebirds migrate (i.e., April–May for spring migration and August–September for fall migration). Monitored sites are visited every

Fig. 7.9 Scan sampling with a spotting scope (**a**), lesser yellowlegs (*Tringa flavipes*) foraging on a mudflat (**b**, *left*), king rail (*Rallus elegans*) responding to a callback survey (**b**, *right*), and a great blue heron (*Ardea herodias*) rookery (**c**) (Published with kind permission of © Clayton Ferrell and Matt Gray 2014. All Rights Reserved)

7–10 days during this period. Shorebirds are counted using a spotting scope or binoculars via scan sampling (Fig. 7.9). Scan sampling involves viewing a defined area over for a specified short duration (i.e., 3–5 min) with binoculars or spotting scope and counting the number of individuals present in the area by species. Wintering grounds surveys are being conducted in similar fashion in the United States, although a standardized effort needs to be developed for Central and South America. Similar to waterfowl, stable isotope analyses have been conducted for some priority species, such as the red knot (*Calidris canutus*), to link breeding areas to specific wintering sites (Atkinson et al. 2005).

7.4.2.2 Monitoring Survival and Recruitment

Survival for shorebird species can be monitored by a variety of methods. Banding return analyses have been useful for documenting range-wide movements of shorebirds; however, banding data generally have not been useful for estimating survival because of low band return rates, unlike waterfowl which are harvested. As such, most banding studies for shorebirds typically are used in a local area to answer questions about short-term survival and population turnover at a particular site (Gratto-Trevor 2004). Radio telemetry has been used to track short-distance movements, survival and habitat use. Satellite transmitters are being used to track long-distance migration by researchers at the Alaska Science Center of the U. S. Geological Survey for species of conservation concern (e.g., curlews [*Numenius*] and godwits [*Limosa*], http://alaska.usgs.gov/science/biology/shorebirds/index.php).

Monitoring recruitment can be done using standard nest monitoring protocols. Once nests are located, they can be checked every 3–5 days until hatching similar to waterfowl. Broods can be monitored post-hatching from blinds or survey stations to estimate brood survival until fledging. Obtaining accurate chick counts in shorebird broods from visual surveys can be challenging due to their small size and cryptic coloration. To increase detection, small radio transmitters can be attached to a subsample of chicks in each brood.

7.4.3 Wading Birds

Species of herons, egrets, storks, ibises, flamingos, and spoonbills are classified as long-legged wading birds. There are 38 species of wading birds in North America. Wading birds exhibit a wide range of life history strategies. Some species are largely non-migratory, others are exclusively migratory, and some species are migratory only in the northern portion of their range. Wading birds are predatory in their foraging approach, feeding in aquatic habitats on fish, amphibians, reptiles, crustaceans, and other invertebrates. Many of these species are colonial nesters in rookeries, which provide opportunities for population monitoring unique to this group.

7.4.3.1 National Programs

An international conservation plan has been drafted for waterbirds, including waders (Kushlan et al. 2002). The U.S. Geological Survey Patuxent Wildlife Research Center hosts a Waterbird Monitoring Partnership, with the goal of coordinating and standardizing efforts to monitor waterbird populations in North America. Because many species of waders are colonial nesters, the best time to monitor populations is during the breeding season in nesting colonies (Steinkamp et al. 2003). Populations can be monitored through a two-stage approach. First, nesting colonies are located (Fig. 7.9). Depending on the area being covered, this can be done by ground-based, boat, or fixed-wing aircraft surveys. Once a colony is located, then colony visits can determine the species composition and the number of active breeding pairs. Breeding pairs can be counted directly or the total number of nests can be counted. The number of nests generally exceeds the number of breeding pairs because not all nests are actively used in a given breeding season. If the colony is relatively small (e.g., <100 breeding pairs), then a complete count may be possible. As colonies become larger, a standardized sampling approach is needed. Typically, fixed-width belt transects are walked through the colony. Breeding pair estimates per unit area from the sample can be extrapolated to the entire colony to estimate population size. Repeat visits may be necessary to account for imperfect detection and temporal variability during the breeding season. Colonies with dense vegetation and poor visibility from below require aerial surveys for monitoring. Aerial surveys can either directly count individual nests or adults, or aerial photos can be taken and inspected in the lab.

7.4.3.2 Monitoring Survival and Recruitment

There are a limited number of studies documenting survival and recruitment in wading birds (Cezilly 1997). Survival rates have traditionally been monitored via banding studies; however, large numbers of individuals need to be banded to yield sufficient returns to estimate survival. This problem can be mitigated by using auxiliary markers, such as color leg bands or patagial tags, so that banded individuals do not require recapture to be identified. Radio telemetry studies can also yield reliable survival estimates, although many species of wading birds may disperse outside the study area, and aerial tracking is required to discriminate between dispersal and mortality.

Monitoring of recruitment can often be accomplished via direct observation at nesting colonies. Nest success can be determined from repeated observation of active pairs. Counts of young produced in active nests can be made from the ground but are typically biased low because of poor visibility into nests. Climbing to a subsample of nests or combining ground-based observations with aerial observation or photography can account for visibility biases.

7.4.4 Secretive Marsh Birds

Species of rails, bitterns, coots, moorhens, gallinules, and grebes are classified as secretive marsh birds, because their skulking behavior makes them difficult to detect by conventional means. These species use freshwater and brackish marshes throughout North America. Most of these species breed across the continent and are migratory, wintering in the southern United States, Mexico, and the Caribbean. They are generally cryptically colored, nest on the ground, and spend most of their time on the ground in dense vegetation. Detection of these species in wetlands for population monitoring is problematic. As a result, specialized protocols have been developed.

7.4.4.1 National Programs

The North American Breeding Bird Survey is inadequate for monitoring secretive marsh birds because road access to wetlands is limited and passive point-count methods are inefficient at detecting these species. As a result, an independent national monitoring program for marsh birds, involving standardized count protocols and a sampling framework, has been developed (Conway 2011; Johnson et al. 2009). The count protocol is based on point-count monitoring stations that are located $\geq$400 m apart in wetlands that are representative of an area. The number of point counts conducted on each site is based on the level of precision desired by the researcher or natural resource manager (i.e., more points typically yield better precision) and available resources. The protocol involves a passive 5-min point count in which all species of interest that are heard or seen are recorded. Focal individuals are recorded during the first 1-min interval, and the distance to the individual at first detection is visually estimated. Playback recordings are then broadcast on a 30-s playback, 30-s silence interval for each focal species. Broadcast of playback recordings of many marsh birds has been shown to increase detection (Conway and Gibbs 2005). These data yield an index of relative abundance (individuals per species per point) that can be adjusted to estimate density using distance sampling and time-to-detection functions, which is discussed in greater detail in Sect. 7.4.5.2.

