

Iowa Multiple Species Inventory & Monitoring Program Technical Manual



Iowa Department of Natural Resources
2025

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Preface

The Iowa Multiple Species Inventory & Monitoring Technical Manual is designed to be both a guide for technicians hired to collect data as part of the multiple species inventory & monitoring (MSIM) program and also a template for other organizations to design their own monitoring programs. The first 7 chapters are the 'office protocols' detailing objectives and tasks to be completed in the office on a computer. The remaining 15 chapters are the 'field protocols', outlining the techniques and methods used to collect the faunal and habitat data.

This technical manual is written in such a manner as to allow interested partners to deploy one, some, or all of the protocols, ensuring that their data are collected in the same manner as that of the Iowa Department of Natural Resources (DNR) for Iowa's species of greatest conservation need (SGCN). Using the same data collection techniques will allow comparisons of data collected at different sites, by different organizations, to the data collected by the DNR. The idea being simply that there is power in numbers.

The manual describes a monitoring program - it is not intended to answer specific research questions. If your organization has a specific question in mind, these techniques may or may not be suitable to collect the data needed to address your question.

This document will be known as version 5. Version 1 was created beginning in 2004 and used through the 2008 field season. Version 2 was updated in 2009 and used through the 2011 field season. Version 3 was updated in 2012 and used through 2015. Version 4 covered years 2016-2024. More changes to the field protocols may occur every 3-5 years or as needed as problems arise. Some minor adjustments (i.e. changing the type of bait used in traps) may not be addressed in these protocols as the adjustments are made.

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Many people contributed to the protocols and ideas described in this book. None of the techniques were invented for this project. Hopefully, all appropriate citations for original work are covered in the Literature Cited section. In addition to the information collected from the literature, many individuals have provided input to these protocols. Stephanie Shepherd wrote the Data Entry handbook, which is a companion to this volume. The Fish Monitoring Protocol - Wadeable Streams was copied almost verbatim from an in-house, Iowa Department of Natural Resources (Iowa DNR) document.

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Chapter One:

Introduction

This manual was written in response to the need for a monitoring program to help fulfill the Iowa Wildlife Action Plan (IWAP) for the state of Iowa. In order for a state to continue to receive funding through the State and Tribal Wildlife Grants Program (STWG), each State was required to submit a WAP which included plans for monitoring species of greatest conservation need (SGCN). In 2006, the state of Iowa designated 296 species as those SGCN in the IWAP, in 2012 this number was revised to 313, and again changed in the 2015 revisions to 405 SGCN. Although there are many parameters by which the IWAP's success will be determined (funding attained, educational programs, recreational opportunities developed, etc.), the ultimate measure of the success of the IWAP will be the impact on the wildlife resources in Iowa. Long term monitoring of all wildlife will be necessary to demonstrate the reversal in declining trends of SGCN and to document that common species are remaining common. Long term monitoring will also be necessary to demonstrate true species declines. This can be accomplished only through the application of a rigorously designed long term monitoring program to track the status of Iowa's wildlife resources.

The current monitoring efforts within Iowa have centered primarily on either game species or been conducted by individuals and groups interested in a specific taxon of wildlife. These surveys are important and will continue but Iowa also needs efforts on other less visible species as several of these surveys are either out of date and/or limited in scope. For clarity, inventory, census, survey, and monitoring are defined as (Thompson et al. 1998):

Census - A complete count of individuals, objects, or items within a specific area and time period.

Survey - An incomplete count of individuals, objects, or items within a specified area and time period.

Inventory - Process of making an itemized list of species occurring within a given area. This may or may not be a complete list of species depending on whether the information was collected through a survey or a census. Alternatively repeated surveys may be used for the inventory of a given area (MacDonald et al. 1991).

Monitoring - A repeated assessment of some quality, attribute, or task for the purpose of detecting a change in average status within a defined area over time.

Long-term monitoring programs give the best picture of the status of wildlife populations over time. Well-designed short-term surveys and inventories can indicate the current status and distribution of wildlife but are often valid only in the area where they are conducted and may quickly become obsolete if habitat or other critical factors change. In Iowa, the rapidly changing habitat availability on agricultural lands as USDA farm programs change is a frequent example.

Purpose of Monitoring Program

The lack of species specific information on the abundance and distribution of SGCN was one of the concerns highlighted in the 2006 IWAP. In some cases, species were added to the list simply because information was outdated or unavailable. The amount and distribution

of potential wildlife habitat is comparatively well known, but in order to relate habitat information directly to wildlife information on a smaller (property) scale, data will also be collected on habitat. The habitat information can then be used as explanatory covariates in species occurrence analyses.

The Multiple Species Inventory and Monitoring Program (MSIM), therefore, is a standardized, statewide survey implemented in order to provide basic inventory information as to the wildlife species in Iowa. The surveys also serve as baseline data for the long-term monitoring program. The program consists of surveys instead of censuses for two reasons: (1). Most likely species will be missed on some properties (i.e. the animals are inconspicuous) and (2). The entire state of Iowa cannot be included in the program (i.e. the area is too large). However, using a randomized sampling design for the property selection, along with the surveys, will allow inferences to be made from the sites examined to the habitats available statewide.

This program can incorporate permanent sampling areas on both public (federal, state, and county owned) as well as

private (CRP, WRP, NGO, etc.) lands. As funding becomes available, the program outlined in Iowa's MSIM Technical Manual can be implemented on additional areas. The SWG-funded program will focus on public lands and private lands and is designed to aid in monitoring private lands enrolled in conservation programs (CRP, WRP, LIP, etc.). The Iowa Department of Natural Resources (DNR) has the primary responsibility for coordinating the program, but the program is designed so that partners (County Conservation Boards, USFWS, NGOs, Universities, etc.) can participate fully in the process.

Background

As developing and maintaining different inventory and monitoring programs for 296 to 405 species is cost prohibitive, the design of the MSIM program is loosely based on the USDA Forest Service's "Multiple Species Inventory and Monitoring Guide" (Manley et al. 2005). The USFS MSIM program shifted from the idea of monitoring indicator species as these programs have been heavily criticized for failing to scientifically show true correlations between indicator or umbrella species and multiple other species of interest (Landres et al. 1988, Niemi et al.

1997, and Lindenmayer et al. 2002). Therefore, the Iowa MSIM program is designed to sample as many species as can be found, including those that are currently considered 'common'. In having unbiased, representative, random samples, the status and trends of all species can be described to the best extent possible. There is no way to predict which common species will be rare in the future, nor which rare species may or may not be common in the future.

The Iowa MSIM program establishes permanent monitoring areas to sample as many species as possible. Each 'core' area encompasses 10.4 ha (25.7 acres), but additional areas will be covered at each location as needed for the species protocols to be implemented. Chapters 8-20 of this manual describe the taxonomic protocols in detail. In addition to the faunal protocols, habitat data collection is described in Chapters 21 and 22. The protocols require various numbers of visits to each property per season.

Objectives

The first stage for implementation of a monitoring program in Iowa was to inventory a random sampling of public and private lands through surveys. The inventory surveys were conducted following the same procedures used in the monitoring program and serve as the first, or baseline, data collection for the long-term monitoring program. More specifically, the primary objectives for the inventory stage of the program include:

1. What proportion of sampled habitat is occupied by a given species?
 - a. What are the detection probabilities for each species? Once the detection probabilities are estimated, it will be possible to estimate habitat occupancy proportions for a variety of scales and specific comparisons of interest, including:
 - i. Iowa as a whole
 - ii. A given region within Iowa
 - iii. A given county
 - iv. A habitat association at the land cover classification level
 - v. Private vs. public ownership
 1. E.g. - Private federal aid program land vs. active agriculture land vs. public land
2. What is the spatial distribution of occupancy based upon these sites?
 - a. Are there any unexpected gaps in species occurrence from a strictly spatial perspective?
3. What are the physical and biological attributes of sampled sites?
4. Are there changes that need to be made to the individual sampling protocols?
5. Do the results illuminate the need for future or immediate research on specific species, communities, or habitats?

Other benefits gained during the inventory stage of the monitoring program include:

1. What are the relationships between spatial distribution of a species and associated habitat conditions?
 - a. Predictive models of species occurrence based upon habitat variables (logs, snags, vegetation composition, etc.)
 - b. This information should be useful for management decisions such as:
 - i. For a given species, is more habitat needed or is it adequate?

- ii. For a given species, is the habitat high-quality or marginal?
 - iii. Are there restoration opportunities or other management options for species of interest?
- 2. The identification of public areas susceptible to the stresses summarized in the IWAP.
 - a. Assess the impact of the perceived stress.
 - b. Continue to determine if there are additional stresses not specified by the IWAP that should be addressed in future IWAP revisions.

These additional benefits may depend upon the availability of either additional resources or interested scientists willing to assist with the analyses.

The initial inventory phase has been completed and a subset of properties are visited annually with that more than 15 years of data collection, the objectives move from those related to inventory into objectives more specifically related to monitoring. Currently, the first priority becomes measuring the trends in each species. Specifically, the primary objectives for the monitoring include:

- 1. Is there a change in species occupancy of sampled properties over time?
 - a. If so, what is the change in occupancy rates and patterns (colonizations and extinctions)?
 - i. These changes may be able to be linked to invasive species or climate change if a long time series data set is collected, Jonzen et al. (2005) suggests at least 15 years of data is needed in this situation.
 - b. Is the change linked to a certain scale or spatial distribution pattern (i.e. is it localized to one region of the state?)
- 2. Is there a change in community composition?
- 3. Is there a change in habitat?
 - a. If so, did the habitat type increase or decrease?
 - b. Did the habitat quality improve or degrade?
- 4. Is there a relationship between changes in species and the habitat conditions?

In addition to the primary objectives for the monitoring phase of the program, we expect to have additional benefits, including information towards the following:

- 1. What are the effects of management actions or natural disturbance on wildlife populations and habitat conditions?
 - a. This information is expected to serve as a starting point for additional research into a given topic.
 - b. In order to examine management actions, we would need a database to collect management actions from the property managers. We do not have such a database in place at this time.
- 2. Provide data complimentary to existing large scale monitoring programs (such as the National Breeding Bird Survey) and continue to strengthen species occurrence patterns predicted by other programs.

Intended Relationship to Other Monitoring Programs

In following the basic outline of the USFS MSIM Program, Iowa will be collecting data that can be compared at a larger-nationwide scale, should the USFS program become nationwide. Currently, the USFS program is limited in scope and not being used in national forests near Iowa. Iowa has no national forest land.

The design of Iowa's MSIM program has been created in a manner to allow other interested partners to utilize all or part of the taxon protocols depending on their interests and available resources. Once the plots are delineated, some of the protocols could conceivably be carried out by dedicated volunteers, others will need to be performed by employed technicians. In any case, this will allow various partner organizations the ability to collect data on species of particular interest to them in a manner which will allow their data to be comparable to a larger dataset for Iowa. This should aid in illuminating meaningful changes or other information in a species of interest. Partners working in Iowa are encouraged to use our on-line database to store their data as long as the data are collected following our standardized protocols.

Chapter Two: Sampling Design & Plot Establishment

The strength of the monitoring program design is based upon the randomized property selection. By using a random selection of areas to include and by not choosing areas specifically because species of interest were known to occur there historically, inferences as can be made for more areas than are surveyed. If only areas known to contain the species of interest were included, then any conclusions or correlations inferred from the monitoring program could only be linked to the areas examined. The expectation is that several of the areas that are known to have species of interest will be included in the study even though they were selected at random as opposed to being selected as a target. The power of the program rests on the idea that any property has an equal chance of being chosen within its given habitat stratification.

However, given that land owned, managed, or affiliated with the Iowa Department of Natural Resources or other non-farming, non-urban entity has a much greater chance of being included in this study than active farm or urban areas, this program may more aptly monitor wildlife associated with these conservation oriented areas as opposed to a true state-wide program. Similarly, the majority of land in Iowa (>80%) is classified as agricultural (including row crop and pasture lands), yet, again due to the majority of land ownership being private (98%) and the focal areas for the monitoring program being primarily publically owned (although both row crop and cool season grasslands are habitat classifications which are included), the monitoring program will not be comprised of >80% agricultural lands. Therefore, the results obtained with the monitoring program will apply primarily to non-agricultural, non-urban areas.

Plot Locations

Public Lands

Due to funding and personnel constraints, the majority of the effort expended by the Iowa DNR will be focused on public lands. Iowa has approximately 2.48% of the total land area in public ownership, with approximately 1.03% being owned by the Iowa DNR. The 2.48% public ownership includes DNR lands, federal lands, county conservation board lands, city parks, and trails. Federal entities and county conservation boards are willing to partner with the Iowa DNR to monitor lands in their ownership.

Of the 2.48% of Iowa land in public ownership, all areas 247 acres or larger (and some smaller areas within target wetland classes or counties) were classified according to the 19 habitats outlined in the IWAP (Zohrer 2005).

Classifications were made using both the knowledge of the land manager and, in some cases, using aerial photos and a 2002 GIS landcover classification layer. These habitats with their definitions, as listed in the 2006 and 2012 IWAPs, include:

Forest - More than 60% canopy of tree species with crowns interlocking.

Wet forest/woodland - Temporarily or seasonally flooded forest or woodland.

Woodland - Open stands of tree species with 25-60% of canopy cover.

Shrubland - Shrubs >0.5 m tall forming >25% cover with <25% tree cover.

Wet shrubland - Temporarily, seasonally, and semi-permanently flooded wetlands or saturated deciduous shrubland.

Herbaceous wetlands - Temporarily, seasonally, or semi-permanently flooded or saturated herbaceous wetlands.

Warm season herbaceous vegetation - Less than 25% canopy cover made up of trees or shrub species. Herbs form at least 25% canopy cover.

Savanna - Temperate grassland with sparse coniferous or cold-deciduous tree layer.

Cool season grassland - Smooth brome, forage crops, and pasture.

Cropland - Worked land normally on an annual basis in corn, soybeans, sorghum, fallow fields or other crops.

River - Large flowing bodies of water, normally with permanent flow and draining over 100 mi².

Stream - Smaller flowing bodies of water, normally permanent, that serve as tributaries to rivers and drain less than 100 mi².

Creek - Even smaller flowing stretches, often intermittent and ephemeral, that flow into streams.

On-stream impoundment - Slowly flowing bodies of water formed from artificial damming of a river, stream, or creek, generally <500 acres in size and having a watershed to lake ratio >200:1.

Backwater - Slow flowing bodies of water associated with large river systems. Back-channel, low-lying areas filled

with water during high flow events but may be completely isolated from the river during low flow and may exhibit no flow during these periods. They are especially prevalent on the Mississippi River.

Oxbow - A sub-class of backwater, water bodies formed in old river channels that are currently cut off from the main channel and flow of a river.

Lake - Large bodies of water exhibiting little or no flow with emergent vegetation over less than 25% surface area. They may be either natural or constructed.

Shallow lake - Open, freshwater systems where maximum depth is less than 10 feet. Normally in a permanent open water state due to the altered hydrology of watersheds and unmanaged outlet structures that maintain artificially high-water levels. May be fringed by a border of emergent vegetation in water depths <6 feet. When clear, they are dominated by emergent and submergent vegetation.

Pond - Smaller standing bodies of water, often exhibiting large swings in dissolved oxygen and water temperature and generally <10 acres in size.

For Iowa, in 2005, habitats within each management district were classified by the managers and areas were randomly selected within each habitat class. This list of areas to be included in the monitoring program is listed in other documentation. The stratified random sample selection of sites followed the ensuing procedure:

1. Areas were listed in Excel and assigned numbers using the random number generator function within each habitat classification (primary stratification) and also for the district (secondary stratification: northeast quarter, southeast quarter, northwest quarter, southwest quarter). The secondary stratification allowed for the selected habitats to be more equally split across the state as opposed to being clustered together within one corner of the state.
2. Sites were then sorted by number and those chosen were rotated such that one selection per habitat was made during each round.
3. Once an area was selected for a particular habitat classification, that area was excluded from future selection in other habitat classifications.
4. In some regions, the number of sites with a particular habitat was limited, e.g. only 8 areas had savanna in the northwest. In this case, those 8 areas were given a higher priority in the limiting class when compared to classes with more possibilities for selection. This will still be considered to be a random site selection as 5 of the 8 possible sites were chosen using the random number generator, although these sites may have been excluded from consideration in the other categories.

We devoted a substantial amount of time for this selection process and if our areas were 100% of the target habitat within a 26-acre core area, this time would have been well spent.

However, due to the fact that most of the publically owned properties in Iowa have a mix of several habitats, we most likely would have had a property list with similar habitat composition had we excluded the extra step with the habitat strata and instead done a random selection within each of the four districts of the state. Now, properties are selected randomly and then assigned to a habitat classification as delineated above. We rely on landscape habitat metrics from GIS layers (e.g. Iowa's 2009 High-resolution Land Cover) for those classes as opposed to using the methodology outlined above now when conducting analyses.

Private Lands

As 97.52% of Iowa is in private ownership, it is imperative that the monitoring program have access to a portion of private lands. Lands owned by Iowa Natural Heritage Foundation, The Nature Conservancy, the Meskwaki Tribal lands, and Whiterock Conservancy are a few examples of private lands owned and protected by non-government organizations. These NGO's are regarded the same as the other public land owner organizations - the DNR hopes to include these areas in the monitoring, but it may be necessary that the organizations hire the temporary staff needed to conduct the protocols.

Again, it is anticipated that funding will be available from the State Wildlife Grant program to fulfill salary and equipment expenses associated with the MSIM program.

Beginning in 2016, we began including private lands where the landowner had worked with DNR staff on one of four

conservation practices: CRP, WRP, Periodic Disturbance, or Invasive Species Removal.

