A Review of Research Methods for Barn Owls in Integrated Pest Management

By

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Assembled from materials and information from the 2018 BARD Conference on the Use of Barn Owls for Agricultural Pest Control, held in Davis, California, 5-7 March 2018. See page four for workshop participants.

Recommended Citation: Huysman, A., D., St. George, M. Johnson, R. Baldwin, M. Charter, S. Hindmarch, C. Wendt, S. Kross, G. Rozman, P. Rivadeneira, and E. Phillips. 2018. A review of research methods for barn owls in integrated pest management. Technical Report of the BARD Conference on the Use of Barn Owls for Agricultural Pest Control, 5-7 March 2018 at University of California Agricultural and Natural Resources. Published by Humboldt State University, Arcata, California, USA. <http://www.barnowlpestcontrol.com/uploads/1/1/7/3/117396152/Huysman_et_al_2018.pdf>

Acknowledgements

This work would not have been possible without the participants of 2018 BARD (Bi-national Agricultural Research & Development) Workshop on the Use of Barn Owls for Agricultural Pest Control, held 5-7 March 2018 at University of California Agricultural and Natural Resources. Funding was provided by BARD, with technical assistance from UC Agricultural & Natural Resources and Humboldt State University Sponsored Programs Foundation. Special thanks to Matchbook Vineyard and Button & Turkovich Ranch for generously allowing access to their properties for field days. Brooks Estes assisted with formatting and copy editing.

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Funded by BARD

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Introduction

The role of barn owls in an integrated pest management (IPM) program to control rodent populations in agriculture is subject to many facets of barn owl and rodent natural history and management decisions made by farmers. For farmers, it is vital to understand if and under what specific conditions barn owls can be an effective means of rodent control. While there are several published guidelines on how to build and install barn owl nest boxes (Marti et al. 1979, Raid 2012, Wade et al. 2012), little else has been published describing the methods and practices necessary for follow-up monitoring and research to determine whether installing nest boxes is helping to control rodents.

The methods described here can be used by landowners, researchers, and agency personnel to contribute to the question of how barn owls fit into an IPM system. This document is a product of an international workshop that assembled over 25 researchers, farmers, and students to share information about field and laboratory methods (BARD Workshop 2018). The workshop also clarified research priorities, both from an ecological perspective (reported elsewhere, Johnson et al. in prep) and from the perspective of farmers seeking to better understand how to use barn owls to help control rodents. This work revealed that farmers are particularly interested in studies that will answer which landscape features, crops, and farming practices lead to effective barn owl pest management, and how this may vary among regions. They also want practical information on how to set up boxes most effectively, how to maintain boxes for longevity, how to incorporate boxes into an IPM framework involving other methods, the best timing to install boxes, and whether crop management practices can be harmful to owls. The answers to these questions lay in the predator-prey relationships between barn owls and rodents and the farm management practices that influence them.

In this document, we review methods useful in barn owl research and monitoring for rodent control in agriculture. Specifically, we describe core options, areas in need of refinement, challenges and considerations, and recommendations for 6 key topics: 1) nest box monitoring and demography; 2) pellet analyses; 3) toxicology; 4) rodent monitoring, 5) video monitoring; and 6) telemetry and movement. Within each of these topics, we also describe the types of ecological and management questions for which the described methods may be most useful. With proper expertise, resources, and a commitment to refining current methods, this document can help those designing studies to enhance our understanding of barn owl as agents of rodent pest control.

1) Nest Box Monitoring and Demography

Applications of the method

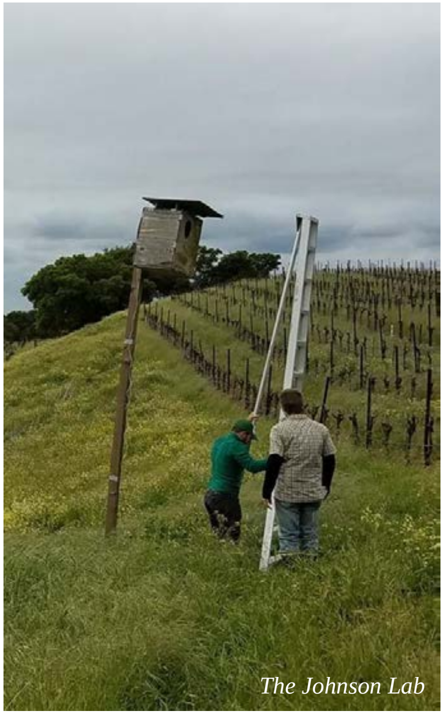
It is convenient that barn owls readily roost and nest in a variety of nest boxes. This makes locating nests and monitoring barn owl reproduction easier than having to locate nests in trees, riverbanks, and caves. Nest box location and design can also be easily manipulated to test research hypotheses. Nest box monitoring is an effective way to monitor barn owl populations, nest success, nest box use, and occupancy. Nest boxes can be used for outreach and education purposes as well as research.

Monitoring the occupancy and nest success of a group of nest boxes can be used to calculate occupancy rates, understand demographic trends, discover interesting natural history, and find the optimal nest box placement and design for a particular region. If the nest boxes are cleaned and maintained annually, barn owls can use them year after year. Monitoring barn owl nest boxes can accommodate short-term or long-term research questions.

Core Options

Field Methods

The field methods for nest box monitoring will vary based on the research questions being explored. Nest boxes can be censused if all nest box locations are known (farm-scale) to address fine-scale questions, or a subset of nest boxes can be sampled out of a larger group of boxes (regional-scale).



In regions where barn owl boxes are not already constructed, resources are available to start a nest box program (Marti et al. 1979, Raid 2012, Wade et al. 2012). In regions where farmers or researchers already have constructed nest boxes, it is possible to begin occupancy and demography studies by conducting regular nest box checks (Hafidzi et al. 1999, Watts and Whalen 2004, Wingert 2005, Meyrom et al. 2009, Charter et al. 2010, Wendt and Johnson 2017). In the initial stages of a nest box monitoring program, it is especially important to collect descriptive data for each nest box, particularly GPS coordinates, height off the ground, construction material, direction and size of opening, dimensions, and condition (Meyrom et al. 2009, Charter et al. 2010b, Charter et al. 2012, Lambrechts et al. 2012, Wendt and Johnson 2017).

The ecological question central to the study will determine the methodology for nest box monitoring. In studies concerned with occupancy, the primary methods are to manually check each nest box by physically looking inside (Meyrom et al. 2009), or by using a camera attached an extendable pole to see inside (Martin et al. 2010, Wendt and Johnson, 2017). There are potential advantages and disadvantages to these two options, including the level of disturbance, effort, and accuracy. While using a camera may result in less disturbance, it can allow for adults to flush from the nest box during the day, and it may be difficult to accurately count the number of offspring. Physically looking inside the box ensures that researchers can accurately count the young, but it requires more effort to climb up and access the box and in some cases nest boxes may be inaccessible. In addition, manually checking boxes seems to be less disturbing for the owls at or after dusk, and if owls do flush during this time they will not be harassed and attacked by daytime raptors and corvids (Hidmarch pers. oberv.). More research is needed to determine which method causes the least disturbance and whether there is a significant difference in accuracy between the two methods. Researchers should consider time constraints, level of disturbance, and importance of accurate estimates when choosing between these methods.

In many cases, it is useful to collect breeding data including egg laying date (can be back calculated using wing length of nestlings to determine age (Roulin 2004), clutch size, number of nestlings (especially at key stages such as hatching and fledging), and age of young (Johnson 1994, Meek et al. 2009, Meyrom et al. 2009, Martin et al. 2010, Hindmarch et al. 2014). If breeding data is being collected, the nest box should be visited repeatedly to determine the fate of the nest, but care should be taken to disturb the nest as little as possible (Martin et al. 2010, Hindmarch et al. 2014, Wendt and Johnson 2017).