7.4.4.2 Measuring Survival and Recruitment

Few telemetry studies have been conducted during the non-breeding season, which would allow for seasonal estimates of survival. Although some of the secretive marsh birds are hunted (e.g., rails and gallinules), banding data are sparse and have been ineffective for monitoring survival (Eddleman et al. 1988). Capture and banding efforts by natural resource agencies and researchers are limited, and hunting pressure is too low to provide sufficient recaptures to produce reliable survival estimates. Monitoring productivity can be done by nest searching, monitoring nest success, and counting number of young fledged. Finding nests of these species can be challenging, because nests are cryptic and often located in areas that are difficult to access. Radio telemetry has aided nest finding for some species if adults can be captured prior to the nesting season.

7.4.5 Songbirds

A great diversity of songbird species use wetlands during part of their life cycle. Wetlands serve as productive breeding, stopover, and wintering sites for songbirds, because of the abundance of invertebrates and seed sources for food. Some songbirds are wetland specialists, such as the marsh wren (*Cistothorus palustris*), whereas other species use wetlands as one option in an array of potential habitats. Riparian zones along waterways provide important habitat for songbirds, even if they are not classified as a wetland.

7.4.5.1 National Programs

Several national programs monitor songbird population status and trends, and may be useful for monitoring populations of some wetland species. The North American Breeding Bird Survey (BBS) was established in 1966, and is the primary continental monitoring program for songbirds. The BBS is based on a stratified random sample of 40 km (25 mile) roadside routes conducted once during the early breeding season (late May and early June) each year. Breeding Bird Atlases (BBA) map the breeding distribution of all birds, including wetland species, across a given state or province. The Christmas Bird Count (CBC) is another long-term national monitoring program conducted by volunteer bird watchers that contains data on distribution and relative abundance of songbirds, including wetland-dependent species, during early winter. The CBC is based on volunteer bird watchers visiting a given area (e.g., portion of a county) for a prescribed period of time (observers $\times$ hours $=$ party hours), and recording the species and number of individuals encountered within that prescribed area. A training program is required to participate in the BBS, and CBC counts are performed by local volunteer groups with an experienced coordinator that is responsible for ensuring data quality. The Monitoring Avian Productivity and Survivorship (MAPS) program sponsored by Point Reyes Bird Observatory is another national program that produces estimates of survival and productivity. The MAPS program is based on constant-effort mist netting during the breeding season. The value of MAPS for monitoring wetland birds, however, is limited because very few MAPS stations are located in wetlands.

7.4.5.2 Count Methods

Songbird populations in wetlands can be monitored using various count methods including point counts, transect counts, passive constant-effort mist-netting, and territory mapping. Point and transect counts are the most easily accomplished, whereas mist-netting and territory mapping are more time intensive. Songbird vocal behavior, visibility within the wetland, and accessibility of the wetland determine which method might be most appropriate. Point counts are used extensively for monitoring during the breeding season (May–June in the contiguous United States) when songbirds are most vocal. Raw counts need to be adjusted for the likelihood of detection to yield unbiased results. Point counts are typically conducted for 5 or 10 min with bird detections recorded in 1-min intervals to allow for time-to-detection (Alldredge et al. 2007) or time-removal (Farnsworth et al. 2002) analyses. Distance to individual birds are usually estimated within 0–25, 26–50, 51–75, 76–100 and >100 m distance bands to allow for distance-detection analyses (Buckland et al. 2001). By taking time to detection and distance into consideration, relative abundance estimates can be converted to unbiased estimates of bird density for each species. Repeated visits to point count stations can be used to estimate bird occupancy (Mackenzie et al. 2006). Territory mapping is an alternative method, which involves typically eight repeated visits to a survey plot (10–20 ha), and mapping the location of all songbirds detected within the plot (Bibby et al. 2000). Thereafter, maps from all visits are overlaid and territories of individual males are mapped based on consistent detections over time. Territory mapping generally yields the most reliable density estimates (breeding pairs per ha) of any of the methods discussed.

Transect counts or constant effort mist-netting typically are more appropriate during migration or winter because detections are lower when songbirds generally are not vocalizing. Transect methods involve walking a fixed distance across a target area, recording all birds seen or heard and the distance from the observer to the bird. Program Distance (http://www.ruwpa.st-and.ac.uk/distance/) can fit probability density functions to distance-based counts and estimate bird density. Constant effort mist-netting involves setting an array of mist nets in a target area and banding all songbird species captured. Capture effort (net-hours) is recorded based on the number of nets used times the number of hours the nets are set. An index of relative abundance can be generated from these data based on captures per species per net-hour.

7.4.5.3 Measuring Survival and Recruitment

Survival estimates for most songbirds are difficult to obtain. Radio transmitters for most songbirds typically have less than 30 days of battery life, thus telemetry is of limited value for estimation of seasonal or annual survival rates. As a result, banding studies with recapture or resighting are about the only method available for estimating seasonal or annual survival. Often individual birds are banded with a unique combination of color bands to enable individual identification upon resighting (Bibby et al. 2000).

Recruitment can be measured by standard nest monitoring methods outlined above or by using videography. Nest success, the number of young fledged from successful nests, the number of renesting attempts, and the number of broods produced define fecundity for a given species. Nest success is typically monitored by determining daily nest survival and analyzing the data with the nest module in Program MARK or the logistic exposure model (Rotella et al. 2004). Patterns of juvenile dispersal and site fidelity vary widely for songbirds (Greenwood and Harvey 1982). Stable isotopes, in conjunction with genetic markers, are being used to link songbird breeding and wintering grounds (Hobson and Wassenaar 1996).

7.4.6 Raptors

Many raptors (hawks, falcons, eagles, kites, and owls) regularly use wetlands for nesting, during migration, or winter. Raptor habitat is defined by the availability of suitable prey and suitable nest and roost sites. Different habitats can often meet these requirements, thus few raptors would be considered obligate wetland species. Monitoring raptor population status and trends in wetlands is challenging, because their low relative abundances and large home ranges make detection problematic.