These protocols have been developed in such a manner as to allow for some basic questions about land management practices to be examined. However, these should not replace a rigorous study design around a specific action. The protocols can be used to look at species occurrence on a large scale and using the habitat and GIS protocols will allow correlations to be made between land attributes and species occurrence. Therefore, these protocols should be adequate to monitor wildlife and wildlife responses to management and conservation actions, at least at the occurrence level for a wide variety of species. This will allow Landowner Incentive Program lands, Wetland Reserve Program lands, and other private landowner aid programs to find the information collected under these protocols useful.

However, it should be noted that if a particular species or management action is in question, a scientific study should be designed to focus on that species or action. It is also expected that the monitoring protocols will elucidate specific species or management questions that will need to be examined through research studies.

Many of the private lands may be smaller than the 1 km² (247 acre) utilized in the selection of the state-owned lands for this program. The protocols can be adjusted to fit a smaller land area by either searching a smaller amount of habitat or by dropping inappropriate protocols, e.g. searching for fish can be omitted in areas without adequate habitat. It will be left to the discretion of the MSIM Leadership Team or partner collecting data as to which protocols are or are not of interest or practicality in implementation.

Likewise, it is expected that the partner organizations will not be able to randomly choose the areas to be monitored as the agency can. However, as long as the majority of areas utilized in this program are chosen at random, it is expected that the non-random site selection of partner organizations, coupled with the somewhat random ownership of land across Iowa, will not impact the statistical strength of the monitoring program when compiling statewide statistics. If a property is not randomly selected, a comment should be included in the Property Comment box of the MSIM database indicating it was not randomly selected. Alternatively, different statistical methodologies can be used to compare parameters collected from random and non-randomly chosen sites.

Monitoring Plot Design

The core area of each plot is contained within the shape of a hexagon. Six poles delineate the hexagon and serve as the bird point count locations (with an additional point located in the center of the hexagon). The hexagon is roughly 10.4 ha (25.7 acres) in size. **However, the protocols are not limited to the area inside the hexagon.** Aquatic species, especially, may need to be searched for within a larger area. Certain sites may require extra effort, but as a general rule, up to 10 wetlands within a reasonable distance (500 m in any direction) to the center of the hexagon could be searched for aquatic species as allowed by the landowner(s). This distance should be sufficient to allow for adequate sampling for fish in lotic systems as well as it would equal roughly a 1 km² (247 acre) area spanning a distance of 1 km (0.62 mi) centered on the middle point.

The protocols have been designed to be implemented in a variety of habitats. On one extreme is the bird protocol, where birds are expected to be found in all 19 habitats and therefore, the bird point counts can be conducted in all habitats. At the other extreme would be the aquatic protocols, the fish and mussel protocols in particular. Should the area being examined have no adequate wetlands to support fish or mussel populations, these protocols would simply be omitted at that property.

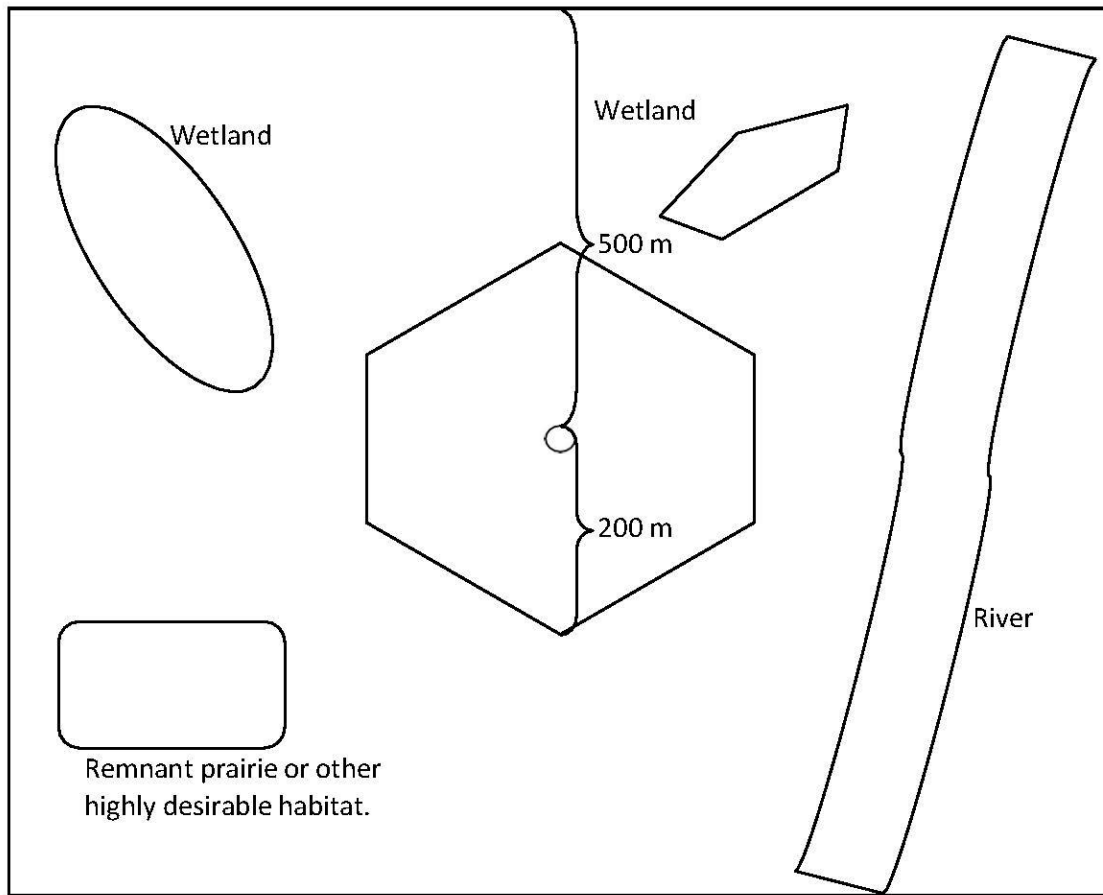


Illustration of a hexagon plot and associated sampling area. The distance from the center point to each outer point of the hexagon is 200 m, the distance from the center point to the outer edge of the additional sampling area is 500 m, or 1000 m centered on the middle point. Additional areas to be searched within this 1-km² area are illustrated and labeled as wetlands, remnant prairie, or other desirable habitat.

Data Collection Methods

By utilizing the larger, 1 km² (247 acre) area, additional potential habitats should be available which should increase the number of species found per site. For example, while the small mammal traps will be placed along the lines of the hexagon, the mammal track searches can encompass any muddy area, and cameras can be set anywhere within the 1 km² area. Bird point count locations are also tied to the hexagon, yet the Bird Visual Encounter Surveys could be done in the larger area. The search for herpetofauna will encompass the larger area as well. Looking for the aquatic species, primarily fish, but also the amphibians, butterflies and dragonflies to a lesser extent, will require searching the larger area for most plots. Individual protocols for each taxonomic group follow in the subsequent chapters.

Remember that these protocols were designed so that organizations with limited resources and specific taxonomic interests can choose which protocols to implement should it be infeasible to employ all protocols.

Chapter Three: Landscape Characteristics Protocol GPS/GIS

Monitoring

Once the randomly chosen properties have been identified, a GIS system should be used to gather information and choose the hexagon point locations within each site. The center of the hexagon should include the habitat classification for the area. If, due to property ownership, it is not possible to center the hexagon over the habitat classification, then the hexagon should be placed such that as much as possible of the area inside is comprised of the habitat by which the site is classified. Placing the hexagon in GIS is the responsibility of the project coordinator. Realize, however, that subsequent ground-truthing of the property may result in needing to adjust the location of the hexagon. Any changes in location of the hexagonal points must first be approved and recorded by the Program Coordinator.

The purpose of the hexagon is simply to place the bird point count locations and Sherman traps in the same orientation at as many properties as possible. The hexagon shape is the most efficient for spacing locations although other shapes can be used. This shape requires the least amount of walking effort while maintaining a 200-m distance between point locations.

The GIS is also used to collect information on landscape characteristics. The data should be ground-truthed by the technicians during the field season. Although the information collected under this protocol will probably not change over several years for the majority of properties, the potential for change is still present. Therefore, this information should be re-collected or re-ground-truthed each additional year of the monitoring.

Survey Methods

The seven points that comprise the hexagon sampling plot will be pre-determined prior to field work through information collected under this protocol. The 7 points include the center point and 6 edge-angle points to form the hexagon shape seen below. Each point is spaced 200 m from the adjacent points.

Choose the center point for the hexagon such that it is either centered over the primary habitat classification for the site or such that the majority of the habitat within the hexagon is comprised of the habitat for which the site is classified. Record the UTM coordinates for the center point location. Use the formulas in the table below to determine the locations of the outer 6 points that form the shape of the hexagon.

Note the location of wetlands in and around the hexagon. The area around the hexagon should be included up to a 101-ha (250 acres) block. Should the property be smaller than 101 ha, then include all information, but unless we have obtained permission from the landowner to be on the adjacent property, this information will not be able to be groundtruthed. Roughly calculate the amount of each habitat type that occurs within the hexagon. Also calculate the amount of each habitat type that occurs within each 101 ha block. Include information on the number and type of roads within and around each site.

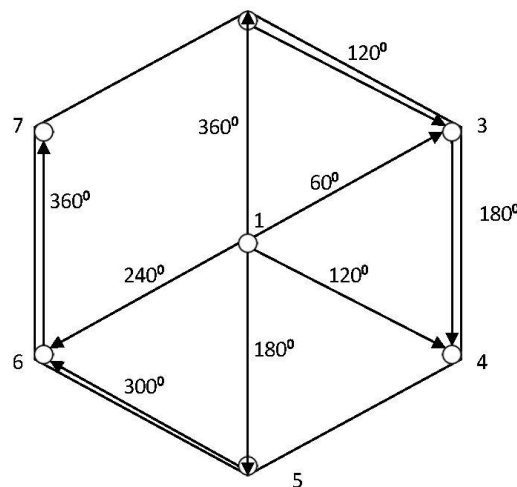


Table 1. Formulas used to choose point locations around the point chosen to center over the representative habitat for the study area.

	UTM E	UTM N
Center point (1)	Choose from GIS coverage (X)	Choose from GIS coverage (Y)
Point 2	X	Y + 200
Point 3	X + 173	Y + 100
Point 4	X + 173	Y - 100
Point 5	X	Y - 200
Point 6	X - 173	Y - 100
Point 7	X - 173	Y + 100

Roads and trails located within 30 m of wetlands should be listed by category and distance. There are different data sheets for lentic and lotic sites. Categories include: 4 lane highway, 2 lane highway, paved road, unpaved road, OHV (off-highway vehicle, i.e. all terrain vehicles) trail, and hiking trail. In addition, the area of compacted soil or impermeable surfaces within 10 m of the wetland edge should be estimated. For lentic sites, also record the number of road crossings along with an estimate of the total length of the stream they impact (so, add up the widths of the roads where they cross the channel). All information should be ground-truthed.

Groundtruthing

Technicians will be assigned specific tasks for ground-truthing. Logically, the fisheries and amphibian technicians can be charged with ground-truthing the wetland areas. The small mammal, bird, and other terrestrial habitat technicians will most likely have the responsibility of ground-truthing the habitat classifications and the rough boundaries for each habitat. Road type and area can be checked by anyone. Once the information has been ground-truthed, it can be entered into the database.

Most wetlands will be identified only in the field as they are often too small to be visible via GIS or aerial photos. Field technicians should clearly label these wetlands and record GPS locations (walking the perimeter if possible) to ensure that all technicians within the field crew used the same wetland/area name for each one during faunal and habitat data collection.

Photostations

Each of the 6 points of the hexagon and the center point of the hexagon should be considered a photostation. Fourteen digital photos should be taken from these points at least once per year, preferably once each season. Leave at least 2 months between each photo session. The following table lists the angles at which the photos should be taken.

Table 2 Photostation locations and angles.

Number	Station	Angle of photo (in degrees)	Comments
1	1 (center point)	90	Due east
2	1 (center point)	270	Due west
3	2 (top of hexagon)	0	Due north
4	2 (top of hexagon)	180	Due south, toward center pt
5	3 (clockwise from top)	60	Away from center
6	3 (clockwise from top)	240	Toward center point
7	4	120	Away from center
8	4	300	Toward center point
9	5 (bottom of hexagon)	0	North, toward center pt
10	5	180	South, away from center

Number	Station	Angle of photo (in degrees)	Comments
11	6	240	Away from center
12	6	60	Toward center
13	7	300	Away from center
14	7	120	Toward center

Be sure to set the digital camera to the automatic mode and zoom out until the photo will cover the largest area. Record the number of the photo with the date, site, station number, and angle at which the photo was taken. When the photos are uploaded to the photo-database, be sure to label all photos with the pertinent information.

Photo/Map Book

A 3-ring binder of aerial photos and maps should be made for each crew. Included in the binder should be maps of each of the properties for which they are responsible. If known, on the aerial photos, the wetlands should each be labeled with a number or name to facilitate in assigning the correct wetland to the data collected under the faunal and habitat protocols. Other areas of interest should be highlighted and labeled on the photos as well. This will prevent technicians assigning different names to the areas which would be confusing when the data is being entered into the database. This book should be left in the field vehicle and should include contact information for each property as well. It is also advisable to create a '911 sheet' with driving directions that could be read to a 911 operator in an emergency.

Digital Maps

Digital maps, such as google maps, should be made available to the crews. These digital maps can provide the property boundaries and site locations within a property shown in the map book. These maps can also be updated through out the year with new site locations or other noteworthy locations within a given property. Lastly, these digital maps can be used to provide specific transects or other designations that would be help for the crews to survey in the correct spot year after year.

Equipment List

- Computer with appropriate ARC GIS software and GIS database/aerial photos
- GPS unit
- Compass
- Surveyors tape
- Data sheets
- Pencils
- Digital camera

Staff & Training

Staff will be trained in the basic use of the GPS unit during the workshop at the beginning of the field season. This training should include practice surveys to ensure that proper procedures are followed.

Data Quality & Management

Ground-truthing of the data collected through the GIS system will serve as the primary quality control for this protocol. Once information has been checked in the field, it can be entered into the database.

Data Analysis

The data will serve to both aid in the selection of specific areas for targeted visual encounter surveys and aquatic trap placement as well as being used to correlate wildlife species presence and absence.

Safety Considerations

Typical field considerations should be followed. Proper hygiene (i.e. hand washing, checking for ticks and other potential parasites) should be maintained. Technicians should take proper precautions around water (i.e. avoiding fast, deep flowing water).

Additional Methods for Special Locations

None

Chapter Four: Data Entry & Database Maintenance

General Database Information

All information collected during or pertaining to the field season should be entered into a computerized database. Information left on scraps of paper and hidden in a desk drawer is not useful to anyone other than the person that already knows it exists. At a minimum, the database should contain information relating to the area(s) surveyed (GIS and habitat data), the times surveyed (time of day and weather conditions) as well as the species encountered and all data collected on each individual.

The data will need to be extracted for use in various analysis programs using different formats. Therefore, it is probably best to use a database that is capable of querying the data by several different manners.

Iowa DNR MSIM Database

In 2008, the Iowa MSIM database was converted to a web-based system. It is maintained by DNR IT staff. Technicians (or others) charged with data entry must first apply to get a password from IT.

<https://programs.iowadnr.gov/msim/pages/login.aspx>

There is a companion manual on the Iowa MSIM database to assist with data entry.

The following information is described the DNR MSIM database:

Vocabulary

- Property - The largest entity being surveyed.
- Site - The survey locations within each property (e.g. BPC1, Pond 1, Creek, etc)
- Plot - The survey locations within a particular site. Mostly used for habitat data.

Database Structure & Fields

The main information should already be entered into the database before the beginning of the field season. These data include information such as owner name and contact, UTM coordinates for bird point count locations and wetlands or other areas of interest. Only the project managers can add properties to the database.

For survey sites on a property but NOT on a pre-programmed site, technicians may suggest names when they send UTMs to the MSIM Coordinator to be added to the database.

Helpful Hints

1. Be sure to update each screen before you leave it to ensure that the data you have entered has been saved.
2. If you are entering information from a previous year, you must first choose the correct year before the other options appear on the screen. For example, if you were attempting to enter data collected in 2018 during 2025, you would not have any choices for property or observer until 2018 had been selected.
3. You are required to validate each survey before you log off the system. This entails looking through the data entered and checking for errors. If you find errors, please correct them before logging off.
4. Ideally, once a week, you should receive an email with one of the surveys you have entered attached. This is for an in-depth data check. Compare each line of data entered against the original data sheet. Correct any errors. If there are errors, continue to check other surveys to ensure all information was entered correctly.

Chapter Five: Data Analysis

Analyses described below represent only a handful of possibilities for evaluating the data. This chapter is not meant to serve as a primer for data analysis, but should provide a starting point for further understanding. For consistency between techniques, information provided under each heading includes the parameters to be estimated, procedures used to collect the data, examples, requirements and assumptions of the analysis, advantages, disadvantages, and additional literature.

The data collected under these protocols can be analyzed with many different methods.

The primary objective, for the first inventory component of the surveys, is to determine the locations of wildlife populations, the characteristics of the habitats they are found in, and the status of those habitats. The primary parameter of interest, then, is the proportion of area occupied by a given species. We know have over 15 years of data and the MSIM Program has moved into the monitoring phase (although we do still inventory properties for the first time). As such, we can compute trends (e.g. - how occupancy changes over time) and correlate those to habitat variables.