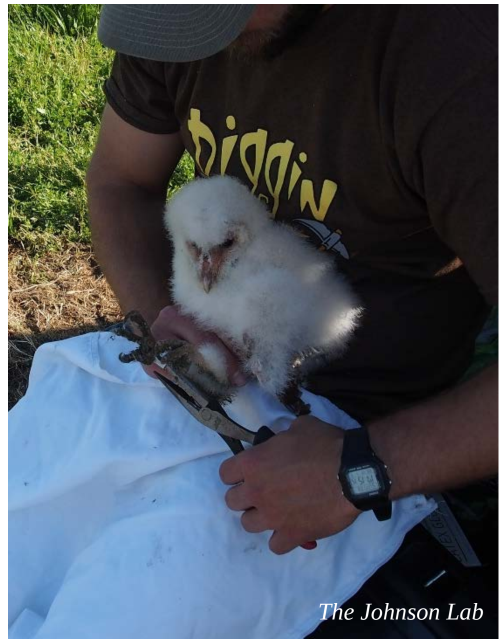
Complementary data

In addition to the basic monitoring methodologies described above, complementary data can be essential for answering more complex questions and for the discovery of new natural history traits.

Many studies collect pellets egested inside or outside the nest box as an index for occupancy or as complementary data on the diets of nesting barn owls, in some cases linking these data to reproductive success (e.g., Charter et al. 2015). Pellet dissection can be used to determine whether owls are removing pests or other prey (Moore et al. 1998, Meek et al. 2009).

Whenever pellets are collected, data on the time of collection, quantity, and location should be recorded.

Banding of adults and nestlings in each nest box can also reveal more complex details about barn owl demography, survival, and dispersal (Altwegg et al. 2006, Meyrom et al. 2009). Whenever owls are banded, researchers can use this opportunity to take measurements including, but not limited to, bill length, wing chord, weight, body coloration and molt, and breeding condition. Barn owl chicks can be aged by feather growth, weight, and wing length before fledging (Taylor 1993, Roulin 2004, The Barn Owl Trust 2015). The degree of whiteness on primary feathers can be used in certain regions to determine the age of adult barn owls as first year, second year, or after second year (Taylor 1993), though the reliability of this should be investigated further (Charter pers. obs.). Recaptures and recoveries of banded owls will help researchers answer questions related to breeding success, movement, and more.



In an applied context, an important element of complementary data is a GIS analysis of the surrounding landscape. Many studies have used this information to determine the habitat features that will best predict nest box occupancy and breeding success and can be used to determine optimal nest box placement (Martinez and Zubergoitia 2004, Meek et al. 2009, Frey et al. 2011, Naim et al. 2011, Hindmarch et al. 2012, Hindmarch et al. 2014, Kross et al. 2016, Wendt and Johnson 2017). Although nest boxes may be placed in desirable locations for owls, in fast changing agricultural settings these nest boxes may actually be ecological traps resulting in poor nest success.

Short-term vs. long-term studies

Barn owl nest boxes are conducive to both short-term and long-term studies. The methods described above can be used for studies over various timelines, though demography studies may require more time to collect data on a sufficient number of individuals.

Areas in need of refinement; challenges & considerations

Focus for refinement should be placed on standardization of field and statistical methods.

Challenges and considerations

In any nest box monitoring program, the most important consideration is disturbance to the nest. Care should be taken at all times to keep disturbance to a minimum and reduce risk of nest abandonment. Nests are especially sensitive during laying and incubation so special care should be taken at this time (Martin et al., 2010). More research is needed to determine which methods cause the least disturbance.

Cost of equipment and nest construction can also be a challenge in nest box programs. In addition, height and location of the nest box can be a challenge for researchers. Access to the box may be limited and safety equipment can be necessary and costly. This should be considered when installing the nest box, or when choosing a ladder and other equipment to use for monitoring.

All of these methods will be most successful when they are agreed upon from the Principal Investigator to the coordinator to manager. Barn owl researchers should work together to agree on methods and achieve standardization within and among studies.

Recommendations

Our recommendations for researchers are to agree on methods within a study team. One of the most important considerations is to minimize disturbance to decrease the risk of nest abandonment. Data should also be standardized within a study and ideally in agreement with other similar studies. When establishing a monitoring program, it is essential to map nest box locations and use a consistent code to refer to each box. Lastly, occupancy data should be relayed back to landowners so results can be applied in a way that achieves optimal nest box placement.

2) Pellet Analyses

Applications of the method


The Barn Owl Trust

The egestion of pellets by owls provides a unique method to address diet related questions (Dodson and Wexler 1979). Pellet analyses can answer questions pertaining to prey species composition, differential prey selection, minimum number of prey items, and estimates of prey mass, age, and sex (Colvin and McLean 1986). In some settings, studies have suggested that barn owl diets can provide an index for local small mammal communities (Hanney 1962, Glue 1971). Although, this suggestion assumes that barn owls hunt at random and that pellets provide a random sample, both of which are difficult to test in the field (Yom-Tov and Wool 1997).

For barn owls, nest boxes allow researchers to conduct more in-depth studies with greater sample sizes both across time and in various habitats (Colvin and McLean 1986, Charter et al. 2007). Data for barn owl diets from pellets are extensive and various forms of agriculture in different biomes are represented globally. Although methods for processing pellets vary, most pellet analyses involve dissecting egested pellets and using skulls, mandibles, femurs, and dentition to identify prey items. Here we discuss common methods and considerations for conducting pellet analyses and some of the strengths and limitations of using pellets to assess barn owl diets.

Core Options in Field Methods

Pellet Collection

Pellet analyses require three steps; pellet collection, pellet processing, and prey identification. Pellets are normally collected from inside the nest box, from the ground immediately surrounding the nesting site, or from roosting sites. Fresh pellets remain moist and may have a sheen for up to two days. Undisturbed pellets can remain intact for almost two years (Barn Owl Trust 2015). Therefore, it is difficult to reliably age barn owl pellets without prior visits to collect sites. To guarantee pellets were from a given time frame, nest boxes can be entirely cleared and all pellets collected at a predetermined date in order to provide an estimate of total prey items over that time period (Charter et al. 2009). It is best when collecting large numbers of pellets to wear gloves and masks to prevent exposure to airborne pathogens such as hantavirus.

Pellet Processing

The first step in processing owl pellets is to sterilize and prepare the pellets for dissection. For sterilization, it is common to bake the pellets for 4 hours at 200 ℃. After this, pellets can be weighed and measured. Pellets can be dissected by hand at this point or for greater ease, pellets can be soaked individually or all together in water or in a 1:10 mixture of sodium hydroxide (NaOH) and water or other cleaning agent (Roth and Powers 1979, Bonvicino & Bezerra 2003, Charter et al. 2009, Marti et al. 2015). This will facilitate the separation of bones from fur and feathers and help clean any impurities from the surface of the bones without damaging bones or chitin-based insect segments. Remaining fur and feathers can be discarded and bones kept for further analysis.

Prey Identification

Skulls, femurs, and dentition from pellets allow researchers to identify prey items to genus and often to the species level. Resources for rodent and small mammal identification are plentiful. For many species, characteristics of the skull, femur, and pelvis bones can provide estimates for body mass and sex of the prey species (Dunmire 1955, Trejo and Guthmann 2003, Balčiauskas and Balčiauskienė 2011). For non-mammalian prey, skulls and mandibles can identify prey items to family and sometimes genus level (Glue 1974). To best organize large samples of pellets, analogous bones from different pellets should be grouped accordingly (Marti et al. 2015). A minimum number of prey items can be quantified by counting the number of any one individual bone in all pellets (e.g. right mandible, left femur).