7.4.6.1 National Programs

Raptor monitoring is included in the previously discussed national monitoring programs (e.g., BBS, BBA and CBC), and these data are useful for estimating range wide or regional population status and trends. Raptors are also monitored across a network of continental "hawk watch" monitoring stations during migration.

7.4.6.2 Count Methods

Raptors can be monitored in individual wetland sites by conducting nest searches and monitoring the number of active pairs. Nests can be located either by aerial, boat, or ground-based surveys. Some species (eagles and osprey) have nests that can be easily detected during surveys. Point counts have been used during breeding and non-breeding seasons to monitor populations of more common species. Road-side point-count routes, similar to BBS, have been used, because long distances (e.g., 40 km) can be surveyed within a morning, increasing the probability of detection. Surveys involving broadcast of species vocalizations have also been effective for monitoring raptors to enhance detections (Mosher et al. 1990).

7.4.6.3 Measuring Survival and Recruitment

Survival rates can be estimated by banding and recapture or resighting of raptors. Color leg bands and patagial tags have been used to aid in individual identification. Survival rates have also been estimated through radio and satellite telemetry studies for many species. Satellite telemetry has also aided in identification of migratory pathways and developing linkages between breeding and wintering grounds. Stable isotopes have been used for this purpose as well.

Recruitment can be monitored through traditional nest monitoring methods. The nests of many raptors are visible to ground-based observers for monitoring nest success and number of young produced. Aerial surveys have also been used effectively for monitoring reproduction in eagles and ospreys (Fraser et al. 1984).

7.5 Monitoring Mammal Populations

A diversity of mammals occupy wetland environments, and sampling methodologies for monitoring mammals are numerous and variable, depending on the species of focus. We have separated mammals into groups including bats, small mammals, large rodents, and carnivores. Monitoring techniques for each group are well established, yet emerging techniques offer ways to improve our abilities to make inferences about mammal behavior, activity, and abundance.

7.5.1 Bats

Bats occupy a variety of habitats, including a diversity of wetland types. Bat activity is primarily nocturnal and crepuscular, and because of their size and mobility, is somewhat difficult to monitor. Bats emit high-frequency sounds, and use the echoes (echolocation) of these sounds to locate food and navigate at night. Echolocation allows bats to judge the size and location of objects in their flight path, and the speed of flight. Bats spend diurnal periods at roost sites, which may include cavities, caves, tree foliage, and various man-made structures such as bridges and buildings. Most bats are insectivores, but some bats eat fruits, vertebrates, and blood. Not surprisingly, efforts to monitor bats typically focus on roosting or foraging locations, and foraging location is ultimately determined by the species of bat encountered. We encourage readers to consult Kunz et al. (2009a, b) for further details on monitoring bats.

7.5.1.1 Acoustical Surveys

Because bats are elusive and sometimes difficult to study, much attention has focused on detecting and analyzing vocalizations to make inferences about the ecology and behavior of bats. Acoustic monitoring often focuses on estimating bat activity across habitat types (Gannon and Sherwin 2004), evaluating resource use, or describing behavior (Barlow and Jones 1997). Acoustic surveys are a noninvasive method to survey bats across broad geographic extents at relatively low cost. Acoustic surveys rely on being able to compare calls heard in the field to reference collections, but they have inherent biases because the likelihood of detection probably differs among bat species (Brigham et al. 2004). Nonetheless, acoustic surveys can provide qualitative and quantitative assessments of bat activity and habitat associations. The quality of data gained from any acoustic survey will ultimately depend on the quality of the library of calls from known species available to the observer. Observers should attempt to standardize equipment used during acoustic surveys if comparisons among sites or habitats are desirable. Acoustic detectors can be used in practically any habitat, and multiple units can be deployed by one person, making them efficient for collecting large amounts of data across broad spatial scales (Rodhouse et al. 2011). Various song meters and recognition software, such as SonoBat, are available, which detect high-resolution full spectrum sonograms. Song Scope® by Wildlife Acoustics (http://www.wildlifeacoustics.com/) is a versatile software that can analyze calls from bats, birds and anurans, thus permitting simultaneous monitoring of these groups. For bats, many researchers use Anabat or Anabat II detectors in combination with recognition software to survey bats along specific habitat features such as streams, as well as in habitats unable to be sampled effectively with other techniques (e.g., open expanses of marsh; Hayes et al. 2009).

7.5.1.2 Live Capture

There are various methods used to capture bats, but mist nets, handheld nets and harp traps are the most common (Kunz et al. 2009a, b, Fig. 7.10). In wetlands, capturing most bat species represented in the community will require multiple capture techniques given the diversity of habitat types available to bats. The type of capture method used ultimately depends on the abundance of bats in the sampling area and the expected number of flying bats that will be encountered. Areas where bats are traveling routinely, feeding or drinking are ideal capture sites regardless of the method used. Once a capture technique is selected, there are many ways to implement the technique, as mist nets, harp traps, and even handheld nets can be used in a myriad of ways (see Kunz et al. 2009a, b). Mist nets are most commonly used to capture bats, and typically set up on the ground (as with songbirds), within the tree canopy, or suspended across narrow waterbodies. Mist nets should not be used if the possibility exists of capturing large numbers of bats in

Fig. 7.10 Mist nets (**a**) are effective at capturing bats in a variety of trapping situations. Harp nets (**b**) are often used at the entrances and exits to roosting structures, and should be used if capturing large numbers of bats is possible (Published with kind permission of © Steven Castleberry 2014. All Rights Reserved)

short time periods. Mist nets should be checked several times per hour, and bats removed as quickly as possible. Harp nets can be used to capture large numbers of bats near the openings of caves and other roosting structures. Handheld nets also can be used to capture bats exiting roosting structures.

Special care should be taken when removing bats from traps to prevent injury to the bat and observer. Similar to birds, determine the direction from which the bat entered the trap, and attempt to remove the bat from that direction. Researchers attempting to capture bats should wear leather gloves. The thickness of the glove should be chosen considering the size and aggressiveness of the bat species handled. Rabies virus is the most significant public health concern related to bats, so individuals that frequently handle bats should be vaccinated.