Proportion of Area Occupied

Single Season Surveys

Since the permanent sampling plots were visited >1 during the season the target species were expected to be present, Proportion of Area Occupied (PAO) will be used to determine both the probability of occurrence and the detection probability of a given species in a given area following MacKenzie et al. (2002 and 2005). The permanent sampling plots can be divided into habitat classes, regional areas, or lumped together to be analyzed state-wide. Previously, Program PRESENCE was created by Darryl MacKenzie and Jim Hines (available for free download at (<http://www.mbr-pwrc.usgs.gov/software.html>) and was adapted and added into Program MARK (also freeware available at same web address) by Gary White. If using Program MARK to compute the calculations, be sure to choose “Occupancy Estimation” for the “data type”, unless the data set contains multiple years of data. All data analyses can be computed in Program R using packages like ‘unmarked’ (Kellner et. Al 2023) or ‘spOccupancy’ (Doser et.al 2022) today.

Parameters Estimated

Detection probability (p) - the probability of finding an individual of a given species at a given site during a given time.

Occupancy (Ψ) - the probability that a randomly selected site or sampling unit in an area of interest is occupied by at least one individual of a given species.

If one ignores the probability of detecting a species (and assumes that this probability is equal to 1 - meaning it is always found if it is present), it is easy to calculate the occupancy probability, simply divide the number of properties with the species by the total number of properties surveyed. This value is commonly called the ‘naïve estimate’. Most species do not have a 100% detection probability, they commonly ‘hide’ from the observer or avoid the trap set to capture them. The occupancy models take this into consideration and incorporate detection probability (p) into the estimate of occupancy (Ψ). For example, the likelihood function of a survey record 01010 for a given property is:

$$\Psi(1 - p_{i1})(p_{i2})(1 - p_{i3})(p_{i4})(1 - p_{i5})$$

The animal was seen during the survey, so a Ψ is included and (1- Ψ) is not included. The p_i denotes that the animal was seen during that survey, while the (1-p_i) indicates it was not detected during that survey. But the survey history of 00000 does not necessarily mean the species is absent and therefore must include the possibility that the species was present but not detected in addition to the possibility that the species was absent. This likelihood function is, therefore, written as:

$$\Psi(1 - p_{i1})(1 - p_{i2})(1 - p_{i3})(1 - p_{i4})(1 - p_{i5}) + (1 - \Psi)$$

Data Collection Procedures

Multiple visits are made to a given property over a single 'season'. The site is searched or trapped during this time frame for the species of interest. It is not necessary to mark the animals captured during a given visit for this analysis.

Example:

Almost any of the faunal protocols in this manual could serve as examples. Specific examples would include visual encounter surveys, and bird point count surveys.

Requirements & Assumptions

Requirements include multiple visits to each site during the 'season' of interest. These models assume that sites are 'closed' during the period of the survey season, meaning that a species is either present or absent on the first day of the survey and the status of the species does not change throughout the duration of the survey. Therefore, it is critical that the appropriate beginning and ending dates for each survey are chosen and followed. It may be necessary to truncate the data within a season to ensure this assumption is met. The other 2 assumptions of the models are that species are identified correctly (& therefore, never recorded as present when in fact it is absent) and that detection between properties is independent. There may be a problem with the independence assumption if plots are located too close together and the same animal is using both areas. The permanent sampling plots should be located to avoid this situation.

Advantages

Advantages of this technique include that it does not require that individuals be marked.

Additionally, the analysis will allow the inclusion of missing observations. If a species was detected during a site-visit, then that presence is recorded as a "1" in the data set. If the species was not detected during a site-visit, then it is recorded as a "0" for that visit. Dates without survey data for a given site are denoted by a "." for missing data in program MARK, a "-" in program PRESENCE, and left blank in program R. Just because a species was not detected does not mean that it was necessarily absent, it may have been absent or it may have gone undetected for a number of reasons, including that it was hidden out of site or that it was not hidden but still missed by the observer (MacKenzie et al. 2002).

Disadvantages

The occupancy parameter is used as a surrogate for population size or species abundance.

Population size and abundance both require additional information that may or may not have been collected with a given protocol. Occupancy analyses rely on species presence and absence data only, not the number seen or captured.

Additional Literature

Burnham, KP, and D Anderson. 2003. *Model Selection and Multi-Model Inference*. Springer-Verlag, Inc. New York, New York.

Cooch, E, and G White. (<http://www.phidot.org/software/mark/docs/book/>). *Introductory User's Guide to MARK*. Last accessed: 9/14/05. This is a user friendly manual For Program MARK.

MacKenzie, DI, JD Nichols, GB Lachman, S Droege, JA Royle, and CA Langtimm. 2002. *Estimating Site Occupancy Rates when Detection Probabilities are Less than One*. *Ecology*. 83: 2248-2255.

MacKenzie, DI, JD Nichols, JA Royle, KH Pollock, LL Bailey, and JE Hines. 2006. *Occupancy Estimation and Modeling*. Academic Press. Burlington, MA.

Single Season Surveys with Covariates

There are 2 types of covariates which can be incorporated into the models. Site-specific covariates are those that do not change between sampling occasions. These variables would be things which would typically be measured only once during a survey season. Examples may include the number of trees in a given area, the amount of woody debris, litter depth, the amount of area of a certain habitat cover, etc. Now, realize that any of the above examples may, in fact, change (e.g. maybe a site is logged during the field season and the number of trees decreases, or a site catches fire and both amount of woody debris and litter depth changes). Should this occur, the covariates may need to be re-measured

and considered as sampling covariates instead of site covariates. However, it is at the discretion of the data analyst to make this decision.

A sampling covariate is a variable which changes between site-visits. Examples include amount of rainfall, temperature, amount of search effort, etc. These variables need to be measured every time the technician is in the field recording data. If someone forgets to take a measurement, the other measurements for that same day could be averaged to use for the missing values, depending on the information in question.

Multiple Season Surveys

If the same sites are visited multiple times over several years, we can compute estimates of colonization (γ) and extinction (ϵ) probabilities in addition to the proportion of area occupied (Ψ) (MacKenzie et al. 2003 and 2005). This is especially useful for tracking species range expansions or contractions and can be considered as a measure of the status (or trend) of populations of a species.

The design for these models is basically the robust design commonly used in mark-recapture studies. The robust design includes several primary periods (usually years) during which the surveys are conducted. Within each primary period the properties are considered 'closed' as they are in the single season surveys. A property is either occupied or unoccupied during the season, it cannot be occupied and then become unoccupied (or vice-versa) during the survey season. Within each primary period there should be 2 or more secondary periods, for our purposes these are the actual dates of the surveys. Since there are several surveys within each primary period and there are also several primary periods (i.e. years of data), we can compute the extinction and colonization probabilities. This is possible because the status (occupied or unoccupied) of any property is allowed to change between primary sampling periods.

Parameters Estimated

Detection probability (p) - the probability of finding an individual of a given species at a given site during a given time.

Occupancy (Ψ) - the probability that a randomly selected site or sampling unit in an area of interest is occupied by at least one individual of a given species.

Colonization (γ) - the probability of a site being unoccupied at time t and occupied at time $t+1$.

Extinction (ϵ) - the probability of a site being occupied at time t and un-occupied at time $t+1$.

Change (λ) - the rate of change in occupancy (not estimated from the software program):

$$\lambda_t = \frac{\Psi_{t+1}}{\Psi_t}$$

As with the single-season surveys, presence is denoted by a '1', absence by a '0', and missing data by a '.'. Again, one could use either Program MARK or PRESENCE to compute the parameter estimates. Should a model that allows year to vary for ϵ and γ be the best fit, it may be necessary to calculate a Ψ for each year by hand using the following equation:

$$\Psi_{(t+1)} = \Psi_t(1 - \epsilon) + (1 - \Psi_t)\gamma_t$$

Data collection procedures

Multiple visits are made to a given property over a single 'season' and multiple 'seasons' (or years) are covered before this can be utilized. The site is searched or trapped during the time frame for the species of interest. It is not necessary to mark the animals captured during a given visit for this analysis.

Example

Almost any of the faunal protocols in this manual could serve as examples. Specific examples would include visual encounter surveys, and bird point count surveys.

Requirements & Assumptions

More than one year of data collection with several visits to a given property within a given year (or season) are required. The assumptions are the same 3 as for the single season surveys (closure within season, correct identification, and

independence between properties) but the closure assumption is relaxed between years. This means, that although the site must be either occupied or unoccupied within a season (or year), the occupancy status is allowed to change between seasons (or years).

Advantages

The advantages are the same as for single season surveys.

Disadvantages

The disadvantages are the same as for single season surveys.

Additional literature

The suggested literature is the same as that for the single season surveys.

Multiple season surveys with covariates

The same covariates as collected for single season surveys can be used in multiple season surveys. Remember that the larger the data file is, the longer the computer program will take to run. There are several ways to incorporate covariates into the models.

Site specific covariates are allowed to change between years or seasons and can be applied to occupancy (ψ), colonization (γ), extinction (ϵ), and detection probability (p). Sampling occasion covariates are allowed to change with every visit and are applicable to detection probability (p) only.

Distance Sampling with Variable Circular Plots (Bird Point Counts)

Using circular plots (e.g. point counts) is a method primarily used with birds and was developed as an alternate to line transects. The point count method is especially useful in rough terrain and in areas of complex vegetation. It is often preferred to line transects as point counts result in less disturbance due to the observer being stationary as opposed to moving through the habitat.

Parameters

Density ($\hat{D}$) - number of individuals per given area.

Data collection procedures

Determine the distance between the observer at the center of the point and the animal detected. The points where the observer stands should be either randomly or systematically placed. Typically, an observer stays in the center of the point for a specified amount of time (e.g. 2 minutes) before beginning the data collection and remains standing at that location throughout the timed count (e.g. 10-12 minutes).

Example

An example would be the data collected following the bird point count protocol.

Requirements & Assumptions

The observer must have the ability to pinpoint the location of the animal and judge the distance to that animal. The specific assumptions for this method include that the observer always detect an animal at the point (i.e. if an animal occurs where the observer is standing it is always seen). Animals are detected at their initial location before they move in response to the observer. A third assumption is that distances are measured accurately or accurately within the distance-group interval. Other assumptions are that the animals are not counted more than once (i.e. the same individual is counted only once), that animals are correctly identified to species, and that point locations are randomly placed in the area of interest. Locations of the animals do not have to be randomly distributed through the area (i.e. can be clumped or flocked together).

Advantages

Since the locations are known prior to the start of data collection, distances at each location can be flagged, if necessary to aid in correct distance measuring. Other advantages (compared to line transect sampling) include that radial distances

are easier to measure and that point counts are easier to employ in patchy, complex habitats.

Disadvantages:

Disadvantages include the initial disturbance caused by the approach of the observer. This can often be eased by having the observer stand still for a specified amount of time prior to beginning the timed data collection. Individuals collected between timed data collections are not usable in this analysis. This analysis may not be efficient for species with low densities.

Additional literature:

Buckland, ST, DR Anderson, KP Burnham, and JL Laake. 1993. Distance Sampling: Estimation of Biological Populations. Chapman and Hall, New York.

Williams, BK, JD Nichols, and MJ Conroy. 2001. 13.3 *Point Sampling*. In: Analysis and Management of Animal Populations: Modeling, Estimation, and Decision Making. Academic Press. San Diego, California.

Mark Recapture or Mark Resight

While these data would be collected under potentially more time consuming and costly protocols, the information gained is probably the most informative. The parameters that can be estimated will depend on the amount of effort expended. For example, survival rates for small mammals could only be computed if additional effort (compared to that required under the small mammal protocol in this manual) were employed such that properties were trapped for multiple nights on more than 1 occasion per year. Survival estimates for anurans could be calculated on the number of visits needed per site, per year, IF the animals were marked, which is not usually done (but could be) under the amphibian protocol.

Parameters

These depend on the design of the field study but may include:

Density ($\hat{D}$) - number of individuals per given area.

Population size ($\hat{N}$) - estimate of the number of animals in the population.

Survival (ϕ or S) - typically the proportion of the population that survives from time t to time $t+1$.

Detection probability (p) - the probability of finding an individual of a given species at a given site during a given time.

Data collection procedures

This involves the capture, marking, and release of animals on multiple occasions within and/or between years. The occasions should be separated by a period of time where the area is not trapped or searched for that taxonomic group. Marked and unmarked individuals should be able to be captured for multiple time intervals.

Example

Any protocol where animals were marked and recaptured with sufficient numbers should be able to be analyzed using this procedure.

Requirements & Assumptions

For these analyses to work, large numbers of animals must be able to be captured and marked on multiple occasions. There must also be the opportunity for re-finding significant portions of the animals. General assumptions include that the captured sample is representative of the larger population. Age and sex are correctly determined. There is no loss of marks. Survival and recapture are not affected by marks. Time of resight, recapture, or recovery is recorded correctly. Additional assumptions may apply depending on analysis (i.e. assumptions may vary if emigration is the parameter of interest instead of population size). Typically, although the area must be closed (no emigration, birth, immigration, death) within a season (or year), the emigration, births, deaths, and immigration are allowed between seasons (or years).

Advantages

Mark recapture studies typically yield the most information about a given species within a given area when compared to presence/absence and distance studies.

Disadvantages

Mark recapture studies are often time consuming. They can be expensive depending on the number of visits needed per site per species and the type of method used to mark the animals.

Additional literature

For population estimates:

White, GC, DR Anderson, KP Burnham, and DL Otis. 1982. Capture-Recapture Removal Methods for Sampling Closed Populations. Los Alamos National Laboratory Publication. LA-8787-NERP. Los Alamos, NM.

Williams, BK, JD Nichols, and MJ Conroy. 2001. Chapter 14: Estimating Abundance for Closed Populations with Mark-Recapture Methods. In: Analysis and Management of Animal Populations: Modeling, Estimation, and Decision Making. Academic Press. San Diego, California.

With multi-year data collection:

For survival:

Williams, BK, JD Nichols, and MJ Conroy. 2001. Chapter 17: Estimating Survival, Movement, and Other State Transitions with Mark-Recapture Methods. In: Analysis and Management of Animal Populations: Modeling, Estimation, and Decision Making. Academic Press. San Diego, California.

Community Parameters

One of the more fascinating potential analyses to which the MSIM data may be applied would be community compositions. Are there species that always occur together? Species that never occur together even in appropriate habitat? Advanced analyses are still emerging (i.e.

MacKenzie et al. 2004), but typical analyses include estimates of species richness and species evenness. Species richness can be estimated from any of the protocols in this manual, but species evenness is dependent upon abundance estimates and can only be computed for taxa where abundance can first be estimated from the data.

Parameters

Species richness - number of species in a community.

Species evenness - incorporates the relative abundance of different species.

Data collection procedures

For species richness any of the protocols in this manual can be used to determine presence/absence for many species. Rarely are all species in a given area found.

Example

Any of the protocols listed in this manual should be able to be used in estimates of species richness. The bird point count and butterfly protocols are 2 examples that could be used in the estimation of species evenness.

Requirements & Assumptions

Often, estimates of community parameters require the assumption of equal detection probability between species. This assumption is impossible to meet. Unequal detection probability often results in the underestimation of the true number of species in a given area. Estimates do exist which relax this assumption (e.g. the Burnham-Overton jackknife (Williams et al. 2001) which requires only that individuals of the same species have the same detection probabilities). To meet this assumption, it may be necessary to search areas of equal size, with observers of equal skill, for equal amounts of time. To estimate species evenness, the assumption that no individual is counted more than once must be met. It would be easiest to meet this assumption by marking each animal encountered.

Advantages

If the parameter of interest is species richness, it will not be necessary to mark individuals (since it is usually easier to determine differences between species) reducing both the amount of time and money needed in the field. Estimates

can be made from a) spatial plot replicates (searching several plots within the same area) and b) temporal replicates (searching the same plot on multiple occasions). If temporal replicates are used, one must assume that the time between first and last visit is short enough to prevent the colonization/immigration or extinction/emigration of species.

Disadvantages

It can be difficult to determine what exactly, the parameters mean. If one property has more species than another but the other property with fewer species has endangered or rare species, how do you decide which property is really more important?

Additional literature

Krebs, CJ. 1999. Ecological Methodology, Second Edition. Benjamin Cummings. Menlo Park, California.

MacKenzie, DI, LL Bailey, and JD Nichols. 2004. Investigating Species Co-occurrence when Species are Detected Imperfectly. *Journal of Animal Ecology*. 73: 546-555.

Williams, BK, JD Nichols, and MJ Conroy. 2001. Chapter 20: Estimation of Community Parameters. In: *Analysis and Management of Animal Populations: Modeling, Estimation, and Decision Making*. Academic Press. San Diego, California.

Chapter Six: Reporting

The primary audience for the MSIM program is wildlife and public land managers, whether city, county, state, federal agency, NGO, or other organizations, that are responsible for the properties used in the program. This information will be important for making and defending management decisions. However, land managers are not the only people that need to have information from this program. The scientific community, general public, and political organizations will also be interested in the information. For this program to be successful, the data must be analyzed and presented at appropriate intervals in such a way as to be most beneficial to each of the audiences. To do this, the information gained from the analyses of the data will need to be presented in differing formats. Using different formats will allow for the content and detail to vary between audiences.

The National Park Service has provided skeletal guidelines for creating reports for different audiences, including a table outlining 8 types of reports which is both summarized and expanded upon below. This 4 page document is no longer in press, but can be retrieved from MSIM leadership team. For the purpose of the Iowa MSIM program, 2 of these 8 reports have been combined into 1, and the final report listed by the NPS (State of the Parks Report) has been dropped as this information would be covered in Iowa's annual report. Additional information can be found in Oakley et al. (2003).

Annual Administrative Report and Work Plan

The purpose of this report is to account for the use of funding and employee time on a yearly basis. It should include information on the yearly objectives, tasks, accomplishments, and products of the effort expended in a given year. It should also include plans and budgets for the next, up-coming year.