Areas in need of refinement; challenges and considerations

Although pellet dissection is a straightforward and well-documented way of assessing barn owl diets, it does come with some limitations. First, assessing the total number of prey items consumed is an imperfect method. Barn owls do not egest all pellets within the nest box and adults will often tear apart prey items before delivering them to nestlings. Therefore, potentially important bones and skulls may be missing from pellets. Further, it is difficult to assess at a fine temporal scale when pellets were egested. For pellets collected from within the nest box, growing nestlings often trample on pellets making collection of individual pellets challenging, however, summary data on all pellets is still possible. Coupling pellet analyses with other methods such as occupancy monitoring or video analysis can help validate estimates of number of prey items and identification of prey from pellet analyses.

Recommendations

Pellet analyses have historically been a convenient and reliable method for assessing barn owl diet. They will continue to be an easy and inexpensive method for assessing barn owl diets and particularly informative in agricultural settings to determine if barn owls are consuming target species. We recommend that researchers practice the nest box clearing method so as to be certain of when pellets were egested. For standardization and sterilization, we recommend baking pellets for 4 hours at a minimum of 200 °C. Soaking pellets in a water and NaOH solution will allow for easiest separation of bones from fur. Last, we recommend using dentition to best identify prey items, supplemented by measurements of the skull, pelvis, and femurs.

Pellet analyses are likely most informative when coupled with other research methods and provide an easy form of validation for other diet related questions. Otherwise, there is merit in collecting and assessing owl pellets, regardless of sampling scheme, if only for summary data.

3) Toxicology

Applications of the method

Methods to study toxicology can reveal the exposure and concentration of anticoagulant and other rodenticides in barn owls. This can help answer questions related to anticoagulant rodenticide application and its effects on individuals and possibly populations. With the careful consideration of exposure rate, concentration, and timing and location of rodenticide application, studies using these methods can elucidate practical information for land managers. The risk of rodenticide exposure of barn owls and how this varies depending on landscape composition and rodenticide application methods needs to be addressed in order to determine whether barn owls and rodenticides can be used concurrently in an IPM approach.

Core options

The core options for measuring toxicology involve necropsies and various screening methods. Tissue samples can come from liver, blood, pellets, feces, or whole carcass residues (Primus et al. 2001, Christensen et al. 2012, Hughes et al. 2013, Gómez-Ramírez 2014, Ruiz-Suárez et al. 2014, Geduhn et al. 2016, Martínez-Padilla et al. 2017). Other methodology for screening can include coagulation assays, immune response, genome screening, and liver enzymes. Because rodenticides may not be detected in all tissue samples, an alternative method is to test prey samples (Geduhn et al. 2016), but this will not directly reveal the secondary exposure levels in owls.

The likelihood of detecting rodenticide residues depends on the tissue sample collected and the type and concentration(s) of rodenticide compound(s) present in the specimen. Though blood is easy to collect and can come from a live animal, the concentrations of rodenticides are more variable and there is currently no test for anticoagulant rodenticides in blood for birds (Booth and Wright 2016, NYS Department of Environmental Conservation and Cornell Wildlife Health Lab). Liver tissue is most commonly used for assessing anticoagulant rodenticide exposure due to the long half-life of rodenticide compounds in the liver (1-12 month) However, it does require dead animals for testing (Gray et al. 1994a, Albert et al. 2010). Pellets are likely the easiest and non-invasive samples to collect from barn owls, but they complicated by uncertainty between what was egested in the pellet and what was ingested by the owl (Gray et al. 1994b, Eadsworth et al. 1996). In all cases, it is important to consider the half-life of different rodenticides and how long they are likely to be present in certain tissues.

Behavioral studies have involved the assessment of live captive and free-ranging barn owls' response to consuming rodenticide laden prey. These studies have measured barn owl behavior (Salim et al. 2014), physical responses (Mendenhall and Pank 1980, Salim et al. 2014), and breeding success (Naim et al. 2010, Naim et al. 2011) under treatment and control conditions. Measurements of breeding success include clutch size, nestling growth, and fledging success (Naim et al. 2010, Naim et al. 2011). Studies of live owls can include blood samples taken from the brachial, jugular, or metatarsal vein.

For field experiments, a measure of rodenticide use should be included, either from surveying farmers or obtained from other records of rodenticide use (Brown et al. 2006, Hughes et al. 2013, Geduhn et al. 2015). By including the rodenticide compound, timing, and concentration of rodenticide applied, studies can help determine the temporal effects of rodenticides on potential secondary exposure to non-target species (Coeurdassier et al. 2014). Farm surveys can also reveal locations of bait application sites, which can be used to calculate the likelihood of secondary poisoning through GIS analysis (Hindmarch et al. 2017).

Whether conducting a lab experiment or a field study, consideration should be given to the most effective sampling method to determine exposure in owls and associated prey. Studies can be repeated spatially, temporally, and seasonally. Studies involving necropsies of barn owls and prey species may contain spatial biases with regard to specimen collection; carcasses are more likely to be detected near areas of human habitation and may have higher levels of rodenticides because of this close proximity to humans. The design of the overall study will depend on the questions being asked.

Areas in need of refinement; challenges & considerations

Focus for refinement should be placed on the need for "safe" IPM guidelines which incorporate the use of both owls and rodenticides. In order to determine this, future studies should, in addition to sampling barn owls for rodenticides, also assess exposure rates in target and non-target rodent species on which barn owls prey (Hindmarch and Elliot 2015). To date, there is little information available on species-specific threshold levels, identifying sub-lethal impacts, as well as the effects of digesting multiple compounds. Furthermore, carcass collection, and prey sampling are all specific methodological areas that would benefit from further refinement.

Challenges and considerations

Several aspects of detecting and diagnosing rodenticide exposure and poisoning create challenges for understanding effects. For example, evidence of poisoning in carcasses may be more difficult to detect than other causes of mortality, requiring specific diagnostic testing that is expensive and not always available. Exposure to rodenticides may also cause an individual to be more susceptible to other risks, requiring distinguishing proximate from ultimate causation of mortality. Poisoning diagnoses can also be complicated by the “cocktail effect” of exposure to multiple compounds, which is often the case for raptors inhabiting agricultural and urban landscapes.

In any study of toxicology, some of the primary challenges are cost and sample size. Consideration should also be given to the sampling strategy and whether it is biased or unbiased. Carcasses retrieved opportunistically are often biased spatially because they are more likely to be found near human habitation, where rodenticide use may be disproportionately high. Retrieved carcasses also do not provide any information on the proportion of the population impacted, and ultimately if these losses impact owl population levels and subsequent rodent control in a given area. Each type of tissue sample has advantages and disadvantages, so care should be taken to understand the sensitivity of a tissue sample to testing for exposure, as well as how invasive sample collection will be.

When analyzing toxicology samples, the meaning of exposure and the concentration of rodenticide detected should also be well defined. It can be challenging to determine which is worthwhile to measure in a particular study system and how to interpret results. The questions being explored in each specific study will determine whether the goal is to estimate exposure, concentration, or both.

Thought should also be given to other toxicants and diseases that may be influencing a study population. Fungicides, neonicotinoids, avian influenza, trichomoniasis, and West Nile virus can also potentially have effects. Researchers should consider the effects of these in addition to anticoagulant rodenticides.