7.5.1.3 Radio Telemetry

Because telemetry involves capture of individual bats, it is a relatively invasive and costly form of monitoring. However, properly designed telemetry studies (e.g., Ratti and Garton 1994; Millspaugh and Marzluff 2001) can provide observers with movement and behavioral data that are impossible to obtain with other techniques. Transmitters can be attached to bats in a variety of ways depending on the size of the bat. Telemetry studies on small bats typically use surgical glue to attach transmitters on the back between the scapulars, whereas larger bats may be able to wear collar or necklace transmitters. Regardless of the attachment, transmitters should weigh less than 5 % of the body weight of the bat, including the adhesive or attachment used. Monitoring of bats with telemetry typically uses either homing (e.g., to locate a roost site) or triangulation, with one or multiple observers. If movement ecology is a primary objective, the use of multiple receiving stations and simultaneous azimuths should be considered (see review by Amelon et al. 2009).

Researchers using radio telemetry to monitor bat populations need to consider the target number of animals to be monitored and the desirable number of relocations per animal. In general, if monitoring fine-scale movements is an objective, more relocations are necessary. Conversely, study objectives may make it necessary to monitor greater numbers of individuals, potentially resulting in fewer relocations per individual. Monitoring bats with radio telemetry requires careful consideration of research designs prior to capture and marking.

7.5.1.4 Roost Surveys

Bats use roosts for extended periods of time and for various reasons (e.g., hibernation), so surveying roosts to monitor bat populations is commonly used. Roost surveys may be used to determine colony size, or to monitor temporal trends in abundance (Warren and Witter 2002). Typically, repeated visits to roost sites through time are necessary to provide inferences on changes in colony size. Because populations of bats may use several roosts through time, it is critical to locate roosts being used in the area, and determine the number of roosts in the area being studied (Hayes et al. 2009). Field staff responsible for monitoring roosts should strive to minimize disturbance to roosts and roosting bats, as increased disturbance can result in roost abandonment and population declines (Mann et al. 2002).

Bats roost individually or in large aggregations in a variety of sites, including caves, under bridges and in buildings, as well as in trees. Monitoring roosts in wetlands could involve any of these sites, and each introduces unique challenges to those conducting the monitoring efforts. Caves and other subterranean roosts, as well as buildings and bridges, can either be sampled internally to directly count numbers of bats, or externally to count bats as they exit. Internal surveys can result in disturbance, but can provide fairly precise estimates of bat abundance. With the recent emergence of white-nose syndrome, field gear and footwear should be disinfected with 10 % bleach for a minimum of 10 min to inactivate the fungus (*Geomyces destructans*) associated with this disease. White-nose syndrome is a disease responsible for significant deaths in bat populations throughout the eastern United States. Infrared cameras and night vision goggles allow precise estimates of the numbers of bats emerging from roost sites (Kunz et al. 2009a, b), as long as all roost openings are monitored simultaneously. Monitoring bats that roost in trees can be difficult, because exit points are difficult to determine and bats roosting in trees tend to show low fidelity to roost sites (Barclay and Kurta 2007). Radio telemetry is an effective way to determine selection of tree roosts and fidelity to roost sites so that monitoring programs can be established in areas.

7.5.2 Small Mammals

A rich diversity of terrestrial small mammals use wetlands. Small mammals are typically secretive and nocturnal, and serve numerous ecological roles important in wetland ecosystems. Specifically, small mammals serve as seed dispersers, prey for many vertebrate species, and are important in nutrient cycling (Dickson 2001). Because of their variable sizes and niche differences, adequate monitoring of small mammal communities requires multiple techniques, with the understanding that small mammal populations fluctuate considerably throughout the year. Typically, population size of many small mammal species peaks during fall and is lowest during early spring prior to the first reproductive pulse. Monitoring strategies should be designed to encompass the entire small mammal community, ranging from the smallest (e.g., shrews) to the largest (e.g., woodrats) species.

7.5.2.1 Live and Removal Trapping

Snap-traps can be used to provide unbiased estimates of species composition, relative abundance, occurrence, and distribution of small mammals. However, because using snap-traps involves removing individuals from the population, this technique should be used only once annually on the sites being studied. Snap-traps should be placed along transects rather than grids to maximize capture rates and increase the probability of sampling all species present (Pearson and Ruggiero 2003). We recommend placing traps along microhabitat attractive to small mammals, such as woody debris and trails. Traps can be placed systematically along transects at regular intervals (e.g., 10–15 m) or stratified in different wetland types. Sampling duration will depend on specific objectives of the monitoring program, but generally traps should be operated for several days to a week to adequately estimate species composition. There are two general sizes of snap traps: those designed to sample smaller mice and shrews, and those capable of capturing larger mice and rats. To improve accuracy of population estimates, both trap sizes should be used. Pre-baiting snap traps can improve capture rates once sampling begins, but if pre-baiting is not possible, field staff should consider lengthening the trapping period by up to 4–6 nights (Ritchie and Sullivan 1989).

Live trapping small mammals is used widely, and various trap designs (e.g., Longworth, Sherman) are available to capture species ranging from shrews to larger rodents. Typically, using multiple live-trap types simultaneously more adequately samples the entire small mammal community (Anthony et al. 2005). Trap arrangement will depend on whether estimating species occurrence or density is an objective. Live traps can be placed along transects similar to snap-traps or randomly within wetland habitats to estimate species occurrence or composition. However, if density estimates are needed, traps should be placed using a grid or web design (Greenwood 1996). Spacing among traps usually ranges from 10 to 25 m, and traps are usually baited with peanut butter or grains (e.g., oats), often in combination.

Pre-baiting live traps can improve capture success, although results among studies are highly variable and often conflicting (Edalgo and Anderson 2007).

7.5.2.2 Pitfall Trapping

As discussed, pitfall trapping can be used to study herpetofauna, but this technique can be equally effective at evaluating occurrence and spatial distribution of some small mammals. For example, fossorial small mammals that have locally confined movements, such as shrews, are readily captured in pitfall traps (Laakkonen et al. 2003). Pitfall traps can be more effective and efficient than other methods at sampling small mammals, particularly with rare species (Umetsu et al. 2006). However, small mammals captured in pitfall traps can suffer mortality in the traps, so traps should be covered while sampling is not occurring. Pitfall traps vary in size and shape, but all consist of a collection device (e.g., buckets, drums, or tins) buried in the ground with the top level with the ground. Drift fences that funnel animal movements into the pitfalls can improve capture rates. Pitfall traps should be placed in areas most likely to be encountered by the species of interest. For additional details on pitfalls and drift fence designs, please see the previous sections on amphibian and reptile monitoring.