The primary audience for this report is departmental supervisors, agency program managers, and administrators. This report should be written annually and probably due in late January to allow for inclusion of all expenditures through the end of the field season for a given year as the Program is currently administered through a sub-recipient agreement between DNR and Iowa State University. Alternatively, if the Program is administered within an agency, it may be better to stick with the agency's fiscal year meaning that the report would include partial information from 2 years for the report, and the current and next year for the plan, covering 3 years total. Projecting budgetary needs across calendar years is, however, more difficult.

This report should be written by the program scientist with help from appropriate budget managers. It should be reviewed and approved by the departmental supervisor and may be used for Congressional or Federal State and Tribal Wildlife Grant oversight reporting.

Annual Report

This report would combine information from 3 of the categories listed in the NPS guidelines (Annual reports for specific protocols or projects, Inventory projects reports, and State of the parks report). The purpose of this report is to record annual data and report on yearly accomplishments, describe the current condition of select species, detail any changes to the protocols used for data collection, identify situations of concern, and highlight potential future research. The report would include information from all properties visited in the given year, but this information could be broken down into specific areas at the request of a land manager. Species lists, occupancy probabilities, detection probabilities and any additional parameters that can be computed on a yearly basis should be included for habitat class, region, county, and state-wide.

The primary audience for this report would be agency staff, scientists, and monitoring partners. The information should include graphics that could easily be pulled from the report (stand alone, in other words) for dissemination to the general public. The report would include information from the given year only for analysis, but may include summary statistics from previous years to help illustrate details. This report should be due before the beginning of the next field season (usually April 1).

The report should be written by the program scientist with help from the appropriate staff and monitoring partners. It should be reviewed and approved by the partners, including those chosen by the Iowa Wildlife Action Plan coordinator to serve on the subcommittees for the IWAP.

Periodic Analysis and Synthesis Report - Trend Analysis

The purpose of this report is to examine trends in the species occurrences (site colonization and extinction rates). In addition, the report should outline correlations to environmental conditions for each species, and should analyze the amount of change that can be detected with the current level of sampling. Any recommended changes should be suggested here, as to management actions and/or sampling effort changes.

The primary audience would be resource managers, staff, scientists, and monitoring partners, but as with the yearly report, stand alone graphics should be included which could be easily disseminated to the general public and legislative entities. This report should be written every 3 to 5 years and include all data acquired with the program in addition to comparisons to historical reports. It should also be written by the program scientist with help from the appropriate staff and monitoring partners. It should be reviewed and approved by the partners.

Program and Protocol Review Report

The purpose of this report is to provide a formal review of the program and protocols. It should review both the protocols used for each taxonomic group and habitat data collection as well as the products of the program to determine if changes are needed. This will help ensure quality assurance through the peer-review process. It should include any suggested changes to the program, including the number of habitats, sites, locations of plots, in addition to the protocols and outcomes.

The primary audience for this report is supervisors, administrators, the IWAP subcommittees, and monitoring partners. The report should be written and the program reviewed every 5 years. Again, responsibility for the report belongs to the program scientist with help from appropriate staff and monitoring partners.

The remaining 2 'reports' will depend on the quantity and quality of the data collected by the MSIM program to determine the frequency of production.

Scientific Journal Articles and Book Chapters

The purpose here would be to express knowledge gained as part of the program or to advance the program itself. The target audience would primarily be scientists and managers. The product would be peer reviewed by journal or book editors and would serve as part of the quality assurance aspect of the program.

Symposia, Workshops, and Conferences

The purpose of participating would be to review and summarize the information collected on a specific species as well as to provide publicity for the MSIM Program. It is expected that this would help identify current and future issues as well as sparking debate for new ideas. The audience and frequency would vary depending on the setting and volume of information collected.

In summary, the Annual Administrative Report and Work Plan as well as the Annual Report should both be compiled annually. The Periodic Analysis and Synthesis Report - Trend Analysis should be compiled every 3-5 years, and the Program and Protocol Review Report every 5 years. Journal articles and presentations should be done as often as possible given data and time constraints.

Chapter Seven: Periodic Review and Evaluation

The final element for a monitoring program is that of Periodic Review and Evaluation. In addition to internal review by the program scientist and the staff conducting the monitoring, external reviews should be made as well. This chapter outlines the potential protocol for conducting the external reviews. The information draws heavily for the National Park Service's "Peer Review Guidelines for the Inventory and Monitoring Program" which can be accessed at <https://irma.nps.gov/DataStore/Reference/Profile/2253227> (last accessed on May 15, 2025) as well as the Oakley et al. (2003) publication.

Definitions

Peer review - Report is reviewed by scientific reviewers and technical experts.

Internal peer review - Review is conducted by DNR staff, chosen staff should have no direct involvement with the program (i.e. do not collect or analyze the data).

External peer review - Review is conducted by independent experts from outside the DNR. For our purposes, these reviewers will most likely be members of the IWAP taxonomic subcommittees.

Reviews should be conducted periodically and should include review of individual protocols, sampling location selection procedure, and findings of the program. It may (or may not) be necessary to appoint a coordinator for the review process to either ensure objectivity or ensure that the most qualified experts are chosen as the reviewers.

This is a formal process and requires the maintenance of written files and approval forms signed by the reviewers. This will serve as the administrative record of review and is to include the original document, instructions to reviewers, reviewer comments, documentation as to how the authors responded to the comments, the final copy of the document, and the coordinator-signed approval form.

Peer reviewers should be chosen based upon expertise in the area and should be able to independently and objectively comment on the document and merit of the work. Therefore, they should not be regularly involved or have a vested interest in the project under review. The panel should include people that are not employees or supervisors of program personnel or product authors. It will be critical to include external reviewers in this process.

Types of Reviews

Annual Report - should be reviewed by at least 1 internal and 2 external peer reviewers before being submitted. One of the reviewers should have a statistical background. It may be best to request reviews by taxonomic sections which would increase the number of reviews but also increase the number of reviewers with expertise in a given area. Comments received from others (i.e. monitoring partners, including the subcommittees for the IWAP) after it is submitted should be considered and incorporated into the next annual report. It may be advisable to request friendly reviews from a select few of the partners before the report is submitted.

Periodic Analysis and Synthesis Report /Trend Analysis - should be reviewed by at least 2 external peer reviewers per taxonomic group as well as a statistician for the whole report. Monitoring partners should also be given the opportunity to comment on the report before it is considered final.

Program and Protocol Review Report - should be reviewed by at least 2 external peer reviewers per taxonomic group as well as a statistician for the whole report. The IWAP subcommittees should also be given the opportunity to comment on the report before it is considered final.

Guidelines to the scientific peer reviewers should include (but not be limited to):

1. Are the objectives clearly defined and reachable?
2. Is the sampling and experimental design appropriate? Did or will it meet the program objectives? Is it

statistically valid?

3. Are field techniques clearly described and sufficient to meet program objectives?
4. Are analytical and statistical procedures clearly described and appropriate?
5. Were analytical and statistical procedures used appropriately?
6. Are the results and conclusions logical?
7. In addition, for future plans:
 - a. Does timeline ensure that objectives will be met?
 - b. Are reports and other products identified and adequate?
 - c. Is the combination of scientific disciplines proposed sufficient to adequately meet the objectives?

Guidelines to the non-scientific reviewers should include (but not be limited to):

1. Is the report understandable and easy to read?
2. Does the report adequately describe the objectives, properties, how the data were collected and analyzed?
3. Are the conclusions logical?

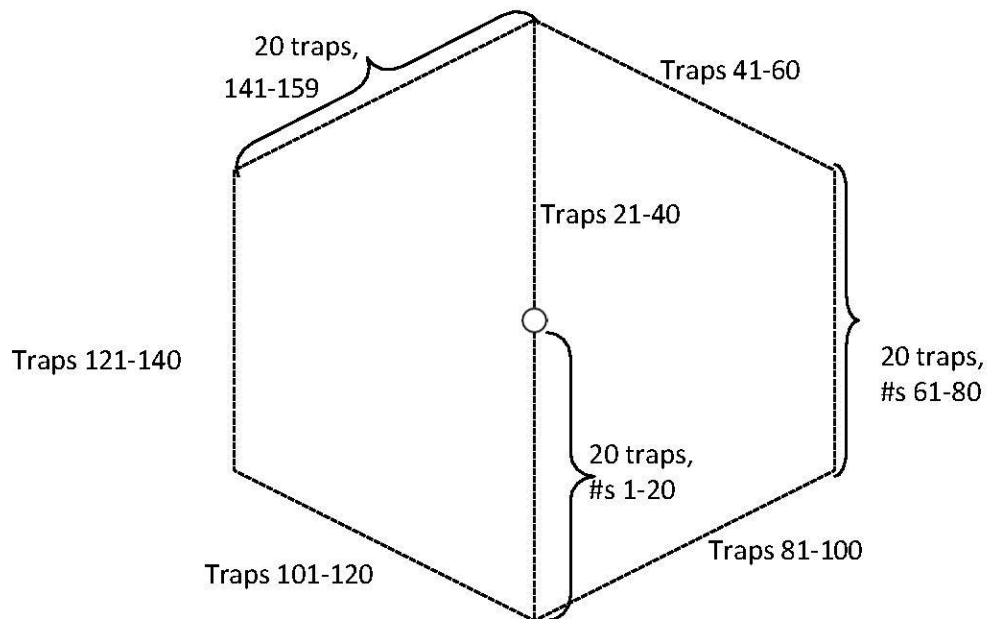
The files maintained as part of the review process should be in the possession of the program scientist (or a copy of these files), with another copy (or the originals) being maintained by the review coordinator. For Iowa, it would be appropriate for the IWAP coordinator to fill the role of MSIM review coordinator, if the coordinator was willing. Additional possibilities for the review coordinator include the Wildlife Bureau Research Supervisor or someone appointed by this supervisor. Examples of report review forms, including the coordinator's form, the scientific peer review form, and the non-scientific peer review form exist.

Chapter Eight:

Mammal Monitoring Protocol for Small, Medium, and Large Mammals

Small Mammal

At each hexagonal sampling plot, the spacing between the traps should be 10 m with the total number of traps to equal 159 per site, arranged around the arms of the hexagon and along the center transect. Trap number 1 should be located 10 m north of the most southern tip of the hexagon, this will allow traps numbered 20, 40, 60, 80, 100, 120, and 140 to be placed at the PVC poles that delineate the hexagon (the bird point count locations 1, 2, 3, 4, 5, 6, and 7 respectively). Using more traps than the FS MSIM program will still allow for comparisons between the 2 programs, as the Iowa data can be truncated so that only data from traps that would match the FS protocol is used for comparisons between the 2 programs, should that need ever arise.



It is important to use the hexagonal transect trap lines for two reasons. The first is to allow these data to be compared to other studies that have followed the same protocol. The second reason is that previous research has indicated that a transect method is more efficient for a basic species inventory when density or abundance data are not needed (Jones et al. 1996). This increase in the number of species encountered is due to the greater area sampled by a line transect as opposed to a grid (in which area of trapping efficiency for any given trap most likely overlaps that of the adjacent traps). Indeed, Pearson and Ruggiero (2003) have found that linear transects of 25 traps resulted in significantly more captures of individuals than did 5 x 5 trapping grids. The linear transects also captured an additional two species not seen in the grids (Pearson and Ruggiero 2003). However, the MSIM leadership team may deem some properties as not feasible to trap in the normal way. In these occasions, the alternative configuration will be recorded. These arrays will maintain 159 traps placed at least 10 m apart.

Survey Methods

Trapping can begin in April through the middle of October based on weather conditions. Under extreme cold (<20 F at night) or hot (>100 F) conditions, trapping will be paused and will begin again once conditions return to acceptable conditions. When temperatures fall below 45°F at night, cotton balls should be provided in the trap for nesting material and crews may want to switch from peanut butter to Crisco bait. In temperatures above 90°F during the day, care should be taken to ensure traps are shaded (e.g. trap forested areas) and crews should ensure all traps are checked no later than 3 hours post sunrise. Longer periods of time off may be taken, for example Iowa MSIM takes the month of August to conduct vegetation surveys which may not allow for the time needed to complete small mammal trapping. In non-standard trapping arrays, GPS location should be recorded for each trap or a GPS track for the entire trapping line. Traps should be numbered before being set and care should be taken to ensure that traps are numbered consecutively.

All traps will be marked with a flag to allow for every trap to be found every day during the trapping event. We typically use peanut butter as bait, knowing that the scent of the bait can attract mammals. Opportunistically, a crew may choose to supplement the peanut butter with carrots, apple slices, rolled oats, or lard/Crisco, depending on availability.

Traps are opened for 3 nights and checked twice daily after the first night, once in the morning and again in the late afternoon. If no animals are captured in the afternoon trap check and the habitat is not suspected to support vole populations, the afternoon trap check can be skipped after the second trap night (be sure to check traps each morning). Bait should be consistent over time and sites as it influences the capture rates of small mammals. Traps should be checked for the last time and removed such that this timing matches that of setting the traps on the first day. No 'seasonality' in captures is expected. This means that it is expected that all small mammals will have similar probabilities of being active over the entire trapping season within a given species. Therefore, each sampling property is trapped for only one session within a given sampling year.

Captured animals are identified, sexed, aged, examined for breeding status, weighed, measured, and marked. Individual marks are administered by using a Sharpie permanent marker to place a number or letter on the belly (or some other easily read location) of the animal. Colors of marker to be used include purple or blue. Other colors (green, brown, red) can be misidentified at a later capture during the week, i.e. is that brownish-red smear on the stomach a former number or fecal/blood material? Be sure to dry the stomach fur before writing the number on the fur/skin of the animal. If the mark is placed on wet or damp fur, it will smear. If it is raining, a crew may choose to become more creative in the individual marks (e.g. blue left ear and purple right ear = #12) to aid in future recognition of individual animals.

Measurements include total length, tail length, hind foot length, and ear length.

The number of traps that have closed without capturing an animal or are missing bait but open are also recorded as either 'tripped but empty' or 'open, no bait'. This information is important in assisting with calculating species abundance due to the need to know how many traps were available to capture animals. All dead animals are collected, frozen, and donated to a museum or mammalogy class. At properties that will be visited every year, it may be preferable to mark small mammals with ear tags as opposed to a Sharpie marker as it is possible that these animals will be captured between years. Shrews would not be given a permanent mark, even on properties trapped yearly, as this would be additional stress on extremely sensitive animals. Shrews would be marked only with a Sharpie marker or by shaving patches of fur.

Dirty traps should be sprayed with Lysol before being placed into the enclosed truck bed. Never place used traps in the enclosed cab of a vehicle with the driver. Dirty traps are cleaned by being placed into either a mild bleach or a 5% Lysol solution in 30-gallon trash cans (if many traps) or smaller sized pails (if fewer traps). Soiled traps are scrubbed. Traps are rinsed with water and allowed to dry completely before the next use. Bedding (used cotton balls) is soaked in the cleaning solution for 10 minutes before disposal. Bedding and bait are thrown away. Technicians need to always carry extra traps with them in order to replace traps that are missing, damaged, or excessively dirty.

Medium & Large Mammals

Camera traps

For medium and large mammals, trackplates and camera surveys were the primary method of detection through 2008. Trail cameras continue to be used in this project, with 2 to 3 cameras per property based on availability. The cameras are attached to a tree. If no suitable trees are found, then cameras can be attached to stakes. Stakes MUST be able to withstand weather and animal activity. Each camera survey period encompasses a 10-day period, which can occur throughout any point in the year. Each site is visited at least once to check battery life and SD card storage.

Camera stations use a basic digital trail camera with infrared flash. The camera should be attached to a tree or some other immovable substrate. Cameras are attached to the tree using a tripod, wires, nylon straps, and/or duct tape. Batteries and flash cards are checked and replaced as needed (Manley et al. 2004). Cameras can be placed throughout the property, but should try to minimize accidental triggers (e.g. tall grass in the wind, high traffic area for cars or people). Instead finding a game trail or stream bank may provide a better representation of mammals on the property.

All photos should be identified to species and entered onto the data sheet. However, select photos will be saved using the following template: Speciesname_property_date_Survey_photographerinitials(optional).jpg

For example: Coyote_McCoyWMA_5_5_2015_TC_PF.jpg would indicate a coyote at McCoy WMA on May 5, 2015 on a Trail Camera and identified by Paul Frese, and uploaded to the MSIM Photo database.

Visual Encounter Surveys (VES)

In 2008, it was determined that a wider variety of tracks were being recorded as part of the visual encounter surveys than were being collected via the trackplates. In 2009, we formally compared the 2 methods for recording tracks: 1) the traditional trackplates used 2006-2008, and 2) timed visual encounter surveys for tracks. We documented more medium and large size mammals with the track visual encounter surveys and therefore no longer employ the track plate method.

Each property should be surveyed for tracks for 1 hour of Visual Encounter Survey effort each of the three seasons (spring (April & May), summer (June & July), & fall (September & October)). As much as possible, these surveys should follow rain events within a few days in order to allow time for tracks to accumulate in muddy areas but not so long that the tracks become obliterated. These visual encounter surveys should also include sightings of live mammals, burrows, scats, skeletons, and any other signs that can be identified to a mammal. The species and location of the mammal sign will be recorded.

Photos should be taken of each new track on each property. Photos should contain a reference object for size comparison. Photos should be labeled with date, property, and site before uploading them to the online MSIM photo database.

Environmental Data Collection

Environmental variables such as air temperature, wind speed, temperature and other weather conditions should be recorded at the time of each survey on the faunal monitoring data sheet.