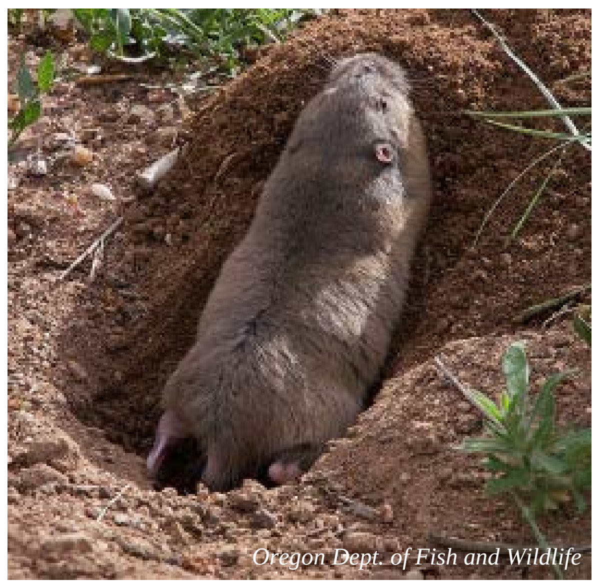
Recommendations

The preferred method for a particular study will depend on the questions being explored and the resources available. In studies which involve blood sampling, the researcher should decide if it is most appropriate to take blood from the brachial vein, jugular vein, or foot. Alternatively, blood can be sourced from the heart or body cavity of a deceased owl. Necropsies can be particularly useful to screen for diseases and toxicology. When collecting samples of live animals or carcasses, a systematic approach should be used, whether the study site is baited or unbaited. For bait stations, information regarding the bait used, timing and spacing of bait should be collected from the farmer. It is also important to collect spatial data of barn owl nest and roost sites. Further attention should be dedicated to assessing sub-lethal effects of rodenticide exposure on barn owls, particularly with regard to fecundity, resource acquisition, and space use. There are many methods available for toxicology studies and the most appropriate methods should be determined based on the goal of the study. However, all studies should use a systematic approach to baiting and sampling to achieve best results.

4) Rodent Monitoring

Applications of the method

Any assessment of the potential benefits of barn owls for reducing rodent damage must inherently rely on estimates of changes in rodent population size or subsequent rodent damage. There are a variety of monitoring tools available for use; the most appropriate strategy will depend on the stated goals of a project, as well as available resources for monitoring. Most rodent monitoring strategies will rely on one of three basic approaches: 1) a general index of abundance, 2) an assessment of population size, or 3) an assessment of occupancy. Specific details associated with these monitoring tools are outlined below, but in general, they allow the land manager, researcher, or agency personnel to monitor either changes in trends in rodent numbers or actual densities of rodents over time. This approach assumes that a reduction in rodent activity or population size will be directly reflected in a reduction in rodent damage. This is often the case, but may not tell the whole story. For example, some forage crops are able to compensate for limited damage by increasing production from remaining tissues or adjacent plants (McNaughton 1981). Alternatively, increased predation in an area may reduce rodent foraging and subsequent reproductive output given the ecology of fear (Brown and Kotler 2004). In both of these situations, simply monitoring changes in rodent numbers may not tell the whole story of how predation can impact losses in crop production. It is important to remember that a reduction in rodent damage is often the ultimate goal, rather than a reduction in rodent numbers. As such, techniques for monitoring temporal changes in rodent damage to cropping systems are sometimes preferred. However, in some cases growers have a “zero tolerance” of rodent presence because of risk of rodent feces or animals themselves ending up in the farm product itself. The following sections provide more detail on monitoring for rodent activity, density, and subsequent rodent damage to provide the opportunity for more robust sampling strategies when researching the impacts of barn owls on rodent populations, and allow land managers to assess potential benefits from rodent predation by barn owls on their property.


Oregon Dept. of Fish and Wildlife

Core options

Researchers often focus on changes in rodent activity, population size, or occupancy when determining the efficacy of a management approach. These approaches can provide additional information compared to damage estimates by potentially identifying species causing damage, additional species present in the area, and demographic information and activity patterns associated with target species. Such information can be critical for developing a better understanding of predator-prey dynamics. To that end, rodent activity is often assessed via a general indexing approach (Engeman 2005), which allows the user to utilize a variety of monitoring tools to assess changes in rodent activity over time.

General indices are frequently preferred given that they are practical to implement, are sensitive to changes in population size, allow for development of precision and variance estimates, and have few assumptions (Engeman 2005, Engeman and Whisson 2006). They also allow the user to select between a binary or continuous response in assessing changes in rodent activity. Continuous response rates have generally tested better than binary responses, but this can vary (Engeman and Whisson 2006). Examples of general indexing approaches include catch per unit effort via live (Sellers et al. 2018) and kill (Theuerkauf et al. 2011) trapping, remote-triggered cameras (Baldwin et al. 2014), tracking stations (Quy et al. 1993), chewing indices (Engeman et al. 2016), visual counts (Fagerstone 1984), and measures of mound/burrow activity (Engeman et al. 1993; see Engeman and Whisson 2006 for additional examples of indexing approaches). Although all of these approaches are sensitive to changes in population size over time, not all provide the same information. For example, live trapping allows the user to identify species present, as well as other demographic data which can be useful for monitoring the effectiveness of management programs. Species identification can sometimes be collected via other monitoring approaches (e.g., species identification from cameras or tracking plates), but will depend on species composition in a given area; if sympatric species are similar in appearance and size, identification to species is difficult, if not impossible. Other approaches (e.g., chewing indices) rarely allow for species identification, and few of the non-trapping approaches allow for easy identification of demographic data. That said, live trapping is rather time-consuming and requires handling of live animals; live trapping is generally not practical for land managers, which is an important consideration given that land managers should be able to quickly employ monitoring tools for easy assessment of the current status of pest populations. Lastly, it bears noting that general indices should be calibrated against rodent numbers (e.g., minimum number known estimates; Baldwin et al. 2014, Engeman et al. 2016)

before full-scale implementation as a monitoring tool. For the index to function appropriately, it needs to reflect actual changes in rodent numbers over time (Engeman and Whisson 2006). This requires foresight into the development of the index prior to initiation of a monitoring program.

In some settings, it is more practical to measure rodent damage rather than use other methods. This approach is commonly employed in rice where damage to tillers is calculated along transects or quadrats at stratified distances (e.g., Hafidzi and Mohd 2003, Singleton et al. 2005). Similar approaches are employed in nut (e.g., White et al. 1998) and vegetable crops (Advani and Mathur 1982) where damage to the respective crop is documented at set intervals throughout the cropping system. This approach is also incorporated into IPM programs for vole control in artichokes, where growers apply rodenticides when vole damage exceeds a defined threshold (R. Baldwin, University of California, Davis, unpublished data). As previously defined, measuring rodent damage to cropping systems has the advantage of providing feedback on the indicator of greatest interest (i.e., the amount of damage caused by rodents). However, this approach can sometimes be more time-consuming than monitoring for rodent activity. Also, rodents do not always cause the same level of damage throughout the year. If damage appears to be minimal during a given sampling interval, land managers may decide that rodent management is not currently warranted. However, this approach could allow rodent populations to build to large numbers. Over time, they may switch to feeding on the crop, thereby causing substantial damage in a short period of time. User experience will be needed to determine if the monitoring of crop damage is the best strategy for determining the potential for rodent damage in that particular cropping system.

Although general index values are very useful in monitoring changes in rodent activity over time, there are some questions that are better answered through the estimation of population size. Density estimators allow for direct comparison of rodent numbers before and after treatment applications, as well as comparisons across space and time. Density estimates are also easier to conceptualize when compared to general index values (e.g., reduction in vole density from 100 to 50 voles/unit area vs. a reduction in catch per unit effort of 1 capture per 5 trap nights to 1 capture per 10 trap nights). However, density estimates generally require greater effort for data collection, making this approach impractical in some settings. Density estimators also have a more stringent set of assumptions, with some studies suggesting few instances where data requirements for proper density estimation are met (McKelvey and Pearson 2001). It also bears noting that land managers will not be able to employ density estimators when tracking changes in population status of rodents over time given the advanced methodology and capture effort required by these approaches.

Traditional density estimates for rodents have focused on mark-recapture models. A variety of models are available depending on local circumstances (see Williams et al. 2002 for overview on options). The general approach entails live-trapping over several-to-many days to individually mark animals. A population estimate is then derived based on the ratio of marked to unmarked individuals over the duration of the trapping period. Density estimates are then established by relating the population estimate to the area sampled. However, it can be difficult to estimate the area sampled given unknown movement patterns of the sampled population. Spatial capture-recapture models circumvent this limitation by including location data on captures to develop detection probability, and subsequently density estimates, without having to rely on an arbitrary delineation of the sample area (Royle et al. 2014). Spatial capture-recapture models are increasing in use, and may have utility in certain settings (e.g., Berl et al. 2018).