7.5.3 Wetland-Dependent Rodents

Nutria (*Myocastor coypus*) are indigenous to South America, but were introduced in North America primarily for production and sale of their pelts (Kinler et al. 1987). They are most abundant in freshwater and brackish wetlands, and construct burrow systems along banks that range from simple tunnels to complex systems with multiple entrances and exits (Bounds et al. 2003). Nutria forage on various species of wetland plants, and their foraging and burrowing activity can significantly compromise the function and quality of wetlands (Shaffer et al. 1992). Monitoring for nutria sign and evidence of foraging can provide estimates of relative abundance and be used to make decisions on when population control is needed. Signs of nutria include burrows, resting structures, paths or runs where animals exit the water, evidence of rooting or excavating plants, plant herbivory, and feces (Gosling and Baker 1991). Generally, these signs increase as density increases, and the amount of destruction often is greater during winter when food resources are limited.

Beavers (*Castor canadensis*) are the largest rodent in North America, and have well developed family units called colonies. Beavers consume primarily herbaceous and woody plants, and the leaves, buds, bark, and twigs of woody plants are typically the most important component of the diet throughout their range. Beavers are a true keystone species, as their dam-building and foraging activities have profound effects on wetlands. Surveying beaver populations often involves estimating numbers of colonies and mean colony size (Novak 1987). Colony size can be difficult to determine, and varies spatially and temporally. Aerial surveys have been widely used to estimate size of beaver populations, and are typically conducted during fall. If precise estimates of beaver abundance are desired, beavers can be trapped in box traps, snares, or specially designed traps (e.g., Bailey or Hancock traps; Baker and Hill 2003). If trapping is used, field staff should use enough traps to capture all colony members as quickly as possible, as beavers become trap-shy quickly.

Muskrats (*Ondatra zibethicus*) are widely distributed in wetlands throughout North America. Muskrats require emergent, submersed, or shoreline vegetation, and often use this vegetation for forage and to construct houses. Houses provide protection from predators and safe areas for raising young, and coupled with burrows and tunnels along banks of wetlands, are signs of muskrat presence. Counting houses is used widely to monitor muskrat populations, and can be conducted by air or boat (Erb and Perry 2003). Abundance of houses can vary seasonally, as water levels and spring breeding activity can result in either decreases or increases in the number of houses constructed (Palmisano 1972). If an estimate of muskrat density is desirable, mark-recapture techniques using cage traps are effective (Clark and Kroeker 1993). Traps should be baited with fruits or vegetables, and covered with natural vegetation to provide concealment and increase capture success. Captured muskrats should be carefully removed from the trap by grabbing their tail, and placed head first into a wire cone for marking with standard ear tags (Erickson 1963).

7.5.4 Carnivores

Terrestrial carnivores are prominent species in wetland systems, and include truly carnivorous species such as river otter (*Lontra canadensis*), as well as the omnivorous raccoon (*Procyon lotor*). Carnivores range in size from diminutive weasels, to larger canids such as the coyote (*Canis latrans*), and even large felids such as bobcats (*Lynx rufus*) and cougars (*Puma concolor*). Carnivores are secretive species, being most active during crepuscular and nocturnal periods. As such, monitoring techniques are highly variable, with most relying on attracting animals to a site where they are captured or sign is collected (e.g., tracks, hair). Additionally, passive techniques (e.g., remote cameras) can be used to monitor abundance or distribution. Similar to many other mammals, monitoring carnivore populations requires a working knowledge of potential species at a site and a willingness to use multiple techniques.

7.5.4.1 Live Trapping

Cage traps are widely used to capture mammals in wetlands, including raccoons, river otters, mink (*Neovision vision*), and beaver. Animals captured in cage traps

Fig. 7.11 Various mammals (e.g., bobcat [*top*], mink [*middle left*], river otter [*middle right*]) can be effectively and humanely captured in foot-hold traps; wire cage traps also are effective at capturing small mammals (e.g., raccoon) (Published with kind permission of © Ryan Williamson and Mike Byrne 2014. All Rights Reserved)

typically must be sedated or restrained if handling is necessary. Field staff responsible for chemical immobilizing mammals should refer to Kreeger and Arnemo (2007) for information on appropriate methods and drugs for safely removing individuals from traps. If marking animals is necessary to meet management objectives, cage traps are effective and efficient. Foot-hold traps and other restraining traps (e.g., snares) can be used to capture larger carnivores, and capture rates for many species (e.g., coyotes, bobcats) increase dramatically when using these trap types (Fig. 7.11). Trapping with foot-hold traps requires skill and training, as well as the ability to remove captured animals safely from the trap. As such, monitoring carnivores through trapping with restraining traps is time and labor intensive. However, if research objectives require radio marking of species such as coyotes and large felids, using foot-holds to capture individuals may prove most efficient (Shivik et al. 2005). Alternative designs to foot-restraint traps, such as the EGG™ trap, have been shown to be more effective and efficient than cage traps in capturing raccoons (Austin et al. 2004).

7.5.4.2 Camera Surveys

Many carnivores are rare and elusive, so remote cameras offer opportunities to observe behavior, estimate abundance, and evaluate occurrence of various species across broad spatial scales. Remote cameras have numerous advantages over other survey techniques, producing relatively large datasets with minimal effort and labor. Likewise, cameras can be set to detect carnivores of varying sizes and because animals do not have to exhibit any particular behavior (e.g., stepping into a trap) to be photographed, they have little bias (Kays and Slauson 2008). However, the cost of initiating a camera survey can be substantial relative to other passive monitoring techniques (e.g., track surveys, scent stations).

Most surveys designed to monitor carnivores should use either active infrared or passive infrared sensors. Active sensors require multiple units, whereas passive sensors only require one sensor component; passive sensors are easier to set up and are manufactured widely. Regardless of the sensor used, most camera sets are aimed at game trails or baited sites. Field staff using remote cameras should become familiar with features of the cameras used, and develop standardized protocols in regards to the height of deployment and the distance between the camera and the intended target.