Equipment List

Per site:

Small mammal protocol:

- 159 Sherman traps, plus replacements
- Bait packs (peanut butter wrapped in wax paper)
- Surveyor's tape
- Compass
- Polystyrene batting (or cotton balls in non-zip sandwich bags) if trapping in cold weather
- 1 gallon plastic bags
- 2 scales up to 300 grams
- 2 mammal field guides
- Latex or rubber gloves
- Leather gloves for each crew member
- Backpacks or containers for carrying traps
- 2 hand lenses
- Hand sanitizer
- GPS unit to record trap locations
- Lysol

Meso- & large mammals:

- Track VES: Stopwatch
- Camera
- Ruler
- Camera stations: 2-3 game cameras and SDcards, 2-3 wires or cables
- 100 feet of 22-gauge bailing wire OR airplane cables
- Camera batteries

Standard field kit: Clip board, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets.

SMT Clean up:

- 2 30-gallon garbage cans or other plastic containers to use as sinks
- Water supply
- Bleach or Lysol
- Hose with nozzle
- Scrub brush
- Protective eyewear
- Rubber gloves
- Garbage bags for garbage

Staff & Training

Training is recommended and should include 1) field guide use and identification, 2) discussion of defining species characteristics, 3) practice of trap setting and animal handling in a variety of environmental conditions (rain, heat, etc.), 4) track and scat ID, 5) set up and maintenance of camera stations. Crews may need a reference guide of most common species' tracks for the clipboards.

Data Quality & Management

Small mammal data can be affected by:

- Trap placement: Should be checked periodically by supervisor. Where possible, traps should be placed adjacent and parallel to downed woody debris (can serve as runways) or underneath tall grass (can serve as shelter).
- Observer handling care: Mortalities can be monitored through data, and should be <1%.
- Error in species ID: Difficult to monitor, therefore, could switch observers during week of trapping or collect vouchers of suspect species. Can also photograph animals and send photos for confirmation of some species.

At the end of each trapping day, field crews should review data sheets to ensure all information is present. At the end of the week, the field crew member responsible for this protocol should review the data sheets for species identifications, escape and mortality rates, trap function, and legibility. Data should be entered as soon as possible and is expected to be entered before the end of the season. Each trap-checking session is entered as a different survey (e.g. Tuesday AM check = visit #1; Tuesday PM check = visit #2; Wednesday AM check = visit #3; and Thursday AM check = visit # 4). The track VES and camera station data are 'independently verifiable and the data are subject to very little interpretation' (Manley et al. 2004, MELP 1998) as long as photos of each track are also documented. This means that there are very few sources for technician errors within this protocol design, therefore there is no need for separate quality assurance teams. However, the set up of the camera stations is critical and should be spot-checked by the crew supervisor periodically throughout the season.

County lists of species are submitted to the IWAP Mammals Subcommittee in January of each year. Any additional information requested by the Subcommittee is to be supplied in a timely manner. Any record rejected by the subcommittee is changed to 'unknown mammal' in the database with notes in the comments as to the original identification.

Data Analysis

The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of sites.

Using a closed population model, such as a Lincoln Petersen estimation method, could allow for the estimation of population size, provided that small mammals are marked and recaptured during the primary sampling period. For more information, see the Data Analysis protocol.

Photos will be archived for species identification verification. Detection probabilities and the number of properties with detections can be evaluated to determine if the number of camera stations per site should be adjusted.

Safety Considerations

Small mammals carry many diseases which can be transferred to humans. Technicians should be aware of the potential diseases and the associated symptoms. Leather gloves are provided for small mammal trapping and it is expected that technicians will wear gloves when handling any animal. Dust masks are to be provided for those who wish to wear them. Normal hygiene, i.e. hand washing, not rubbing face before hand washing, should be followed at all times. It is also possible that a technician may encounter a feral dog or other potential hazard; therefore maybe pepper spray should be carried, should the technician so choose. Crew members should carry cell phones. When cleaning traps, crews should wear rubber gloves and goggles to prevent bleach/Lysol getting into cuts on hands or being splashed in the eyes.

Target Species

The following list of target species represents the species of greatest conservation need as chosen by the Steering committee for the 2015 Revision of the Iowa Wildlife Action Plan (Reeder and Clymer. 2015). Bats are considered in a different chapter and are therefore not included in the following list. Distribution maps for these species can be found in Mammals of Iowa (Bowles 1975) and also in Iowa GAP (Kane et al. 2003) (except for the red-backed vole which was not included in GAP due to a lack of reliable, recent data (Erv Klaas, personal communication)). Appendix 1 contains a list of additional, more common, mammal species which may also be encountered during the monitoring efforts. An asterisk (*) indicates species considered to be Data Deficient in the IWAP.

Target species:

Common Name	Scientific Name	Habitat
Hayden's shrew*	<i>Sorex haydeni</i>	Grassland, woodland, riparian
Elliot's short-tailed shrew*	<i>Blarina hylophaga</i>	Forest, woodland, savanna, grassland
Southern short-tailed shrew*	<i>Blarina carolinensis</i>	Forests, shrublands, grasslands, wetlands
Least shrew	<i>Cryptotis parva</i>	Woodland, savanna, grassland, riparian
White tailed jackrabbit	<i>Lepus townsendii</i>	Shortgrass prairie & pasture
Franklin's ground squirrel	<i>Spermophilus franklinii</i>	Tallgrass prairie & roadsides
Southern flying squirrel	<i>Glaucomys volans</i>	Forest
Plains pocket gopher	<i>Geomys bursarius</i>	Prairie
Plains pocket mouse	<i>Perognathus flavescens</i>	Prairie, sand & loess
Southern bog lemming*	<i>Synaptomys cooperi</i>	Moist grassland
Woodland vole	<i>Microtus pinetorum</i>	Forest
Gray Fox	<i>Urocyon cinereoargenteus</i>	Forest, woodland
Long-tailed weasel	<i>Mustela frenata</i>	Generalist found in all habitats near water
Least weasel	<i>Mustela nivalis</i>	Meadows, fields, shrubby areas, and open woodlands
Spotted skunk	<i>Spilogale putoris</i>	Grassland, forest, farmsteads
Ermine	<i>Mustela ermine</i>	Shrubby or woody areas near water

Additional Methods for Special Locations

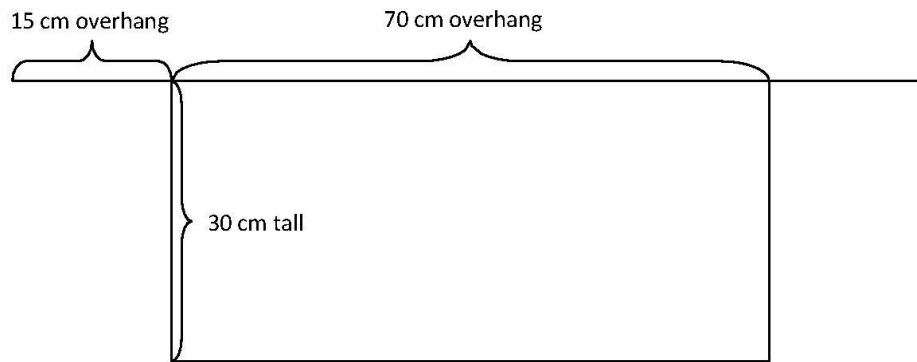
The following are additional techniques which may be implemented at certain properties *in addition* to the core methods described above. These could be used in areas where there are known populations of species of concern or when supplemental funding has been acquired for a given area. However, the basic core protocol must still be followed to allow for comparison of all sites, both across the state of Iowa and also for a regional comparison, provided that other states or areas are following the same protocol.

Trackplates

The trackplate covers are wooden boxes. The final dimensions of most of the track plates are 70cm long x 30 cm wide x 30 cm tall. The bottom tray is made of a piece of aluminum flashing that is 70 cm long x 29.5 cm wide. The track plate can be attached to the bottom of the box using Velcro (Drennan et al. 1998) and the contact paper can be attached to

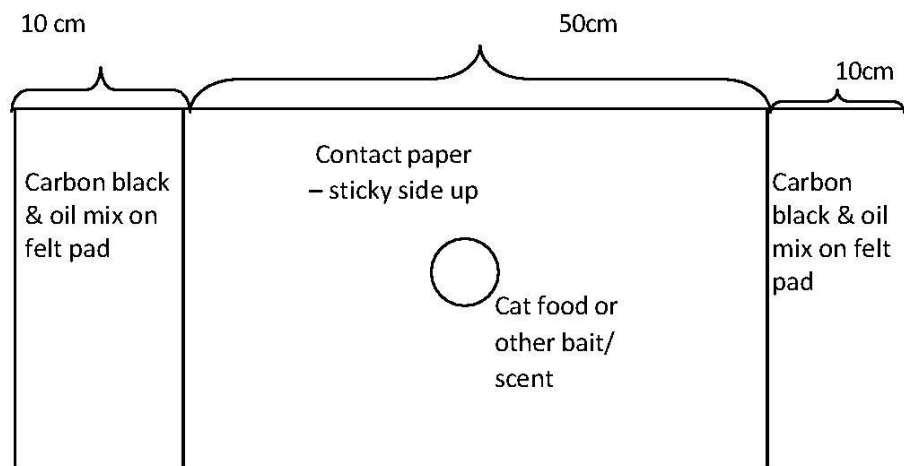
the bottom plate using poster putty or tape. The front & back of the box are unobstructed.

The top piece should be longer to allow each opening to have a 15 cm “overhang” to prevent rain splatter.



Side view of a trackplate box.

Plates are covered with a carbon black (same as newspaper ink; from a Xerox machine) mix on felt pads and CONTACT paper (sticky side up) which is used to record the tracks (Manley et al. 2004, MELP 1998) as illustrated in the diagram below:



Track plate for bottom of box.

There are several ways to coat the track plate. For this project, a felt pad covered with Xerox carbon black (mixed 1 to 1 with paraffin oil or mineral oil) can be attached to the track plate (Weiwel 2003). Alternatively, a mix of 1 part newspaper ink (Xerox carbon black) and 1 part mineral spirits (or paraffin oil) can be applied to the plate using a roller brush (Lord et al. 1970) or that area could be sprayed with a mixture of 1 part blue carpenter’s chalk mixed with 2 parts alcohol (Drennan et al. 1998). Plates should be baited with cat food, scent, or some other appropriate attractant. Old bait is packed out and the track plate is changed if tracks are detected or plate has been damaged by rain. A large feather could be hung approximately 1.5 m above the trap, perhaps on a pole, to act as a visual cue (Manley et al. 2004). The contact paper is covered with clear tape before being removed from the plate bottom and stored with the data sheet.

Numbers of animals estimated from the trackplate method have received mixed reviews as to their correlation with actual population size estimates, with some authors finding high correlation between the two methods (for sciurids: Drennan et al. 1998) and no correlation (for raccoons: Smith et al. 1994). The focus of this protocol, however, is to document species presence, not necessarily to determine population sizes.

Sherman Trap Array Augmentation

1. Trap for longer duration.
2. Increase the number of traps (and therefore, the size of) the trapping array.
3. Arboreal small mammals (i.e. Southern flying squirrels) are best trapped in trees, so in forested areas, could place additional Sherman traps on shelves attached to trees.
4. The IWAP Mammal Subcommittee has suggested pre-baiting and pre-setting traps, especially in areas which may support populations of southern bog lemmings or red-backed voles. To do this, popsicle sticks would be used to lock the Sherman traps open, and each trap would be baited. Traps would be placed in the proper location 2 to 14 days prior to the start of the trapping session. Bait would need to be replaced periodically.

Camera Array Augmentation

1. Camera locations - Some species, such as bobcats or coyotes, are believed to avoid bait stations. For these animals, it may be best to place camera locations along travel routes.
2. Sampling intensity - If needed, the number of stations could be increased or the sampling duration could be extended.
3. Visual attractant - A large feather can be hung 1.5 m above the ground to act as a visual cue.

Track plate Augmentation

1. Sampling intensity - If needed, the number of stations could be increased or the sampling duration could be extended.
2. Polyethylene enclosures - In areas with heavy precipitation, the track plates boxes should be covered with polyethylene.
3. Open track plates - For species that are less likely to enter the enclosed track plates or are too large to enter the enclosures. Open track plates consist of 1 square meter of metal, covered with soot, with the bait placed in the middle. These are less effective due to rain, fog, and other weather which can wet the tracks. Alternatively, to attract larger animals, such as coyotes, a patch of ground could be bared, tilled, wetted to create mud, and a white disk (not bait) placed in the center to serve as an attractant. The site should be visited every other day and a cast of the tracks could be made using plaster. This bare-earth method, with a scent tablet for bait, has also been used for raccoons (Smith et al. 1994).

Tomahawk Live Trapping

Established and used during the small mammal monitoring protocol, a tomahawk trap can be placed within 3 m of every 4th Sherman trap for a total of 40 traps with 5 being on each transect of the hexagon. Traps are baited with the oat/seed mixture used for the Sherman traps, along with apples, alfalfa pellets, and an open can of tuna or cat food. Traps are checked twice daily. The data sheets should include boxes for each trap - to be checked off when the trap is checked with each visit to ensure that no tomahawk traps are missed. Animals are identified, sexed, and released. Due to the low capture probability it may not be feasible to mark animals, although this could be done by using colored hair spray.

Tomahawk trap cleaning should follow the same protocol as for the Sherman traps. In addition, should a trap be sprayed by a skunk, that trap can be washed with a mixture of baking soda and hydrogen peroxide.

Tomahawk equipment needed includes 40 Tomahawk traps, plus a few extra as replacements, trap bait, knife for apples, plastic bags, rulers, mammal field guide or key, scissors, paint or colored hair spray, large garbage bags, rubber gloves, leather gloves, capture cones, backpacks for traps.

Tomahawk staffing, skills, & training: An additional crew member may need to be added to each crew if tomahawks are used. All employees involved with the tomahawk trapping should be vaccinated for rabies.

Pitfall Traps

Especially useful for capturing shrews and gophers. Pitfall traps often cause large mortality rates for small mammals, however.

Spotlight Visual Counts

This method is reliable for some but not all species of ungulates and lagomorphs. Errors with detection probabilities can occur due to vegetation, terrain, or species habits (such as being secretive, solitary, etc).

Molecular DNA

Scat, hair, or blood samples could be collected for species identification. Where morphologically similar species occur together, molecular ID would be best for distinguishing the species. Field technicians would need to be trained to collect sufficient amounts of hair, blood, or feces in a manner which protects the DNA from both degradation and contamination.

Chapter Nine:

Bat Monitoring Protocol

Bat Monitoring

Bat detections were done primarily using ANABAT detectors between 2006-2022. Currently we use Song Meter SM4BAT from Wildlife Acoustics. Walking surveys are conducted by 2 technicians through various habitats. These walking surveys (or ANABAT Surveys) shall be done on 3 occasions (once May 15 to June 15, once June 16 to July 15, and once July 16 to August 15). Each survey will be at least 2 hours in duration (of recording time). Alternatively, stationary placement of detectors on a property could be done following the USFS NABat program (Loeb 2015). MSIM started to implement stationary detectors in 2025 at a handful of properties.

Survey Methods

Walking Survey

Two technicians will carry a SM4BAT while they walk around the property. These surveys can start between a half hour after sunset and go until about 2 am. Technicians should walk through a variety of foraging habitats. Good areas to survey include forest edges, around buildings or lights, roadways through forest (as long as road is safe to walk), but surveys can happen anywhere on the property. The general locations that technicians should be noted on the datasheet. Alternatively, a GPS route could be taken for each survey.

These surveys should be completed on 3 occasions during the summer (May 15-Aug 15), although dates for the summer may differ with the potential change in bat behavior. This time frame should reflect the breeding and roosting of most species in the state. Each visit should be at least 1 week apart.

Operating SM4BAT

- Slide the power switch from EXT to INT (for internal battery)
- Attach the microphone to the SM4BAT. Turning the grey locking ring on the microphone or cable clockwise firmly until it locks into place.
- Press check status to verify time, microphone connection, SD memory, and battery.
- Verify the recorder is turned on and displaying the main menu. If the recorder is sleeping, press and hold SCHEDULE STOP to wake up.
- To Start recording
 - Quick Start
 - record Always
 - Overwrite schedule? = Yes
 - Press SCHEDULE START to begin
- As data are being collected the screen will show “Currently recording” and light will blink green and red.
- Press and hold SCHEDULE STOP when done
 - Slide the power switch from INT to EXT to turn off unit.

Stationary Survey

At each property, one SM4BAT will be set up to automatically record bat calls. The detectors should be placed within 50m of BPC1 in the best habitat. Best habitat is considered to have limited dense vegetation, not directly next to a roost, and not directly over hard surfaces like still water or pavement. The detector will be set out for 2 survey events between May 15 and August 15. Each survey event consists of 4 consecutive nights of automated detection and survey events should be separated by at least 10 days. Ideally each location is surveyed once between May 15 and June 30 and once between July 1 and August 15. Each crew should have at least 2 SM4BAT Units to meet this timeline.

The detector should be set to record the entire night from 15 minutes before sunset to 15 minutes after sunrise (Loeb et al. 2015). Survey events should try to occur in warm temperatures, low wind, and little to no rain.

Operating SM4BAT

- Slide the power switch from EXT to INT (for internal battery).

- Attach the microphone to the SM4BAT. Turning the grey locking ring on the microphone or cable clockwise firmly until it locks into place.
- Verify the recorder is turned on and displaying the main menu. If the recorder is sleeping, press and hold SCHEDULE STOP to wake up.
- Attach the GPS module and wait until for it to get the GPS coordinates. Once the SM4BAT says “[FIX]” on the home screen the coordinates have been received and stored in the device. Once FIX is on the screen, remove the GPS module.
- Press “check status” to check microphone connection, and SD memory and battery levels.
- Schedule should already be set to run.
- 15 min before sunset to 15 min after sunrise.
- To start recording press SCHEDULE START - it will display a screen when the device will be asleep until (15 min before sunset).
- Set up the detector (see DETECTOR SETUP instructions below).
- After 4 nights come back to take apart detector set up and turn off the device.
- Press and hold SCHEDULE STOP when done (main menu will appear).
 - Slide the power switch from INT to EXT to turn off the unit.