John Van Niel

Mark-recapture and general index approaches allow for the observation of individuals, thereby asserting their presence at a given location. However, a lack of observation does not imply their absence. A "zero" observation can be a result of two mutually exclusive events, either that individuals are not present at that given location, or alternatively, they are present, but they were not captured, photographed, or left any signs of activity. To overcome this problem, short-interval repeated monitoring can be used. Such monitoring schemes facilitate teasing apart the probability of detection vs. the probability of presence of individuals within a site to determine occupancy (MacKenzie et al. 2002). Various programs such as PRESENCE (MacKenzie et al. 2002) allow for the development of occupancy models that can be used to more accurately assess the presence of individuals. Furthermore, extensions of this approach may be used to assess the abundance of individuals. Thus, going beyond the binary measure of presence-absence, one can estimate abundance levels. For example, N-mixture models (Royle 2004) provide robust statistical methods to infer population sizes based on repeated samplings. Like mark-recapture approaches, occupancy modeling and N-mixture models require increased sampling effort and time to collect sufficient amounts of data for analysis, again making them unsuitable for use by most land managers.

Regardless of the monitoring tool, both transect and grid sampling designs can be effectively used to monitor rodent populations and damage. Generally, transects or grids are set up following a stratified design to best represent the conditions throughout the sample area (see Hopkins and Kennedy 2004, Baldwin et al. 2014, Engeman et al. 2016, Hafidzi and Mohd 2003, Singleton et al. 2005 for both transect and grid examples). These transects or grids are usually combined in a randomized complete block design with a control plot to ensure that results are robust. Alternatively, a before-and-after monitoring approach could be used to more definitively assess the impact of a management strategy on rodent numbers and subsequent damage (Labuschagne et al. 2016). However, the use of control plots or monitoring before and after treatment are not always possible for a variety of reasons (e.g., lack of desire by land managers to leave rodent populations unchecked, lack of sites without natural predators to serve as controls when determining efficacy of natural predators, insufficient funds to include control sites). In lieu of such a strategy, extensive spatial and temporal replication will be needed (likely via a meta-analysis) to better control for natural changes in rodent population size or activity.

One final key to assess the effectiveness of any management tool is proper replication. This applies both spatially and temporally. For example, burrow fumigants tend to be far more effective in moist soils, yet moist soil conditions often vary throughout the year (Proulx et al. 2011, Baldwin et al. 2017). Likewise, raptor diet composition and the number of rodents consumed by a breeding pair can vary throughout the year (Browning et al. 2017 Kross et al. 2016) and across years (Salamolard et al. 2000, Paz et al. 2013) given the potential impact of varying climatic conditions, disease, and other factors on rodent population size and composition. These potential temporal differences must be accounted for to better understand the impact of the selected management tool on rodent populations long-term (Labuschagne et al. 2016). The same applies to spatial replication. Just because a management tool works in one area does not mean that similar results will be widespread. Ultimately, much thought must go into any assessment of efficacy of rodent management tools and strategies.

Areas in need of refinement; challenges & considerations

Methods for rodent monitoring require extensive effort and expertise. One area of focus for refinement should be placed on methods that make rodent monitoring more accessible to the landowner. Special focus can be directed towards determining whether measuring rodent damage is an effective means of indexing rodent population numbers, and more importantly, if these measures of damage can discern potential reductions in rodent damage from applied management actions. Advances in technology including digital trackpads, artificial intelligence, game cameras, DNA-recovery techniques, satellite imagery, and thermal imagery may be able to overcome many of the current limitations if applied to rodent monitoring. For example, researchers are currently examining the use of drones to assess crop damage efficiently and accurately (Malkinson, unpubl.). With current available methods, each crop system will have its own best practices for rodent monitoring, meaning regional studies are needed for each system where barn owls are used for rodent control.

Challenges and considerations

Further refinement of the methods described above will require a large-scale study, thus practicality and available resources will be a limiting factor. Consideration should be given to the expertise necessary to monitor rodents and rodent damage when using these methods. Where methodology is more advanced, or an extreme amount of effort is necessary, it may be beyond the abilities of a land manager to utilize these methods. Ultimately, it will be necessary to consider whether the goal of a monitoring approach is to determine a reduction in rodent activity, a reduction in rodent damage, or a reduction in actual rodent numbers determined via a population estimate. These factors will drive the level of sophistication of the monitoring strategy, as well as the resources required to implement the monitoring program.

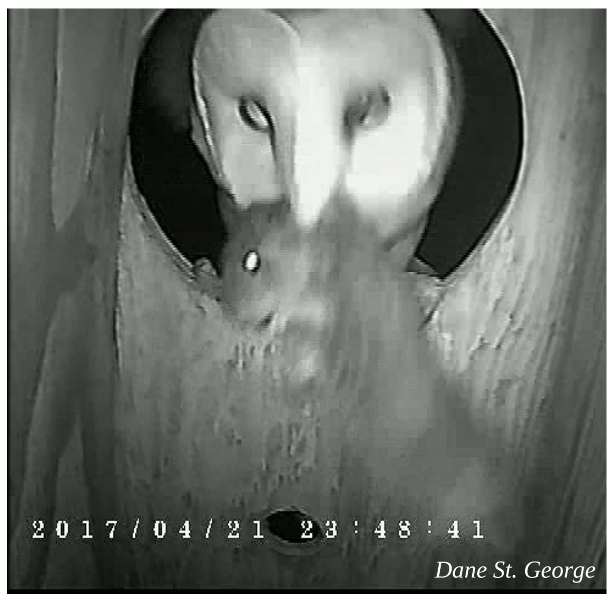
Recommendations

Given the variability in rodent populations within and among crop systems, studies should be customized to the crop, season, species, and resources available for monitoring changes to rodent populations. Keep in mind that many methods will require calibration to a population index, and that proper study design should include consideration of temporal and spatial replication and controls. All monitoring designs require sufficient expertise and resources, so these should be considered before choosing a monitoring method.

5) Video Analyses

Applications of the method

Nest box cameras offer a minimally invasive method to observe various nesting behaviors of barn owls (Taylor 1991). Nest box cameras are used to assess nest success, number and rate of prey deliveries, prey species composition, resource distribution, and nestling survival (Currie et al. 1996, Radford et al. 2001, Steen 2009, Browning et al. 2017). Using these videos, we can gather data on fundamental phenological questions such as the timing of the nesting period and nest box use, fecundity of individuals and communities of barn owls, as well as produce an index of rodent removal in agricultural landscapes (Roulin and Bersier 2007). Further, the affordability, and feasibility of using nest box cameras allows ecologists to observe many nest boxes over greater temporal scales (Bolton et al. 2007).



Dane St. George

Core Options in Field Methods

Nest box cameras require three principle components: camera, recording device, and power source. Among these three components, there are near infinite combinations of models regarding quality, price, convenience, and performance; however, a review of features and models is beyond the scope of this report. Instead, to aid those considering or embarking on a project involving nest box cameras, we offer primary considerations when selecting and deploying a camera, recording device and power source.

Cameras

The model and design of the camera are the first considerations when designing a study and will be influenced by the research question. For cameras mounted inside the nest box, the camera should be small enough to cause minimal interruption to the movement and behavior of the owls. Individual cameras should have durable housing so that an owl cannot easily break or move the camera. To ensure adequate night vision, cameras should be equipped or supplemented with infrared light emitting diodes (LEDs). However, some infrared LEDs may need to be masked if the subject becomes “washed out” as it gets close to the camera, and models should be chosen that illuminate adequately while minimizing disturbance to the owls. Similarly, a large field of view is important if subjects are in close proximity to the camera and can be achieved with a wide-angle lens.