Beyond deciding which types of cameras to use and how to deploy them, those using remote cameras to survey carnivores also need to consider whether or not to use bait, and whether camera avoidance (through flashes) is a concern (Wegge et al. 2004). Likewise, the cost of purchasing individual cameras, monitoring units in the field, and reviewing photographs should be considered before initiating a camera survey. Designing a camera survey will ultimately be an exercise in balancing the best data with the most efficient use of available cameras. Typically, cameras should be spaced relative to the home range characteristics of the species' studied (Kays and Slauson 2008), although the arrangement of cameras within the surveyed area can be highly variable (Gompper et al. 2006).

7.5.4.3 Genetic Sampling

The use of noninvasive hair sampling methods has increased rapidly during the past decade, and is now being used widely to sample carnivores in a variety of habitat types, including wetlands (Kendall and McKelvey 2008). Hair collection methods vary, but generally use baited sets to encourage carnivores to deposit hair (e.g., hair corrals, Fig. 7.12), or unbaited sets where natural behaviors facilitate the animal

Fig. 7.12 Non-invasive hair collection site for black bears (*Ursus americanus*) with bait (*top inset*) and barbed wire (Published with kind permission of © Michael Hooker, Jared Laufenberg, and Carrie Lowe 2014. All Rights Reserved)

leaving hair samples (e.g., rub trees used by bears). Regardless, hair collection provides genetic samples that can be used to estimate density using mark-recapture methods, and can allow researchers to track individuals on the study site (e.g., estimate survival through time). Hair samples also can be used for stable isotope analyses to learn about carnivore diets or migratory movements (Fox-Dobbs et al. 2007). However, analyzing large numbers of hair samples can be costly, and genetic material recovered in the field can degrade rapidly in wetland environments. Monitoring of collection stations should be frequent enough to recover hair in a timely manner so as to minimize degradation of DNA. This monitoring protocol will differ by species and site (Goosens et al. 1998). Degradation of hair samples is influenced by environmental factors, but a good rule of thumb is to collect samples at least every 7 days.

Amplifying DNA from fecal samples has gained attention recently as a way to monitor carnivore abundance and individual ecology (e.g., prey selection by known individuals). Feces contain sloughed epithelial cells, and a single sample typically provides enough material to recover DNA multiple times. Species depositing feces can often be identified with considerable success, although identification of individuals is highly variable (McKelvey et al. 2006). Fecal samples rapidly deteriorate, so field staff should be trained on ways to dry samples in a manner that allows them to be stored prior to analysis. Drying and storage protocols differ greatly among species, hence someone interested in initiating a study using fecal genotyping should either conduct pilot studies to determine which technique(s) are most effective in their particular situation, or use techniques proven successful on similar species (Schwartz and Monfort 2008).

7.5.4.4 Track and Sign Surveys

During the course of normal daily activities, carnivores leave signs that can be used to determine species distribution, relative abundance, and occurrence across the landscape. Sign surveys are inexpensive, use the tracking medium on site (e.g., snow, sand), and can be conducted by practically anyone trained to identify the track of interest. Likewise, scat surveys are commonly conducted to determine carnivore diets, but the ability to identify species and individuals, as well as the recent use of dogs to locate scat (see MacKay et al. 2008 for a thorough review) have increased the usefulness of scat surveys. Additionally, some species deposit scat in a way that is easily detected or can be used to survey population trends (e.g., river otters; Kruuk et al. 1986; Heinemeyer et al. 2008). Regardless, the probability of detecting a species and monitoring abundance ultimately depends on survey effort and access to the study site and survey area (Harrison et al. 2004).

Beyond simply noting tracks left through normal activities, monitoring carnivore populations using tracks typically involves the use of track plots, track plates, or scent stations. Track plates require the use of manufactured materials, whereas track plots and scent stations typically rely on prepared substrates (e.g., sand, sifted dirt). Scent stations are sites where tracking substrate (soil, sand) is prepared or placed, then baited with something that serves as an olfactory attractant to stimulate visitation by carnivores. Scent stations have been widely used to estimate occurrence, distribution and relative abundance of carnivore species (Sargeant et al. 2003; Zielinski et al. 2005). Track plates are metal plates coated with smoke residue that provides a high-quality tracking medium, and have been used to survey numerous species worldwide (Ray and Zielinski 2008). Track plates are either designed to be closed (contained within a structure that the animal enters) or open (no structure), and may be used to target specific species or groups of species. Scent stations and track plots will readily detect most felids and canids, hence they are widely used in a variety of habitats. However, a number of factors, such as weather, season, space use patterns, and species density, are known to affect how individuals respond to track stations (Harris and Knowlton 2001); these should be considered when designing and implementing track surveys. Likewise, scent stations provide more reliable data when used across large spatial scales and with large samples of stations. Ultimately, numbers of stations deployed and the choice of survey design will depend on logistical constraints. We encourage researchers using scent stations to carefully consider recent assessments of sampling designs (Sargeant et al. 2003) when implementing scent station surveys.

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Student Exercises

Laboratory Exercise #1: Herpetofaunal Sampling Laboratory

Location and Time: Herbaceous or forested wetland with standing water during the growing season (ideally spring or summer).

Description: The goal of this field lab will be to build competency in common techniques used to sample amphibian and reptile communities. Activities described herein could be assigned in their entirety or only portions of the exercise used. If completed in its entirety, students will have an understanding how to use pitfalls, funnel traps, and nets to capture amphibians, use basking traps to capture turtles, and collect biological information on captured amphibians and turtles.

Supplies and Equipment: Silt fencing with attached wooden stakes, 3-lb sledge hammer, rake, tape measure, shovel, six 19-L (5-gal) buckets, six sponges, six rectangular funnel traps, dip nets, seine net, basking turtle trap, snake tongs, calipers, organism bags, spring scales, and disposable gloves (worn while handling animals). Given that live animals will be captured and handled, scientific collection permits must be secured, and an Institutional Animal Care and Use Committee (IACUC) protocol may be required by your institution.

Set-up Instructions: Identify a wetland for sampling and remove leaves, debris, and herbaceous vegetation in a 0.5-m wide band 10 m above the high waterline and parallel to the wetland for 40 m. Dig holes for 5-gal pitfalls every 10 m, with two pitfalls paired at each end. Pitfall tops should be flush with the top of the ground. Erect fencing such that it passes next to each pitfall and 0.5 m past the end pitfalls. Cover the base of the fence with soil to prevent trespass of animals. Fill pitfalls with 2.5 cm of water from the wetland and place one sponge in each pitfall. Place one funnel trap on each side of the fence, and one trap in shallow water (<10 cm) in each cardinal quadrant of the wetland. Construct a basking trap following Brown and Hecnar (2005), and place in water >1 m depth.