Detector Set Up

- Once the detector is ready to record, you can set up the station.
- Using a tripod that extends to at least 1.4 m (about 4.5 feet), attached the microphone head to the top of the tripod using Velcro zipties or standard zipties, making sure it is secure. The microphone should be attached to the detector prior to this point, to make sure the connection is working.
- Extend the tripod to the maximum height. Making sure to extend the legs and center pole. Tighten all of fixtures, so it stays in place.
- Secure the tripod to the ground by attaching 2 bungee cords to anchors in the ground. Using metal spiral anchors provides a strong hold in a variety of soil habitats. Make sure the bungee cords are placed in at least 2 different directions to provide counter balance to the tripod.
- Lock the SM4BAT to a tree or tripod (tree is preferred if close by)
 - Tree: Feed a python lock through the detector at the top and loop the lock ends around a tree. Tighten the lock until the detector is snug against the tree.
 - Tripod: Feed a python lock through the detector at the top, leaving the locking mechanism very close the device. Place the device on one of the inner supports and cross the end of the python lock over and through the bottom of the detector, creating an x over the center pole of the tripod. Tighten the lock to ensure the detector has limited movement.
- Finally secure down the extra microphone cable and python lock end to reduce noise.

Environmental Data Collection

Environmental variables such as air temperature, wind speed, and other weather conditions should be recorded at the time of the survey on the Bat Acoustic Survey data sheet. In addition to the above data, potential roosting sites seen on the property should be noted on the data sheets. These will most likely be caves, hollow trees, large dead trees with loose bark, etc.

Equipment List

Acoustic Equipment - Walking

SM4BAT FS with microphone attachment

Extra batteries (D batteries)

Extra data memory cards GIS

Headlamps

Standard field kit: Clip board, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets.

Acoustic Equipment - Stationary

SM4BAT FS with microphone

Extra batteries (4+ D batteries)

Extra data memory cards
 Garmin GPS module
 Tripod
 Bike/Python Lock
 Additional Lock for SM4BAT
 Bungee cords (18-24")
 Landscaping anchors (metal screw anchors are most universal)
 Extra zipties
 Standard field kit: Clip board, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets.

Staff & Training

SM4BATs can be utilized by any field crew. A lab technician will be needed to analyze calls recorded with the SM4BATs detectors.

Data Quality & Management

SM4BAT

Correct equipment set up is crucial to ensure that the calls are actually being recorded and stored. As the calls are automatically recorded by the SM4BAT, it will be easy to store the data for future scrutiny. All calls should be kept on suitable electronic storage (hard drives, cloud storage, etc). For species identification purposes, these calls will be compared to calls of known individuals to determine species.

At the end of each survey, field crews should review data sheets to ensure all information is present. For additional data quality control, hand vetting of acoustic recordings can be done by trained professionals.

Data Analysis

The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of areas occupied using Program R. See chapter 5 (Data Analysis) for more information.

Safety Considerations

The SM4BAT survey technicians should work in groups of at least 2, as this work will be done late at night, after hours for most businesses. These technicians should also carry a cell phone, GPS unit, maps, and first aid kit, in addition to flashlights or headlamps and possibly a hard hat if working in a forested or rocky area. These crews should also have a sign in/sign out system so that someone is aware of their location and status.

Target Species

The following list of target species represents the species of greatest conservation need as chosen by the Steering committee for the 2015 Revision of the Iowa Wildlife Action Plan (Reeder and Clymer. 2015). Distribution maps for these species can be found in the Distribution and Biogeography of Mammals of Iowa (Bowles 1975) and also in Iowa GAP (Kane et al. 2003).

Appendix 1 contains a list of additional, more common, bat species which should also be encountered during the monitoring efforts. An astrisks (*) indicates species designated as Data Deficient.

Target bat species:

Common Name	Scientific Name	Habitat
^A Northern long-eared bat	<i>Myotis septentrionalis</i>	Forest, woodlands
Little Brown Bat	<i>Myotis lucifugus</i>	Woodland near waterways, readily rears young in buildings and bat houses
^A Indiana bat	<i>Myotis sodalis</i>	Forest, upland, & riparian
Silver haired bat	<i>Lasionycteris noctivagans</i>	Woodland edge, riparian
Eastern pipistrelle	<i>Perimyotid subflavus</i>	Woodland edge, riparian

Common Name	Scientific Name	Habitat
Evening bat*	<i>Nycticeius humeralis</i>	Forest, riparian

*Federally listed under the Endangered Species Act.

Additional Methods for Special Locations

The following are additional techniques which could be implemented at certain sites *in addition* to the core methods described above. These could be used in areas where there are known populations of species of concern or when supplemental funding has been acquired for a given area. However, the basic core protocol must still be followed to allow for comparison of all sites, both across the state of Iowa and also for a regional comparison, provided that other states or areas are following the same protocol.

Roost Site Monitoring

If the target species have potential roost sites (bridges, caves, etc) within the sampling unit, it may be beneficial to monitor these in addition to mist netting at habitat locations. Field visits will be needed to search for potential roost sites and determine the best location to watch bats exit the roost. If cave-like structures are identified, 2 or more observers watch the site and count the number of bats leaving using hand held counters. If species cannot be identified as they exit, additional techniques will have to be utilized to capture bats as they leave (or search the cave).

Roost under bridges can be inspected using a flashlight with a red filter at least 3 hours after sunset. Attempts can be made to count the number of bats seen and if species identification cannot be made, attempts to catch an individual of each un-identified species/cluster will need to be made.

Remember, it is the species that is of interest more than the number of individuals so emphasis is on species identification. In either case, at least 2 visits separated by 1 week need to be made to each potential roost site.

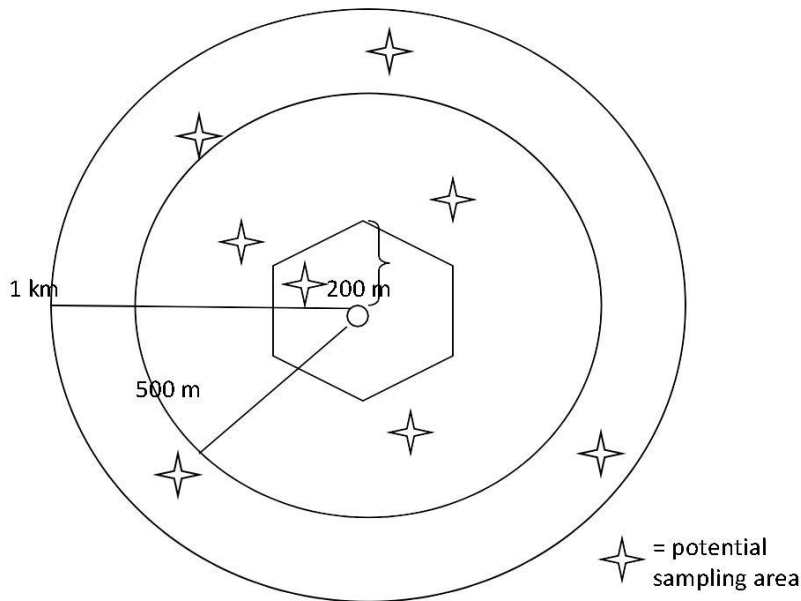
Due to the threat of White-Nose Syndrome, it is highly unlikely that this technique would be implemented through the Iowa DNR MSIM Program. Instead, any suspected roost sites should be reported to the Iowa DNR Threatened and Endangered Species Coordinator (Kelly Poole) for further examination.

Bat Trapping Surveys

At each location chosen for sampling, between 2 and 5 mist nets (depending upon area to be covered & number of technicians) are used. Nets are opened at sunset and operated for 3.5 to 5 hours, being checked frequently, at least every 20 minutes (USFWS 1999). Leaving bats in nets can result in injury to the bat or the bat chewing through the net and escaping (MacCarthy et al. 2006). Care should be taken to prevent any unnecessary disturbance near the net site which may influence capture probability.

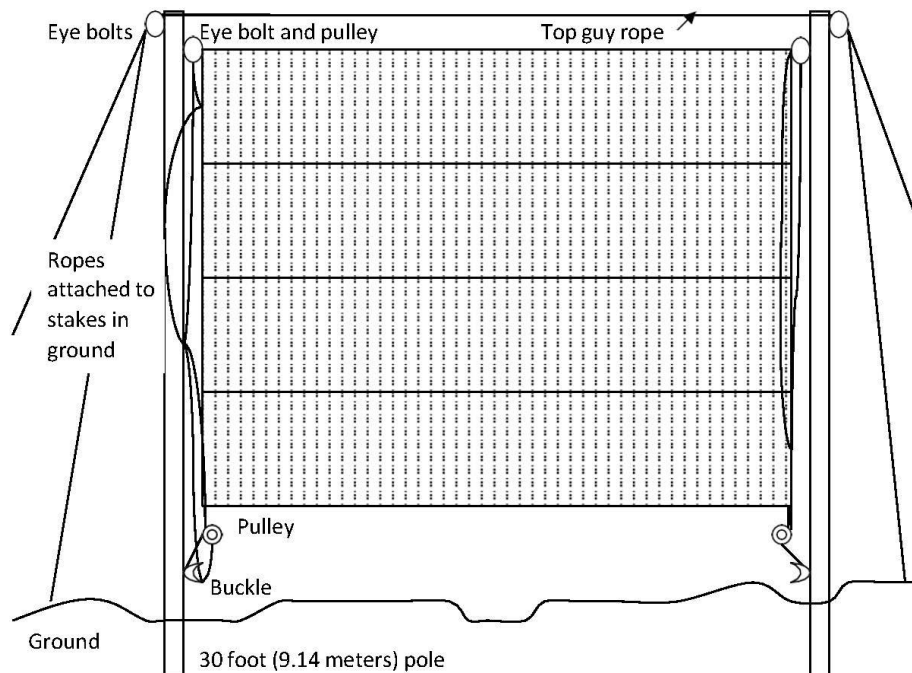
Net mesh size should be the smallest available, approximately 38 mm (1 ½" x 1 ½") openings, although 50 mm (2" x 2") can also be used. Nets should be placed such that they stretch perpendicularly across the corridor opening. Nets should cross the corridor/stream completely and reach from the stream/ground level to the canopy. This set-up often includes 3-4 nets stacked such that the nets reach a height of 7 m (22.97 feet).

Inclement weather conditions, including temperature below 10°C (50°F), rain, and strong winds should halt or prevent trapping efforts. Bats may also avoid nets or be less active on bright moonlit nights (USFWS 1999).



Information recorded at each location will include ambient temperature (if temperature changes significantly during the 3.5 to 5 hours of net operation, this needs to be recorded as well), wind, and cloud cover. Information on each captured bat to be recorded includes: time of capture and net of capture, species, reproductive status, age, and forearm length. The ear, thumb, tragus, and foot length should all be recorded and the calcar should be checked to determine keel.

Gear decontamination protocols: White-nose syndrome has been detected in Iowa (K. Poole, personal communication, 1/2015). Appendix 3 contains full decontamination protocols for bat trapping supplies as adapted from the U.S.F.W.S. [White-nose Syndrome Decontamination Protocol](#) (1/25/2011) and the Wyoming Game and Fish Department “A Strategic Plan for White-nose Syndrome in Wyoming” (Abel & Grenier 2011). Basically, a clean holding bag will be used for each bat captured. All bags will be washed before another bat is placed within the bag. Alternatively, paper bags will be used and thrown away after one use. All equipment and gear will be wiped down with Lysol and clean water at the end of each survey. See Appendix 3 for full procedures.



Four nets (7 ft x 30 ft) stacked on top of each other suspended between 1" galvanized steel poles

Equipment List

Headlamp
Batteries
GPS unit
Compass
Kestrel thermometer/wind gauge
Flagging
Standard field kit: Clip board, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets, nail polish or spray paint.
Waders &/or duck boots
6 10-meter poles (easier to transport if in 3 sections) with appropriate eye bolts and buckles
4 pulleys to run the ropes attached to the nets through
Ropes - 30 foot (9.14 m) for top guy rope
60 ft (18.29 m) ropes between pulleys to attach nets to 4 50 ft (15.24 m) ropes to attach top of poles to ground (or trees)
Stakes
Stake driving device to make a 'pole hole'
Bat mist nets (4 shelves, 38 mm mesh, length= 7 ft high x 30 ft long)
Clips (should be attached to each mist net loop) to aid net movement
Bat holding bags (to be used only once) (e.g. small cotton, GSA 'mailing bags' 8x10 inches or paper bags)
Sunrise/sunset chart
Batting or gold gloves (leather)
Field guides
Night vision scope
Digital camera
Lysol Disinfectant Wipes
Lysol Disinfectant Cleaner to soak cloth holding bags

Staff and Training:

Trapping staff will be trained by an experienced bat biologist as to best placement of nets, safe handling techniques, correct measurements, and species identification. In addition, training should include 1) field guide use and identification, and 2) discussion of defining species characteristics. Since these are delicate animals which may carry various zoonotic diseases, no seasonal staff will be allowed to assist with bat trapping unless they have extensive previous experience and proper vaccinations.

Data Quality and Management:

Trapping

All trapping technicians will be trained as to the proper selection of monitoring habitats, net set up (proper placement and tension), determining when to stop surveying based upon wind or precipitation. Crew members could be rotated among crews (if possible) to reduce the potential for identification or 'escape of animal' bias.

As *Myotis* species, in particular, are difficult to distinguish, morphological measurements are critical for these captures. The measurements will allow the supervisor or data entry person to determine whether the measurements fall within the correct range for the species as which it was identified.

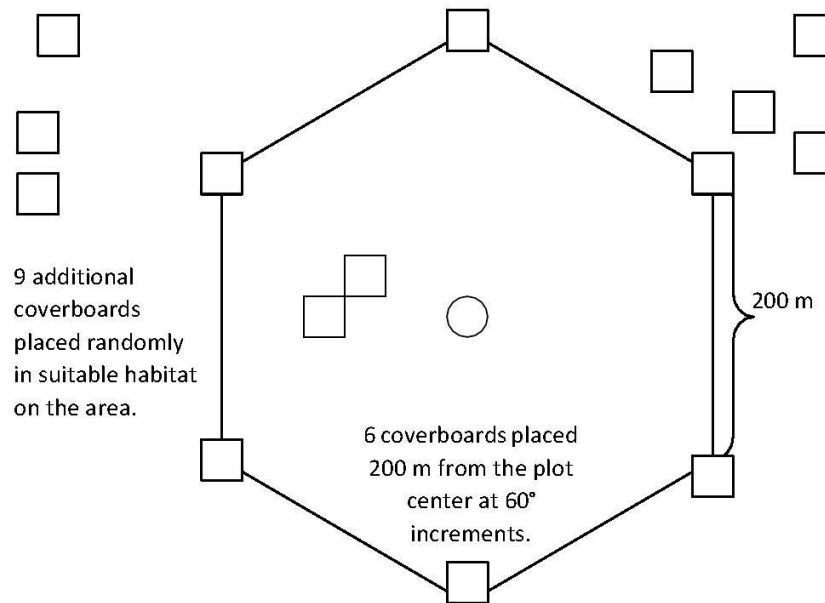
Chapter Ten:

Amphibian & Reptile Monitoring Protocol

Amphibian and Reptile Monitoring

Visual encounter surveys (VES) will be one of the 2 methods used in this protocol. VES is inexpensive, easy to implement, and efficient over diverse habitats (Manley et al. 2004). Additional benefits of VES include low site disturbance, low animal mortality, ease of implementation in terrestrial or aquatic environments, and other animals can be detected at the same time. The entire 26-acre (10 ha) hexagon will serve as the primary sampling unit with additional sites being located outside of the hexagon but within the 101 ha block surrounding the center point.

This area will allow for a large variety of habitat types to be searched.



The other method used in this protocol include aquatic traps. The diagram above shows the placement of cover boards which are checked at each VES and opportunistically while conducting other surveys. Aquatic traps should be deployed in wetlands for 3 days (and 2 nights) (Kolozsary and Swihart 1999) and checked daily.

Survey Methods

Visual Encounter Surveys

Each of the sampling properties will be subjected to a VES of 4 hours total per visit. This 4 hour timeframe may be broken into 2 hours with 2 technicians, 1 hour with 4 technicians, etc. Sites should be visited twice between mid-April and mid-June (quasi-spring), mid-June and mid-August (summer), and again mid-August and mid-October (quasi-fall), for a total of at least 6 visits per year. There should be at least a 10 day lag between all surveys. Since the goal is to find as many species of amphibians and reptiles as possible, searches should be focused on areas within and around the hexagon that appear to be suitable for these animals. For example, areas with rocks or logs that can be turned over should take precedence over areas that have no suitable cover. Each general site location (i.e. field 1 or Pond A) searched should be recorded on the datasheet regardless of if you found a reptile or amphibian or not. If no animals are found, record this a "None" species for the site within the property.

In addition, wetlands should be walked to a reasonable depth (the shoreline to about a 0.5 m depth) to search for egg masses, larvae, and amplexed frogs. In the wetland areas, surveys are conducted by walking the edge of the water body and zigzagging through wet meadow habitat. Two technicians can walk in opposite directions around a water body, ending the survey when they meet. If water is too deep to walk through, technicians stay on the edge of the water body. The entire wet meadow area should be searched. For streams, one technician surveys each side (a 500 m stretch, moving upstream from the starting point) simultaneously.

It is expected that technicians will spend approximately 15 minutes per 100 m of walking effort, stopping the stopwatch when extra time is needed for species ID or to move around obstacles (Manley et al. 2004). Searches are conducted using long-handled dipnets, and overturning logs and rocks. Be sure to replace all disturbed habitat as carefully as possible.