The location of the camera will also depend on the nature of the research question. Exterior mounted cameras can capture more behaviors of adults coming to and leaving the box but capture little with regard to nestling behavior within the box, though they capture nestling behavior outside the box (e.g., Charter et al. 2018), which interior cameras miss. Interior cameras can capture chick behavior and some adult behaviors but may be confounded by obstructions of the camera by chicks depending on box design. Selecting nest boxes that are deeper and placing the camera higher in the box can help alleviate this.

When selecting camera resolution, several trade-offs exist. If a study requires great detail in images (i.e. individual identification or identification of small prey items), a camera with high resolution will require a greater power supply, need more storage space, and will likely cost more. Recent studies employing nest cameras provide details on their models and pricing, such as Prinz et al. (2016), Zárybnická et al. (2016), and St. George (2018). Costs range widely, depending on features, and can be as low as $12 US to as high as $500 US per camera. For studies in which photos are the more pragmatic solution for the research question, see Rovero et al. (2013) for recommendations on specifications for camera traps (aka trail cameras).

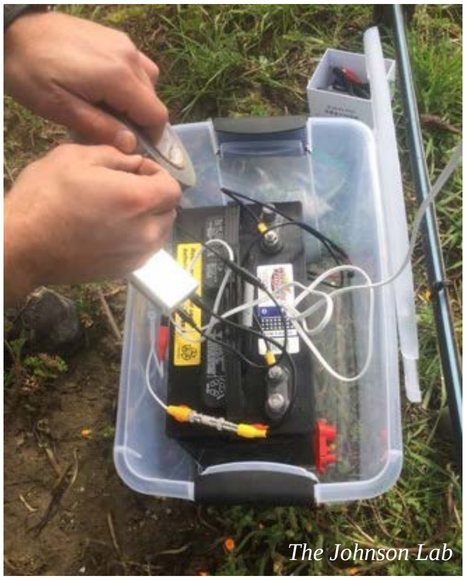
Recording Device

Advances in digital video technology have resulted in a diversity of models, functions, and costs of recording devices. Again, technical settings for recording devices should be catered to the needs of the research. Motion detection is ideal if reducing data storage is a main priority but may require some adjusting of sensitivity to reduce false triggers. Identifying the mechanics of the motion detection of the device is important and should be tested before deploying equipment. For many all-in-one devices such as game or trail cameras, “motion” is detected by infrared sensors which triggers the camera to begin recording images or video onto an external memory device (See Welbourne et al. 2016 for more details on mechanisms). This helps prolong battery life, but often results in a small delay between a motion event and the beginning of a recording, which could be problematic for cases in which the researcher is trying to identify prey items or the moment of prey delivery, which can be brief.

Systems with separate recording devices function differently; recording devices receive a constant digital video signal from a camera which consists of numeric values for each individual pixel. The recording device will store pixel values for several seconds and rewrite them if no change occurs between frames. If a motion event occurs and a certain threshold of pixel value change is exceeded, the recording device will create a video file beginning a few seconds prior to the motion event, thus eliminating any delay between a motion event and the start of a recording. These recording devices often allow users to "mask" parts of the frame so that non-target motion (i.e. stirring nestlings) will not trigger a recording.

Most modern digital video recorders (DVRs) allow users to adjust additional settings; Video recording can be set to specific times, resolution can be increased or decreased, videos can record in various file types (e.g. MP4, ASF, AVI), date and timestamps can be added, file naming can be specified, and so on. The simplest models are usually equipped with adequate functionalities for research purposes and can be purchased for less than $100 US.

Power Source



Since most nest boxes are too far from stationary alternating current (AC) power, electronics must be powered by large direct current (DC) power batteries. Photovoltaic power generally could be employed at the site of a box, but most relevant camera data will occur during the night, again requiring storing power in batteries. Most 12 Volt batteries will be adequate for the power that a camera and DVR require. These batteries can be very heavy to carry, so consider how remote the nest box is before selecting one. It is always best to test the energy consumption of the system so over-discharging batteries can be avoided (over discharging 12V batteries can permanently damage them). Depending on how much video is being recorded per night and the battery capacity, a single charge can range from 2-3 days to 3-4 weeks. For further details on energy demands of cameras and DVRs, see Bolton et. al. (2007).

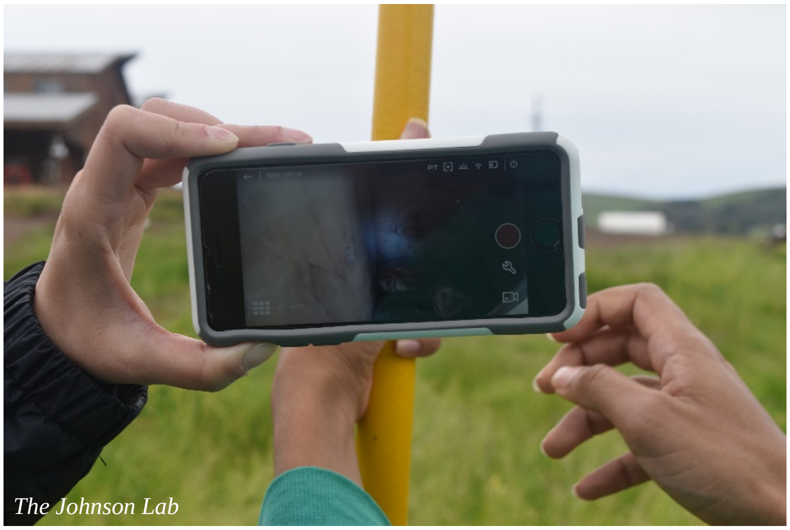
For longer studies, a system for recharging batteries should be established. If budgets and facilities permit, photovoltaic solar panels provide the lowest maintenance energy supply. However, if solar panels are not feasible, batteries must be replaced and recharged as needed. Many devices exist that use AC power to charge batteries and can be purchased for less than $50 US.

Areas in need of refinement; challenges & considerations

While video analyses offer unique opportunities to observe barn owl behavior, it is important to acknowledge shortcomings of the methodology. Like any method involving electric components, the risk of technical failures or malfunction is ever present. Nest box camera studies are also limited to the breeding season because owls do not use nest boxes much outside the breeding season with the exception of occasional roosting (Marti 1979). Identifying prey species is less reliable via video than the typical method of barn owl pellet dissection. Estimating the number of prey items delivered to chicks may also be less reliable than pellet analyses due to the obstruction of the camera view by nestlings, although this is yet to be rigorously tested. Improving the energy efficiency of video systems will be important in making video studies more logistically and financially feasible. Additionally, finding ways to reduce the amount of storage space needed without compromising the quality of video will be another challenge for researchers moving forward.

Recommendations

Technical Recommendations



There is no equipment that will fit all studies perfectly; however, our review suggests that small closed-circuit television (CCTV) security cameras meet most requirements for video studies. They have durable housing, are easily mounted on nest boxes, have wide-angle lenses, come with infrared LEDs, are inexpensive (<$25 US) and widely available, and are compatible with most DVRs.

Portable, single channel DVRs that save to Secure Digital (SD) memory cards work well with security cameras, are inexpensive, easy to obtain, contain simple but sufficiently sophisticated programming, require low energy input, and can save to a variety of video formats. A standard 32 GB SD card can be purchased for $12 US. The memory capacity of SD cards has improved in recent years with some cards capable of storing 1 TB of data.

If accessibility and transportation are not major concerns, deep cycle marine batteries perform well. They are ideal for supplying gradual power over time, can be recharged via solar panels or commercial battery chargers, and hold a charge better than most comparable 12V batteries. They are similarly priced to other 12V batteries (~$100 US) and are widely available.

Weather proof plastic tubs to house batteries and recording devices may be necessary in some settings.

Sampling & Analyses

Like most studies in ecology, there are a myriad of questions to address when deciding how to sample. How many sites and which sites should be sampled? Is it best to capture a small temporal window across many nest boxes or entire seasons in fewer nest boxes? How should sample locations be spaced across time and space to best avoid autocorrelation of data?