Sampling Instructions: After deployment of traps, check in <24 h. Identify all captured juvenile and adult amphibians and reptiles in pitfall and funnel traps, and measure the snout-to-vent length and mass. Amphibians can be placed in plastic bags when processing but should be rehydrated with water from the wetland before release. Lizards can be placed in cloth bags or plastic containers. When handling lizards, avoid grabbing by the tail because most species will autotomize it as an anti-predator response. Captured snakes should only be handled after verifying they are not venomous; non-venomous snakes can be temporarily held in a well-secured pillowcase. Venomous snakes should not be handled and be removed from traps using snake tongs or snake tubes. Aquatic and terrestrial turtles can be placed in 5-gal buckets or large plastic containers. Care should be taken when handling snapping turtles because their bite can cause injury. Larval amphibians can be sampled using dip and seine nets following Schmutzer et al. (2008). Identify and enumerate all larval amphibians by species. As a second exercise, determine developmental stage according to Gosner (1960).

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Schmutzer AC, Gray MJ, Burton EC, Miller DL (2008) Impacts of cattle on amphibian larvae and the aquatic environment. Freshw Biol 53:2613–2625

Laboratory Exercise #2: Amphibian Marking Laboratory

Location and Time: This laboratory can be completed in combination with the Herpetofaunal Sampling Laboratory or in the classroom with preserved laboratory specimens.
Description: The purpose of this laboratory is to practice common herpetofaunal marking methods. Activities in this laboratory can include marking techniques for adult or larval anurans. If this laboratory is completed in its entirety, students will have the experience necessary to mark amphibians using scissors, Visible Implant Elastomers (VIE), and Passive Integrated Transponder (PIT) tags.

Supplies: Wild captured or preserved amphibians (3 per student or team), surgical grade scissors, VIE mix and injection syringes (Northwest Marine Technologies, Inc.), PIT tag supplies (PIT tags, tag reader, and injection syringe [Biomark, Inc.]), 95 % EtOH or 2 % chlorhexidine diacetate solution, disposable gloves, and appropriate marking schemes. If live amphibians are captured, scientific collection permits must be secured, and an IACUC protocol may be required by your institution.

Set-up and Instructions: Wild amphibians can be captured during the Herpetofaunal Sampling Laboratory, and preserved specimens (e.g., *Necturus*, *Lithobates*) can be acquired from most biological supply companies. If wild animals are used, it is important to sanitize all marking instruments using 95 % ETOH or 2 % chlorhexidine diacetate solution. Prior to the lab, review and select one of the published marking schemes for amphibians (see Donnelly et al. 1994 or Ferner 2010). Students should work on a stable, clean surface. Distribute at least three amphibians to each student or group (i.e., one amphibian per marking technique). Students should wear disposable gloves and change them between animals. Practice toe clipping as described in the chapter. The excision should be made at the most proximal joint; bones and thumbs should not be cut. Excise the appropriate toes to number the individual as #234. Once completed, use a different animal for VIE marking. The elastomer should be injected under the skin where very little pigment occurs; the ventral side of most amphibian legs is a good location. Care should be taken to slide the needle under the skin (forceps can help) so as to not pierce muscle or organs. When working with live amphibians, students should work in pairs, with one student holding the animal securely. Students should conceive schemes that allow for individual or batch marking using different VIE colors and marking locations on the amphibian. Finally, practice injecting a PIT tag under the skin mid-body on ventral and dorsal sides. Scan the tag prior to and after injecting. At the conclusion of the laboratory, students should discuss advantages and disadvantages of each technique. Processing time for each technique should be recorded and considered.

Donnelly MA, Guyer C, Juterbock JE, Alford RA (1994) Techniques for marking amphibians. In: Heyer WR, Donnelly MA, McDiarmid RW, Hayek LAC, Foster MS (eds) Measuring and monitoring biological diversity: standard methods for amphibians. Smithsonian Institution Press. Washington, DC, pp 277–284

Ferner JW (2010) Measuring and marking post-metamorphic amphibians. In: Dodd CK (ed) Amphibian ecology and conservation: a handbook of techniques. Oxford University Press, Oxford, pp 123–142

Laboratory Exercise #3: Small Mammal Trapping Laboratory

Location and Time: Perimeter of a forested or emergent wetland in areas not subject to permanent flooding, or the adjacent upland can be used; performed preferably during fall.

Description: The goal of this lab will be to expose students to basic live-trapping techniques used to estimate abundance, density, or distribution of small mammals. This lab will require students to establish trapping grids prior to setting traps, set traps in the afternoon, and check all traps the following morning. Students will gain an understanding of how to establish trapping grids, set traps, capture and mark small mammals, and collect basic morphological and demographic data.

Supplies: Sherman or Longworth traps, flagging tape, peanut butter and oats, wax paper, clear plastic bags, spring scale, and disposable gloves. Given that live animals will be captured and handled, scientific collection permits must be secured, and an IACUC protocol may be required by your institution.

Set-up Instructions: Identify an area that is not permanently flooded with suitable herbaceous or woody vegetation to harbor small mammals. Establish a trapping grid with a minimum of 25 traps (e.g., 5 $\times$ 5 matrix) with each trap placed 10 m apart. Place flagging at each trap site so that traps can be quickly relocated. Prior to setup, mix oats and peanut butter together. Cut 3 $\times$ 3 in. squares of wax paper, and place ½ teaspoon of peanut butter mixture in center of wax paper, fold the ends together, and twist so that the peanut butter mixture is inside a pouch of wax paper.

Bait each trap by placing the bait balls (inside of wax paper) at the back of each trap. Traps should be placed in areas likely to be used by small mammals, such downed woody debris, stumps, etc. Place one trap at each trapping site during the afternoon or evening prior to the day that traps will be checked. Trap sites should be spatially referenced using a GPS unit to ensure re-location of the sites during monitoring.