Coverboards

Coverboards are used as a source of cover by many species of herpetofauna (Corn 1994, Bennett et al. 2003). Typical size for boards is a 1-m² sheet that is at least 1 cm thick. Ours tend to be 1.2 m by 0.6 m. Six coverboards should be placed approximately 200 m from the center of the sample plot in the 6 directions of the point count stations (the outer BPC locations). This means the first coverboard should be placed due north, 200 m from the center of the plot. Every 60 degrees (so, 0°, 60°, 120°, 180°, 240°, and 300°), another board should be placed 200 m from the plot center. Incidentally, this coincides with the placement of the poles to mark the bird point count locations. Place the board 1-2 m from the pole to prevent people from accidentally standing on the coverboard.

Nine additional coverboards should be placed in suitable and logistically feasible habitat to attract snakes (ie. near parking lots, on the edge of fields, close to rock piles or brush piles). Be sure that the boards will get some sun throughout the season. Locations of these boards should be recorded with a GPS unit and marked in the mapbook and google map file to allow them to be found in the future. If possible, hang a piece of flagging nearby to assist in relocating the boards.

Coverboard placement may require the removal of litter on the surface as the coverboard should be flush with the soil (Manley et al. 2004). However, place some coverboards on litter/vegetation as this may provide additional cover snakes would find attractive. Compare capture numbers under the 'bare' and 'vegetated' coverboards to decide which would be most appropriate on each site.

Aquatic Traps

Minnow traps and other aquatic traps may be an effective way to find additional tadpoles (Shaffer et al. 1994). Aquatic traps should be deployed in water at least 0.5 m but not more than 2 m deep. Place an empty, capped plastic soda bottle in the trap to keep an area buoyant, allowing the animals to get oxygen. Alternatively, traps can be placed shallow enough that part of the trap is above water while sitting on the bottom of the wetland. Minnow traps do not need to be baited. For these traps, try to find areas of vegetation. Traps should be checked daily (no longer than 24 hours) and left in the water for 3 days (2 nights).

Traps should be set at least once per each of the three seasons. This can be done concurrently with one of the VESs for each season. Turtle-traps should be deployed concurrently with the minnow traps and checked daily. Turtle traps will be baited with sardines, although frozen fish, or fresh carp can be used as well. Turtle traps (hoop nets or fyke nets) should be placed in areas with numerous basking sites (log jams, sandbars, etc). For each trapping effort at each property use at least 3 hoop nets, 3 crab/crawfish nets, 6 minnow traps. If habitat is appropriate 1 fyke net can be used as well. See diagram below for more description of these traps.

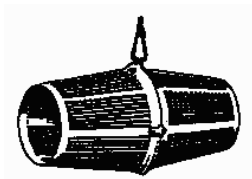


Diagram of a minnow trap. Taken from the British Columbia

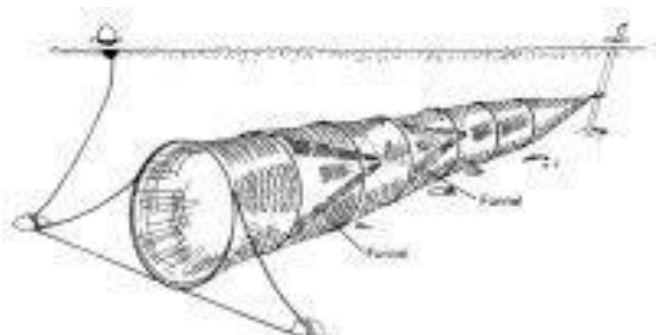


Diagram of a hoop net. Taken from the EPA's website (10/22/2011).

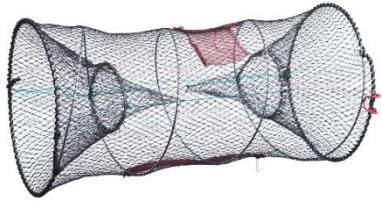


Diagram of a crawfish trap. Taken from BassPro Shop's website (4/8/2024).



Diagram of a crab trap. Taken from Cabela's website (10/22/2011).

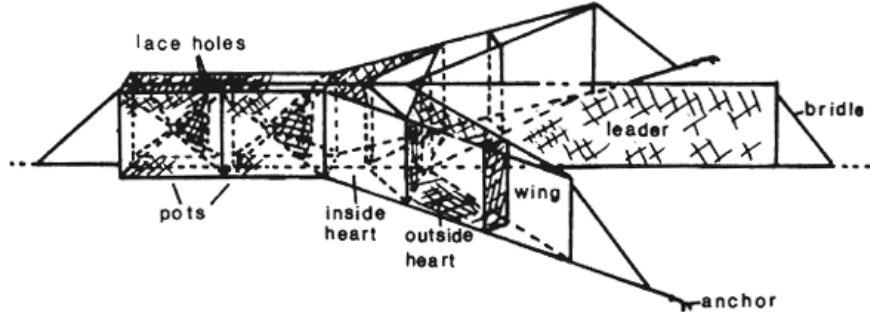


Diagram of a fyke net. Taken from the Food and Agriculture Organization of the United Nations website (10/22/2011)

When possible, traps should be placed in two wetlands/waterbodies each season. A singular waterbody will be randomly selected prior to the field season to be sampled every season and year. At some points, sampling this waterbody may be just taking note that the waterbody is dry or too low for traps to be placed. A short list of up to 3 other waterbodies will be provided to the field technicians for the use of aquatic trapping that season. These waterbodies are randomly selected before the field season and will be reselected every year a property is sampled. When properties are new or do not have waterbodies labeled, we encourage the field technicians to complete a VES prior to aquatic trapping to label as many waterbodies as possible. From there, the MSIM team will randomly select the permanent sampling wetland, as well as the other wetland options for that year.

Record all empty traps as such both on the data sheet and in the database. These null or non-captures are critical information too! Although we do capture fish in aquatic traps, these surveys are done primarily for amphibians and reptiles. Therefore, even if you do not capture any amphibians or reptile in a trapping effort, be sure to enter the survey into the database along with at least 1 trap with the species as "Null" so that we know a trapping session was done, even if no amphibians or reptiles were captured. You also need to create a second survey for each trapping effort to denote fish captures (and nulls) within the traps.

Recording Captures

To protect the amphibian, it is critical that the technician's hands be free of any chemicals or lotions. Insect repellent can be absorbed through the skin of the amphibian, resulting in the animal's death.

For each observation, record the species, detection type (visual, auditory, sign), age class (adult, sub-adult, juvenile), and substrate type (rock, log, bare ground, etc.). In addition, animals that are captured should be measured (snout to vent and total length), and assigned to sex and status. Animals found dead on the road or dead in a trap should be kept as voucher specimen. Place these animals in a plastic bag, label the bag with the day, location and species, and freeze until transport to either the DNR diversity program or another designated facility. Bags should be kept on ice when transporting. **For living captures, photo documentation should be made of each new species for a property, including common species.** Follow the guidelines in Appendix 4 (Herpetofauna Photo Voucher Guidelines) for amphibian and reptile photo vouchers.

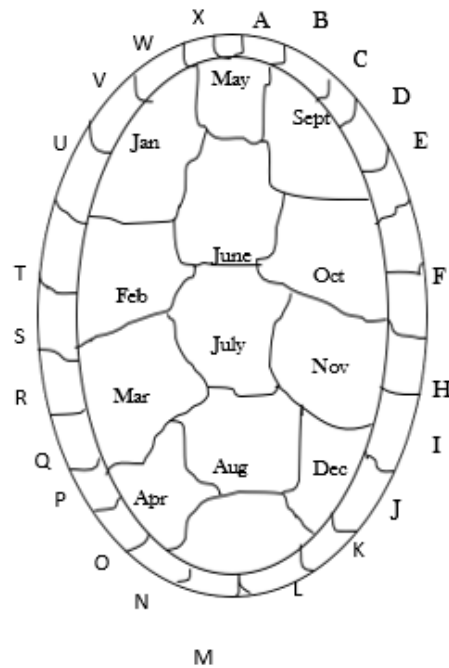
Turtles should be marked using a shell notch system (see Appendix 2). Notches can be created using a 3-sided file. By marking 1-3 marginal scutes, over 10,000 animals can be given a unique mark for Emydidae and Chelydridae turtles. Kinosternidae turtles are more difficult to mark as several of the marginal scutes are not broad enough to use a 3-edge file. In this situation, approximately 4,000 turtles can be marked individually. Unless more than 4,000 turtles of a given species are expected to be captured within a county, following the marking codes in Appendix 4 regardless of family is

advisable. Do not notch, file, or drill into scutes attaching the carapace and plastron together (typically E-H and Q-T in the diagram below).

Although not currently used for Iowa MSIM a paint pen or fingernail polish can be used to indicate in which month a turtle was captured. A paint pen could be used to write the year of capture in the costal or vertebral scute corresponding to the month of capture. Baby turtles can be batch marked by a small shell notch created with fingernail clippers. The marking system for Trionychidae (softshell turtles) is illustrated on the next page.

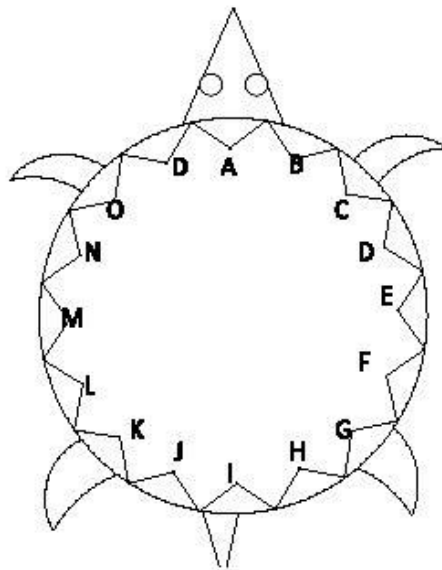
Shell marking diagram for turtles

Use a 3-edge file to mark the marginal scutes. Always count the number of marginal scutes on each side before marking the turtle. The top marginal scutes will always be 'A' and 'X'. The posterior two are always 'L' and 'M'. In addition to the individual mark using these scutes, a dot of paint (from a paint pen or fingernail polish) can be applied to the costal or vertebral scutes to indicate the month of capture.



Marking diagram for softshell turtles

Scute marks should be more slender than indicated in the diagram. Marks should be wide enough to be visible only (<5 mm at widest outer edge). Use a 3-edge file to mark the marginal scutes



Environmental Data Collection

It is expected that the data collected at the center of the hexagon and at each of the 6 hexagon-points will adequately describe the terrestrial component of the area. However, additional measurements are expected to be needed from wetlands searched as part of the VES and trapping design. In depth details concerning the aquatic data acquisition can be found in Chapter 20 (Aquatic Habitat Classification). That chapter includes information on collecting data on the habitats stratified into a wetland classification (i.e. river, stream, creek, impoundment, lake, etc.). Any additional wetlands (i.e. creeks, streams, ponds, etc.) which were surveyed for amphibians and reptiles would also need to have aquatic habitat characteristics measured. These measurements should be collected as outlined in Chapter 20. In addition to these measurements are data that can be determined from GIS coverages in the lab prior to field work (see Chapter 3 GPS & GIS Coverage). Measurements include amount of roads and other impacted soils adjacent to the water body, locations of, and numbers of water bodies. These will still need to be groundtruthed in the field.

Equipment List

Kestrel temperature and wind gauge
Pair leather gloves (for large snake captures)
Hand spade or rake
Field guides (leave in truck)
Hand lens
Phone (stop watch and camera)
Pair hip waders
15 Coverboards
Minnow traps
Hoop traps
Crab traps
Fyke nets
Animal marking equipment: Nail polish, 3-edged file, cuticle scissors, &/or VIE, CWT, PIT.
Standard field kit: Clip board, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, plastic zip-lock baggies, & data sheets.

Staff & Training

Training is recommended and should include 1) field guide use and identification, 2) discussions of defining species characteristics, 3) field practice with an experienced observer, and proficiency testing. Training should also include instruction on how to mark and hold turtles safely. Also need training on habitat data collection.

Data Quality & Management

VES can be difficult to rate for quality:

- Examination of data will not reveal missed detections or misidentifications.
 - Misidentifications can be checked by the use of digital cameras, or by the field supervisor working periodically with each technician.
- Manley et al. (2004) suggests rotating crew members such that each site is visited by more than one crew to reduce the effect of observer bias.
- All photographs should be reviewed by at least 2 additional people to verify species identifications. This is done by the IWAP Amphibian & Reptile Subcommittee each January to confirm new county records
- All records determined to be mis-identified are to be changed to unknown in the data base with a note in the comment of the record as to what the original identification was.

At the end of each trapping day, field crew pairs should review data sheets to ensure all information present. At the end of the week, the field crew leader should review the data sheets for ID, escape and mortality rates, and legibility. Also each day, digital cameras should be downloaded and the photos labeled as to site, date, and species. Be sure to follow the photo labeling guidelines: speciesname_propertyname_Survey_date_Obs.jpg

Data Analysis

The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of points occupied, or other occupancy modeling techniques (see Chapter 5, Data Analysis). The species list can be used to calculate basic diversity indices. Depending on the numbers of animals recaptured, the data may also be able to be used to estimate population size, although this is unlikely. See Chapter 5 for additional information.

Safety Considerations

Venomous Snakes

Never reach underneath a coverboard, rock, or other substrate covering without first flipping it over to see what is underneath. These animals are rare enough that they do not need to be marked, and therefore would not need to be handled. If possible, photograph the animal such that the coloration of the dorsal (back) surface can be compared to subsequent captures with photographic-pattern-recognition software.

There is no reason for an Iowa MSIM technician to handle a venomous snake.

If, however, there is reason to mark these animals as part of a different study (probably marking with passive integrated transponders (PIT-tags)), the safest way to handle a venomous snake would be to use a snake stick to place it into a plastic container (such as a Rubbermaid container at least 43 cm deep). Then, using a snake tube, entice the snake to climb into the tube. Once it is in far enough that it cannot scrunch backwards to escape and yet not in so far as to come out the other end, grasp the belly of the snake at the end of the tube. This immobilizes the snake so it can be properly marked and measured. However, do not attempt this unless you have been trained. As stated above, most likely venomous snakes do not need to be handled, but as part of this program, do photograph it (from a safe distance without disturbing it) if possible.

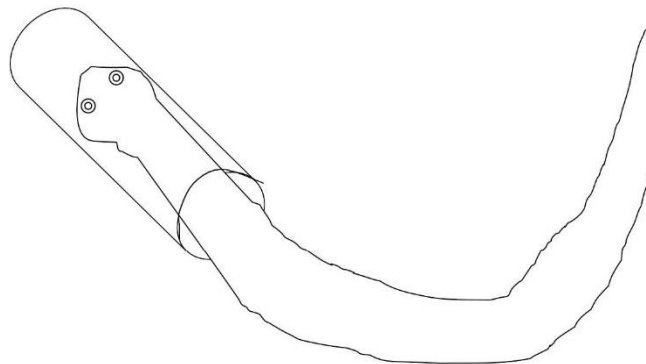


Diagram of snake in a snake tube

Hygiene

Several amphibian species, particularly toads, are capable of producing an irritant from their skin. Do not rub your eyes or face or eat after handling an amphibian without first washing your hands. Should you get the amphibian secretion in your eye (it will burn), wash with water immediately. If this does not help, seek medical treatment.

Care should be taken in order to lessen the probability of spreading an infectious agent, such as a fungus or virus, between wetlands. One way to reduce the chance of spreading an infectious agent between wetlands is to allow waders and equipment to dry for 3-4 days between sites. This may be impractical given the short time frame available for aquatic surveying in Iowa. As an alternative, it may be best to rinse all equipment with a solution of hot water and bleach. Spray bottles and bleach are provided for this purpose.

Target Species

The following list of target species represents the species of greatest conservation need as chosen by the Steering committee for the 2015 Revision of the Iowa Wildlife Action Plan (Reeder and Clymer. 2015). Distribution maps for these species can be found in The Salamanders and Frogs of Iowa (Christiansen and Bailey 1991), The Snakes of Iowa

(Christiansen and Bailey 1990), The Lizards and Turtles of Iowa (Christiansen and Bailey 1997), and also in Iowa GAP (Kane et al. 2003). Appendix 1 contains a list of additional, more common, herpetofauna species which may be encountered during the monitoring efforts. An astrisks (*) indicates species designated as Data Deficient.