Several response variables can be gathered from nest box cameras including visitation rate, frequency and rate of prey deliveries, prey species composition, time between deliveries, nest success, fledgling survival, and resource allocation, among others. Nest boxes allow researchers to make comparisons across many habitats both during the same breeding season and across years. Sampling schemes can be altered between years or repeated easily between years to yield more rigorous analyses.

Given financial, time, labor, and other logistical constraints, there are probably no sampling methods that will apply to all circumstances. Generally, we recommend that data be collected on individual nest boxes through the entire breeding season in order to capture behavioral variation due to energetic demands of raising nestlings. If there are many potential nest boxes in the study site, we recommend maximizing space between focal nests. Estimates for how far barn owls travel from the nest box vary from 1-9 km and may vary regionally (Taylor 1994, Castañeda unpub. Thesis, Charter pers. comm.). Coupling video studies with telemetry data may help validate the sampling method.

For reviewing video data, software such as MotionMeerkat (Weinstein 2015) allow for automated video review and helps cut down processing time by exporting frames with motion as images and skipping over frames with no activity. The software also allows for sensitivity adjustment. Scripts written in Python or other programming languages can also provide a way to return frames where motion was detected, thereby enabling faster review times.

6) Telemetry and Movement

Applications of the method

Telemetry and movement methodologies can be used to answer questions related to population dynamics, dispersal, migration, habitat selection, foraging success, exploratory movement, and home ranges. Answers to these questions will help increase our understanding of the pest control services that owls are able to provide to farmers by revealing how far owls travel within and between years and the types of habitats that they use for hunting. In addition, the technology involved in these methods can contribute to advances in bioinformatics, engineering, computer science, and statistics.

Core Options

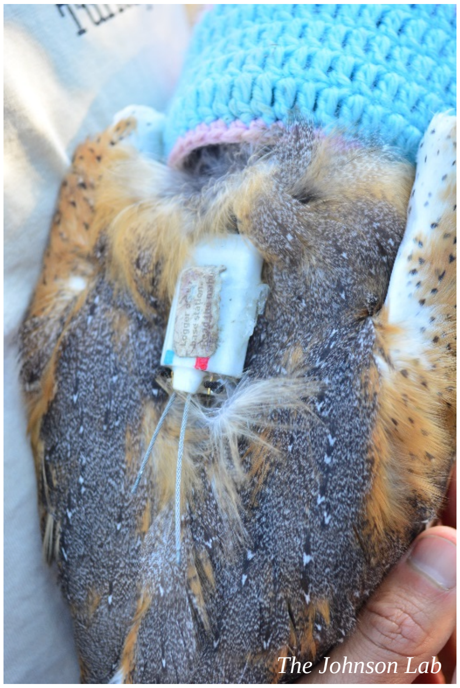
Field Methods

The primary method of capturing nesting barn owls is by hand through a door built into the nest box (Colvin and Hegdal 1986, Taylor 1991). Other options include mist nets (Hegdal et al. 1984), verbail traps (Stewart et al. 1945), bal-chatri traps (Hindmarch et al. 2017), and manually operated traps at nest box entrance (Massa et al. 2015). After capture, marking options include federally-issued aluminum bands (Stewart 1952, Henny 1969, Duffy 1992), color bands (Thomsen et al. 2014), and RFID.

To monitor movement, options include GPS (Massa et al. 2015), reverse GPS (Weiser et al. 2016), and VHF radio telemetry (Seel et al. 1983, Hegdal et al. 1984, Hafidzi et al. 2003, Thomsen et al. 2014, Hindmarch et al., 2017). The choice of technology is an important decision and there are strengths and weakness associated with each option (Latham et al. 2015). GPS utilizes satellite so does not require the construction of receiver stations, can be useful for large-scale questions, and can provide high resolution spatial data. Some weaknesses of GPS include limitations for data storage on the device, weight, battery life, and cost. Reverse GPS overcomes some these issues because it is smaller, does not store data on the device, can provide high resolution spatial data, and data retrieval does not require recovery of the device. However, reverse GPS requires the costly and logistically difficult construction of towers to receive signals and is limited to local populations.

After choosing a type of device to monitor movement, there are many options for attachment of the device. These include tail clips (Hegdal et al. 1984), loop harness (Thomsen et al. 2014), backpack harness (Seel et al. 1983, Hafidzi et al. 2003, Naim et al. 2012, Massa et al. 2015, Weiser et al. 2016), glue (Kenward 1985), and leg loop harness (Hindmarch et al. 2017).

The core options for data retrieval will depend on the choice of technology. For some archival technologies, recapture is required (Duffy 1992, Massa et al. 2015). Other methods allow data retrieval through a base station (Weiser et al. 2016, Castaneda unpubl. data), satellite (Massa et al. 2015), GSM (Gradev et al. 2011), or an antenna, either handheld, mounted on a vehicle, or another setup (Seel et al. 1983, Hegdal et al. 1984, Hafidzi et al. 2003, Thomsen et al. 2014, Hindmarch et al. 2017).



Analysis Methods

Data analysis is highly dependent on the chosen technology, scale of the data collected, and questions being asked. The following options have not all been used in the literature on barn owl movement but have the potential to be applied given the appropriate combination of technology, scale, and questions. For each method, we describe the appropriate application and limitations.

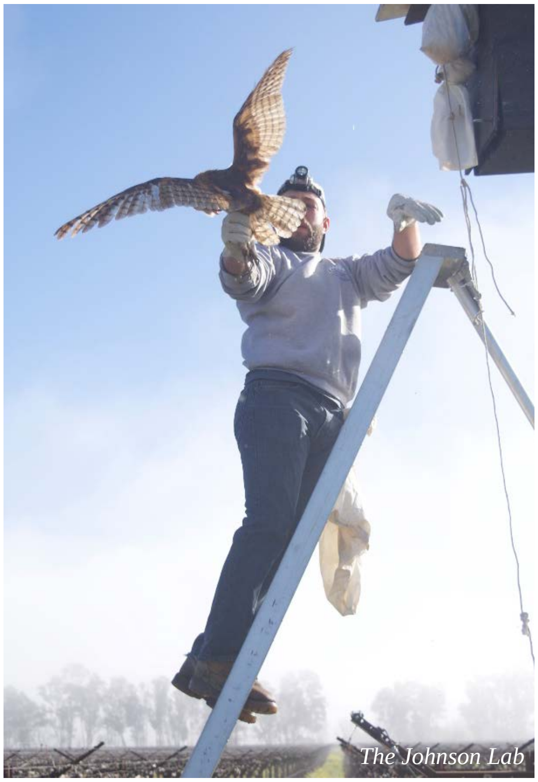
The most basic methods to estimate home ranges include minimum convex polygon (MCP) and kernel density estimates (KDE). MCPs uses observed locations to create a convex polygon that reflects the area used by an individual (Seaman and Powell 1996, Hemson et al. 2005). This method does not distinguish between locations visited frequently and infrequently by an individual (Seaman and Powell 1996). KDEs create a utilization distribution based on observed locations of an individual (Hemson et al. 2005). This method can be used to analyze the locations that are most densely visited by an individual. While basic, these methods are still used often to estimate home ranges.

A more statistically advanced home range analysis can be done using Brownian bridge (BBMM) and Dynamic Brownian bridge movement models (dBBMM). BBMM is used to estimate home range size (Bullard 1999), determine migration routes, and estimate movement paths of individuals as it relates to fine-scale resource selection (Horne et al. 2007). BBMM incorporates temporal structure of wildlife tracking data and models movements by including the sequence of locations and amount of time spent between locations, which improves utilization distribution statistics (Bullard 1999, Horne et al. 2007, Kranstauber et al. 2012). BBMM assumes animal movement patterns within a single track follow one constant property that defines the variance of the Brownian motion, a measure of the irregularities in an animal’s path. This can be problematic because animals change their distinct behavioral movement patterns as it relates to foraging, resting, or even breeding and migration on a greater time scale, which may result in biases in the variance of Brownian motion and utilization distribution models. Dynamic Brownian bridge models combine BBMM with behavioral change point analysis to make Brownian motion parameter more dynamic to provide a more accurate estimate (Kranstauber et al. 2012).