Sampling Instructions: All traps should be checked <16 h after being set. Mammals captured in traps can be removed by opening the door of the trap, placing a clear plastic bag over the trap door, and turning the trap over to drop the captured individual into the bag. Each person handling mammals should wear disposable gloves. Mammals can be carefully removed from the bag by pinching the fur around the back of the neck. Larger mammals, such as cotton rats (*Sigmodon hispidus*), can be further restrained by holding their tails with the other hand. Identify mammals to species and determine sex. Small mammals can be identified using Peterson (2006). Body weight can be determined by closing the plastic bag briefly, and hanging the bag from a spring scale. Capture data should be summarized by species, and inferences made about their association with different cover types. For instance, students could simultaneously measure habitat characteristics (e.g., canopy cover, vegetation type, vegetation density) at sites where mammals are successfully captured and compare them to sites where mammals are not captured.

Peterson RT (2006) Peterson field guide to mammals of North America, 4th edn. Houghton-Mifflin, Boston

Laboratory Exercise #4: Evaluating Wildlife Sign for Surveys

Location and Time: In-class laboratory with slide presentation.

Description: The goal of this lab is to train students to identify tracks and sign of common mammals likely to be encountered in various wetland habitats. The slide presentation is designed to provide students with information on how to identify tracks based on numbers of toes, distance between front and rear feet, morphological characteristics of feet among species, as well as gait patterns. The lab is most effective if students have plaster casts of the species covered in the presentation so that they can view tracks and study them.

Supplies: Slide presentation (PDF format) by Mike Chamberlain is available for use at: http://fwf.ag.utk.edu/mgray/WetlandBook/WildlifeSignsLab.pdf. If track casts are unavailable, a reference collection can be created using Plaster of Paris available at craft stores.

Set-up Instructions: The presentation describes how to identify tracks of mammals based on numbers of toes. Specifically, students should be instructed on ways to recognize 2-toed hooved species, 4-toed species with heal pads, 5-fingered species, and species with 4 front toes and 5 hind toes. Mammals occupying wetlands vary by locale, but larger, more common species, such as white-tailed deer, feral hog, coyote, red fox, gray fox, bobcat, cottontail and swamp rabbits, raccoon, opossum, muskrat, beaver, mink, river otter, and black bear, are covered in this lab.

Laboratory Exercise #5: Waterbird Identification, Sexing and Aging Laboratory

Location and Time: Indoor laboratory during fall or spring. This lab should be conducted prior to the Waterbird Population Monitoring field lab.

Description: The goal of this lab will be to expose students to waterbirds that are of management and conservation interest in the state and region. In addition, sexing and aging techniques by plumage will be demonstrated for species where this information is of management interest, such as waterfowl. The students will be responsible for identification of the species that are presented in the lab and sexing and aging for a subset of those species.

Supplies: Photographs of species of interest and study skins where possible, waterfowl wings for males and females of species of interest, bird field guide, and waterfowl wing sexing and aging guide (Carney 1992). Also, a slide presentation by Matthew Gray on the identification of North American waterfowl is available: http://fwf.ag.utk.edu/mgray/wfs560/WaterfowlID.pdf.

Classroom Instruction: Develop a list of waterbirds that students will be responsible for learning including waterfowl, wading birds, shorebirds, and secretive marsh birds. Include species that are generally of management interest for the state or region, including species that will likely be encountered during the field lab. Develop a slide presentation in which the instructor reviews the identification characteristics of the species on the list. The instructor should also review the sexing and aging techniques for species of interest. After the presentation is complete, the students will break into 2-person teams to review the specimens that are available using their field guides to make a positive identification. In addition, they can use the U.S. Fish and Wildlife Service sexing and aging guide for waterfowl wings as additional practice (see below).

Lab Proficiency Quiz: When each student (or team) believes they have mastered identification, they can attempt a proficiency quiz. The quiz should include images of birds at varying distances. They will be declared proficient if they correctly identify >70 % of the birds. Students should demonstrate proficiency prior to the field lab.

Carney SM (1992) Species, age and sex identification of ducks using wing plumage. U. S. Department of the Interior, U.S. Fish and Wildlife Service, Washington, DC. Northern Prairie Wildlife Research Center, Jamestown. http://www.npwrc.usgs.gov/resource/tools/duckplum/index.htm

Laboratory Exercise #6: Waterbird Population Survey Laboratory

Location and Time: Wetlands with open water and mudflats during fall or spring.

Description: The goal of this field lab will be to demonstrate population survey methods for waterfowl, shorebirds and wading birds, and allow students to practice the methods, analyze the data, and interpret the results.

Supplies: Binoculars and spotting scopes, laser rangefinders, study area maps, bird field guides, clipboards, data sheets, and 1-m stakes for each student team.

Set-up Instructions: Identify a wetland with open water and mudflats for survey. Divide the wetland area into sampling units based on geographic area (e.g., cardinal quadrants) or habitat. Use the maps, laser rangefinders, and stakes to delineate the spatial extent of each survey location. Each location should survey approximately the same viewable area. Divide the class into 2–4 person teams and assign each team to a location.

Survey Instructions: At the beginning of the laboratory, explain the goals of the exercise and review the count protocols and identification for species likely to be encountered. If there are species that are difficult for novices to identify (e.g., various sandpiper species), group these as morpho-species (e.g., western, least and semi-palmated sandpipers might be counted simply as "sandpipers"). Deploy the teams to conduct the counts, ideally within 3 h of sunrise or sunset. Each team should spend the first 30 min scanning the area and identifying the waterfowl, wading birds and shorebirds to species. The last 15 min of the count period will be spent estimating a count for each species. As an additional exercise that students can practice focal surveys, where bird activities are recorded (e.g., feeding, walking, swimming, inactive, sleeping, antagonistic, alert) for 1 min. Students are encouraged to read Davis and Smith (1998) for an example of collecting and analyzing activity budget data.

Data Analyses: Data from each team should be entered into a database, including date, time, study area(s), environmental conditions (temperature, wind speed and direction, precent cloud cover, precipitation), observers, species and counts. Each team will summarize the data collected by the entire class to make inferences about waterbird use of the area. After conducting this exercise over several years, students can begin to look for seasonal or yearly changes in species composition and abundance, and develop hypotheses for why these demographics may be fluctuating.

Written Assignment: Each team will be responsible for writing up a lab report, summarizing the objectives, methods, and results from the surveys and discussing the implications of the results and answering critical questions about changes in waterbird populations. Additionally, each team may present their results orally, and a class discussion can explore the lessons learned from this experience.

Davis CA, Smith LM (1998) Behavior of migrant shorebirds in playas of the Southern High Plains. Condor 100:266–276