Behavioral change point analysis (BCPA) is used to detect and describe significant shifts in an animal’s behavior without assumptions based on previous knowledge (Gurarie et al. 2009). BCPA can identify discrete and concealed shifts and gradual changes in parameter values, ultimately revealing complex behaviors within and between movement tracts for a single individual. The benefits of using BCPA for studying movement is its ability to provide a robust analysis and reveal structure on typical wildlife tracking data that has heterogeneous behaviors, temporal gaps, and autocorrelation. One limitation of BCPA is that individual movements and behaviors are not classified categorically, and therefore, may limit ecological interpretability and some standard statistical analyses. The BCPA does not provide explanatory models of factors that influence how an animal moves or behaves, rather it simply describes patterns in the movement data.

Time local convex hull (T-LoCoH) can be used to estimate home ranges using local kernels that are created based on the time between relocations (Lyons et al. 2013). This method incorporates velocity to construct a utilization distribution that reflects the animal's movement both spatially and temporally (Lyons et al. 2003). This method is appropriate for questions where the animal's behavior is being analyzed in time and space. Castañeda (unpublished) has used this method for barn owl hunting behavior.

Resource selection functions (RSF) are used to analyze an animal's use of its environment by comparing observed locations to "available" locations where the animal has not been observed (Boyce 2006). RSFs can be estimated using various statistical methods and can be used to determine habitat features that an animal selects for. Important considerations with this method include the scale of the data and the habitat covariates used (Boyce 2006, McLoughlin et al. 2010).



Step selection functions (SSFs) are a type of resource selection function that use the movement ("steps") between consecutive observed locations compared to "available" steps as an animal moves through a landscape (Thurfjell et al. 2014). "Available" steps are randomly sampled based on step length and turn angles to represent movements that the animal could have taken between consecutive locations. SSFs are similar to RSFs in their comparison of observed and available locations, but SSFs differ in their explicit use of movement rather than point location observations as in RSFs.

Whichever method is chosen for a particular study, it is important to publish, share, and communicate data and findings. As field and statistical methods are developed and refined, communication has become a crucial part of the process.

Areas in need of refinement; challenges & considerations

Areas that should receive special attention for refinement include tracking tools, data retrieval methods, data management, statistical power, and workshops to share knowledge.

Challenges and considerations

Challenges to consider when beginning a study include field methods for when, where, and how to trap; the materials, technologies, and impact of tags; and the most appropriate harness. The choice of technology should include the consideration of trade-offs in resolution, lifespan, battery, cost, and expertise needed for each technology. In addition, acquisition of complementary data should be considered and can include, but is not limited to, GIS, video, manipulations, individual traits, diet, and reproductive state. All choices will also include challenges in infrastructure, cost, data storage and processing, and training. Lastly, all decisions should be made while considering the intended audience and methods for publication and outreach.

Recommendations

Our recommendations for telemetry and movement methodologies begin with training in harnessing, statistics, and modeling. Resources for the latter include Max Planck Institute and Smithsonian Institution. Most importantly, the tools chosen must fit questions and logistical constraints. For example, GPS can be used for movement and habitat use at any scale, but it is limited in battery life and cost. Reverse GPS can be cheaper and can be used for foraging success and movement at a regional scale, but it is complicated to implement. Once these trade-offs are considered, it is important to consider methods for complementary data. Dynamic and static GIS data, manipulations, video, and collaboration with farmers and agencies will allow movement data to be put in the context of farm management practices, foraging habitat selection, and IPM systems that include pesticides. The most appropriate choice of methods will include consideration of how to best combine tools, scale of data collection, ecological questions, and complementary data.

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includes all citations from the document, as well as some additional relevant resources

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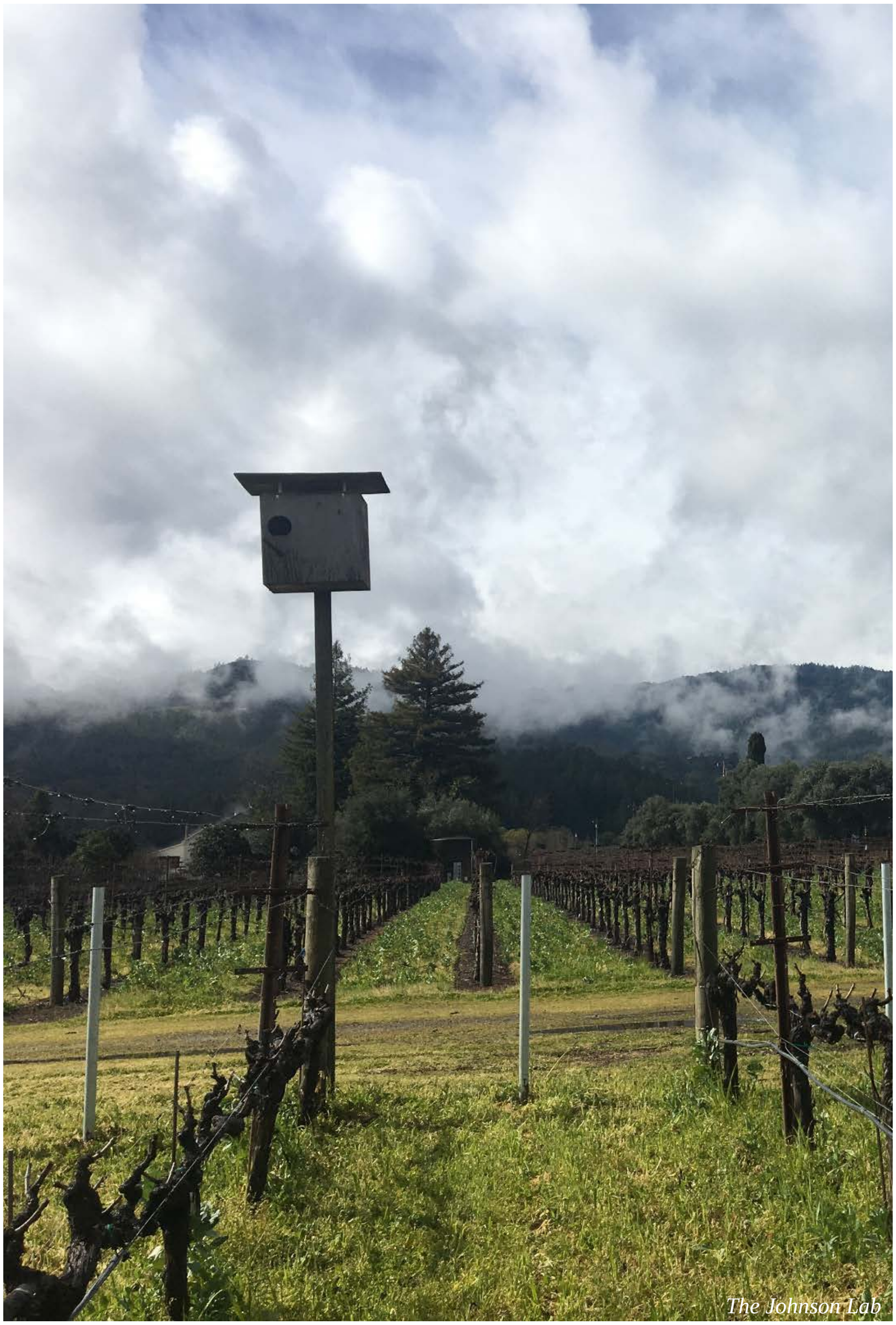
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