Target amphibian species:

Common Name	Scientific Name	Habitat
Mudpuppy*	<i>Necturus maculosus</i>	Clean rivers, streams, lakes, reservoirs
Eastern newt	<i>Notophthalmus viridescens</i>	Vegetated woodland ponds, roadside flooded ditches, & adjacent habitat
Tiger salamander	<i>Ambystoma tigrinum</i>	Generalist
Smallmouth salamander	<i>Ambystoma texanum</i>	Woodland pools, open woods
Blue-spotted salamander	<i>Ambystoma laterale</i>	Woodland pools, open woods
Great plains toad	<i>Anaxyrus cognatus</i>	Prairie, nonnative grassland
Fowler's toad	<i>Anaxyrus fowleri</i>	Grasslands & woodlands
Woodhouse's toad	<i>Anaxyrus woodhousii</i>	Woodlands & savanna
Blanchard's Cricket frog	<i>Acris crepitans</i>	Shallow wetlands & streams
Cope's gray treefrog	<i>Hyla chrysocelis</i>	Wetlands, savanna grasslands, woodland edges
Eastern gray treefrog	<i>Hyla versicolor</i>	Forests, woodlands, riparian areas
Crawfish frog	<i>Lithobates areolata</i>	Prairie marshes, ponds, river floodplains
Pickrel frog	<i>Lithobates palustris</i>	Cold water streams, rivers, and impoundments
Northern leopard frog	<i>Lithobates pipiens</i>	All aquatic habitats
Southern leopard frog	<i>Lithobates sphenoccephalus</i>	On-stream impoundments, wetlands, ponds, and backwaters
Plains spadefoot toad	<i>Spea bombifrons</i>	Grasslands in the Loess Hills
Snapping turtle	<i>Chelydra serpentina</i>	All aquatic habitats
Blanding's turtle	<i>Emydoidea blandingii</i>	Shallow, well vegetated wetlands
Wood turtle	<i>Clemmys insculpta</i>	Floodplain forest, rivers
Northern map turtle	<i>Graptemys geographica</i>	Rivers & streams
Southern map turtle	<i>Graptemys ouachitensis</i>	Mississippi River and Associated oxbows
False map turtle	<i>Graptemys pseudogeographica</i>	Slow portions of large rivers
Ornate box turtle	<i>Terrepene ornata</i>	Sand prairies, savanna
Yellow mud turtle	<i>Kinosternon flavescens</i>	Shallow, ephemeral pools, adjacent areas with nearly pure sand soils
Eastern musk turtle	<i>Sternotherus odoratus</i>	Backwaters and spring fed ponds adjacent to sandy uplands
Smooth softshell turtle	<i>Apalone mutica</i>	Large rivers
Spiny softshell turtle	<i>Apalone spinifera</i>	Rivers, streams, large lakes
Slender glass lizard	<i>Ophisaurus attenuatus</i>	Prairie, pasture, forest edge, savanna
Common five-lined skink	<i>Plestiodon fasciatus</i>	Deciduous forests along the bluffs of the Mississippi River and large eastern Iowa rivers
Great plains skink	<i>Plestiodon obsoletus</i>	Rocky prairie, forest edge
Northern prairie skink	<i>Plestiodon septentrionalis</i>	Sandy prairie-forest edge, wetland edge
Six-lined racerunner	<i>Aspidocelis sexlineatus</i>	Sand prairies, savanna
Western worm snake	<i>Carphophis amoenus</i>	Rocky woodland
Prairie ringneck Snake	<i>Diadophis punctatus</i>	Woodlands, savanna & adjacent grasslands

Common Name	Scientific Name	Habitat
Western (plains) hognose snake	<i>Heterodon nasicus</i>	Sand prairie
Eastern hognose snake	<i>Heterodon platirhinos</i>	Woodland, savanna, & grassland
Prairie kingsnake	<i>Lampropeltis calligaster</i>	Woodland edge, open woodland, grassland, savanna
Speckled kingsnake*	<i>Lampropeltis getulus</i>	Prairie, woodland edge, savanna
Copperbelly water snake	<i>Nerodia erythrogaster neglecta</i>	Backwater sloughs and forested wetlands
Diamondback water snake	<i>Nerodia rhombifera</i>	Quiet pools and backwater sloughs
Common water snake	<i>Nerodia sipedon</i>	Lakes, ponds, marshes, streams, backwaters

Target reptile species:

Common Name	Scientific Name	Habitat
Smooth green snake	<i>Opheodrys vernalis</i>	Old field, savanna, wet prairie, marsh
Western rat snake	<i>Elaphe obsoleta</i>	Heavily wooded bluff lands along rivers
Western fox snake	<i>Pantherophis ramspotti</i>	Wooded rivers, streams, savanna & grasslands near lakes and marshes
Gopher (Bull) snake	<i>Pituophis catenifer</i>	Sand and bluff prairies, savanna, pasture
Graham's crayfish snake	<i>Regina grahamii</i>	Ponds, sloughs, marshes, floodplains, creeks
Northern redbelly snake	<i>Storeria occipitomaculata</i>	Savanna, woodland and adjacent grasslands near water
Western ribbon snake	<i>Thamnophis proximus</i>	Herbaceous wetland, ponds, streams, rivers
Plains garter snake	<i>Thamnophis radix</i>	Generalist
Northern lined snake	<i>Tropidoclonion lineatum</i>	Grassland, pasture, woodland edge
Smooth earth snake	<i>Virginia valeriae</i>	Rocky woodland
Copperhead	<i>Agkistrodon contortrix</i>	Forested, rocky hillsides
Timber rattlesnake	<i>Crotalus horridus</i>	Forested areas near rock outcrops, woodland, hill prairie
Prairie rattlesnake	<i>Crotalus viridis</i>	Prairie
Eastern massasauga rattlesnakeA	<i>Sistrurus catenatus catenatus</i>	Early successional wetland, upland grassland
Western massasauga rattlesnake	<i>Siturus turgemins</i>	Prairie wetland close to rivers and adjacent upland prairie

*Indicates species listed as Federal Threatened under the Endangered Species Act.

Additional Methods for Special Locations

The following are additional techniques which may be implemented at certain sites **in addition** to the core methods described above. These could be used in areas where there are known populations of species of concern or when supplemental funding has been acquired for a given area. However, the basic core protocol must still be followed to allow for comparison of all sites, both across the state of Iowa and also for a regional comparison, provided that other states or areas are following the same protocol.

VES Augmentation

1. Nocturnal surveys - Conduct at least one additional search at night to detect those species most active at night.
2. Extend the survey time - In habitats with many species of amphibians and reptiles, it may be necessary to increase the amount of time each crew spends looking for animals, but the data will need to be recorded such that the first 4 hours (2 for each technician) can be extracted for comparisons to other areas.

Pitfall Trapping

Pitfall traps with (or without) drift fences are time consuming to install. They also only catch species that are not able to

jump or crawl out, mostly limiting the use to salamanders, toads, small snakes, and some lizards. They can result in high mortality for small mammals, or herpetofauna if not checked daily. Should the decision be made to include pitfall traps into the monitoring regime, several references (Corn 1994, Karraker 2001, and Manley et al. 2004 draft) should be incorporated into the design.

Nocturnal Auditory Amphibian Counts

In addition to the VES, each sample plot could be visited one night during each of 3 'seasons'. This would follow the methodology used in the Iowa Frog and Toad Survey protocol, except the locations would be in the permanent sampling plots as opposed to being close to roads. The technicians would visit areas with standing water within the sampling plot, at night, and listen at each wetland for 5 minutes. All calls heard would be recorded for each species and given an 'index ranking' of 1, 2, or 3 depending upon the number of individuals heard. A ranking of 1 is equal to being able to count the number of individuals; there should be space between the calls. A ranking of 2 is equal to being able to distinguish individuals but there should be overlap between the calls. A ranking of 3 would mean that it is a full chorus of calling, with constant, continuously overlapping sounds. Ideally, these visits would be conducted at least twice, preferably 3 times during the 'spring' (mid-April through mid-June) and the 'summer' (mid-June through mid-July) seasons. In actuality, each site should be visited at least once during April, once between May 7 and June 4, and once between June 13 and July 10. If conditions and resources are available for additional visits during these timeframes, those visits should be made.

Additional Marking Techniques

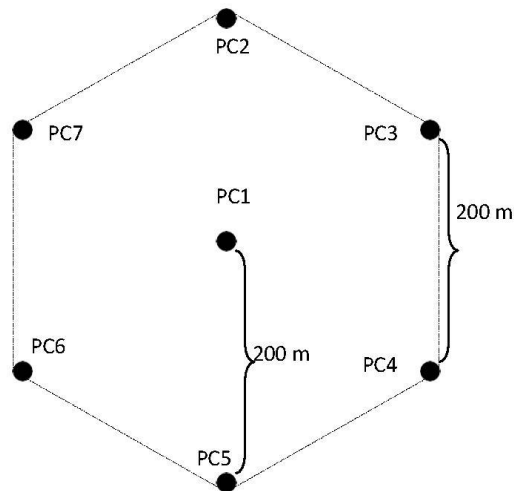
Amphibians could be marked with either toe-clipping (cheaper) or Visible Implanted Elastomer depending on how often a site is expected to be visited. In most situations it probably is not feasible to mark amphibians, due to the typically short amount of time for which they would be capture-able. Salamanders, however, could be marked if the site is to be visited each year

Chapter Eleven:

Breeding and Migratory Bird Monitoring Protocol

Bird Monitoring

The primary method used to document birds in this program is point count surveys. This method will be implemented in such a way as to be able to be compared to other standard 10-minute point count data. In addition to the 4-point locations utilized by the U.S.F.S. (one in the hexagon center and 3 of the hexagonal edge points, Manley et al. 2005), the Iowa MSIM program will use all of the hexagon points as well as the center point for a total of 7 stations per site.



Survey Methods

Point Counts

From the interior center point (point count location 1), the azimuths for the remaining 6 point count locations are 0°, 60°, 120°, 180°, 240°, and 300°, respectively. The point count stations are 200 m apart. If the station should fall on a dangerous (e.g. cliff) or noisy (e.g. road) place, then the station should be moved to the closest available spot with care being taken to keep the station spacing at a minimum of 200 m. Stations should never be located less than 200 m from another station. The new location for stations should be reported to Database Manager.

The timing of observations at the point counts will include 3 seasons. The first (spring: April-May) and last (fall: September-October) will focus on migratory birds. The middle season (summer: June-July) will focus primarily on breeding birds. However, ALL birds seen or heard during any field visit should be recorded. Since migratory birds are not as vocal or showy as breeding birds, the surveys conducted during these 2 seasons may not necessarily be restricted to the morning hours. The IWAP Bird Subcommittee (at least the Chair and a small subset of members) will decide when and if bird observers are allowed to vary the timing of the surveys. Unless granted permission by that subcommittee, all bird surveyors should follow the guidelines established for the summer season (starting near daybreak, one property per day, etc.) for every survey.

All of the point count stations in a single hexagon will be visited on the same day. Once at the station, the technician will record 10 minutes of information, divided into 3 time frames: the first 3 minutes, the middle 2 minutes, and the last 5 minutes. However, upon first arriving at the point count station, the technician should wait 2 minutes, standing quietly before beginning the timed data collection. During the summer, data collection should begin within 15 minutes of sunrise (before or after) and be concluded for the day by 4.5 hours after sunrise. Depending on travel time both within the hexagon between stations and between property locations, it may or may not be possible for 1 person to cover 2 properties in one day. The order of the stations visited (e.g. 1, 2, 3, 4, 5, 6, & 7 vs. 2, 6, 7, 1, 3, 5, & 4) is left to the discretion of the observer but should be randomly mixed or mixed by choosing a different starting point each visit.

Within the spring and summer seasons, 3 visits will be made to each site, with at least 4 days in between visits. During the fall season, however, bird species composition may change quickly; therefore 4 visits per site would be preferred,

although due to time constraints, this is unlikely to happen. If granted permission by the IWAP Bird Subcommittee, it may be feasible to cover two properties each day as it is not as critical that data be collected only during the morning in the fall (i.e. it does not get as hot so birds are still active). Inclement weather, following the [Breeding Bird Survey rules](#), and including fog, steady drizzle, prolonged rain, and wind >20 km/h (12 mph), could result in stopping the survey.

In addition to recording the species seen or heard, additional data will be collected for every observation, including the distance bin of the individual and the type of observation (visual or auditory). In using DISTANCE SAMPLING for the point count locations, it is critical to correctly be able to estimate the distance of the bird from the observer. On the data sheet, the distance (in meters) is divided into categories to aid in this estimation. The technician must also be careful to record birds where they are first detected and to avoid double counting the same individual. By recording the distance estimates, one may calculate the species density.

If birds are seen in flocks or with broods, be sure to indicate those on the datasheet so that a bird with a brood of 8 chicks is recorded as such instead of being recorded as 9 birds assumed to be adults. Likewise, an observer could see a flock (grouped together) of 8 birds, or they could see 8 individual birds of the same species spread out on territories within the sampling area. These distinctions are critically important in analyzing the data. Individual birds should be recorded as such, and the others should be grouped together.

Other data to be collected at every sampling hexagonal plot include the date, cloud cover, wind speed, and start & end times and temperature. At each BPC location, record the start time for that location as well. Species other than birds which are seen or heard during the day should also be recorded (including calling amphibians or vocal mammals, for example). Birds that fly overhead without landing in the sampling plot should be recorded as 'flyovers'. In addition, new bird species seen or heard as the technician moves through the sampling plot should be recorded as incidental sightings. These individuals will be noted on the species list for the property, but no distance measurement will be recorded outside the point stations and these individuals will not be used in the density estimates.

Visual Encounter Surveys

During the summer season (June and July) an additional 30 minutes should be spent on each property after the BPC has been completed on 2 of the 3 visits, for a total of 1 hour of extra search time. This search time should be spent in the 'best quality' habitat (left to the discretion of the technician, with input from the project leader). This survey will not be split between field technicians. All birds (and other species) seen or heard should be recorded along with information as to what area the animal was in. The areas should be delineated on the site information sheet/aerial photo maps compiled under the protocol in Chapter 3 (Landscape Characteristics) to ensure that the same name is used for a given area between multiple people.

Environmental Data Collection

Environmental variables such as air temperature, wind speed, and other weather conditions should be recorded at the time of the survey on the bird monitoring data sheets.

Equipment List

- Point count surveys: Binoculars
- Small digital recorder (if needed, to record unrecognized bird calls)
- Stopwatch/watch/phone
- Range finder (if observer needs assistance in determining distance)
- Standard field backpack with clipboard, datasheets, pencils, notebook, and field guides, weather Kestrel, GPS unit

Staff & Training

Point count survey technicians should be hired based upon their ability to already be able to identify birds by call and sight (at least most birds). They can gain experience on the job but should have at least limited prior experience. People hired with a greater amount of experience could be given the extra responsibility of helping to train and test the more inexperienced technicians.

Although technicians should be hired based upon previous experience, there should also be training at the beginning of

the season, including field trials and museum visits. Technicians will be tested and leaders can adjust training to the needed level. Technicians need to learn when to halt surveys due to bad weather. Training should include judging distance as well. This can be done by flagging different distances and have them practice recording the distance.

Data Quality & Management

To aid in the management of the data quality, care must be taken to ensure technician proficiency in bird identification. This can be addressed by testing technicians before the beginning of the season and also during the season. Survey times should also be limited to a given timeframe (the 4.5 hours after sunrise for point counts). Technicians should know when to halt data collection during inclement weather, to move away from noise, and to wear muted colors. Every Friday, each birder is required to email a weekly species list to the Chair of the IWAP Bird Subcommittee and the DNR Avian Ecologist. This allows for a quality control during the field season.

Things that the project leader should look for when ‘testing’ technicians include:

1. Are technicians quiet and attentive?
2. Are they turning their heads and bodies to listen in all directions?
3. Are they looking at the sky?
4. Scanning up and down vegetation?
5. Looking at the ground?
6. Are they using binoculars?
7. Are they recording directions correctly?
8. Are they double counting birds?
9. Are they correctly estimating distance?
10. Are the data legible?

Data sheets should be examined daily by the recording technician to ensure all fields are filled in. Data sheets should be checked at least weekly by the data manager to prevent time lags in case more information is needed from the recording technician.

Data Analysis

Using program R and several different packages, probability of detection estimates and proportion of points occupied for all of the data collected during these surveys can be calculated. Program MARK, as well as program R, has analysis capabilities using either the “Occupancy Estimation” or the “Robust Design Occupancy” data type selection depending on how many seasons are being analyzed. Since the point count station data includes distance estimates between the birds and the observer, additional analyses can be done, including density estimation. Currently most of the analyses are completed under similar frameworks in program R. See Chapter 5 (Data Analysis) for additional information on these techniques.

Safety Considerations

The point count technicians will be working alone and therefore should carry a reliable cell phone, GPS unit, maps, and first aid kit. The crew should maintain a sign in/sign out method to ensure everyone returned from the field as well as to know exactly where each crew member is assigned to work every day.

Target Species

The following list of target species represents the species of greatest conservation need as chosen by the Steering committee for the 2015 Revision of the Iowa Wildlife Action Plan (Reeder and Clymer, 2015). Birds have been divided into 2 groups: breeding birds and migratory birds. Distribution maps for these species can be found in Birds in Iowa (Kent and Dinsmore 1996) and additional maps for some species can be found in Iowa GAP (Kane et al. 2003). Appendix 1 contains a list of additional, more common, bird species (again, these have been separated into breeding and migratory bird species) which may also be encountered during the monitoring efforts. An asterisks (*) indicates species designated as Data Deficient. Species listed under the Federal Endangered Species Act are designated by ‘A’.

Target breeding bird species:

Common Name	Scientific Name	Habitat
Trumpeter swan	<i>Cygnus buccinator</i>	Wetland
American wigeon	<i>Anas americana</i>	Wetland
Blue-winged teal	<i>Anas discors</i>	Wetland
Northern pintail	<i>Anas acuta</i>	Wetland, grassland
Canvasback	<i>Aythya valisineria</i>	Wetland
Redhead	<i>Aythya americana</i>	Wetland
Ring-necked duck	<i>Aythya collaris</i>	Wetland
Lesser scaup	<i>Aythya affinis</i>	Wetland
Northern bobwhite	<i>Colinus virginianus</i>	Grassland, shrubland
Ruffed grouse	<i>Bonasa umbellus</i>	Dense forest, open woodland
Sharp-tailed grouse	<i>Tympanuchus phasianellus</i>	Grassland, shrubland
Greater prairie-chicken	<i>Tympanuchus cupido</i>	Grassland
Red-necked grebe	<i>Podiceps grisegena</i>	Wetland, hemi-marsh
Eared grebe	<i>Podiceps nigricollis</i>	Wetland, hemi-marsh
American white pelican	<i>Pelecanus erythrorhynchos</i>	Wetland
American bittern	<i>Botaurus lentiginosus</i>	Wetland
Black-crowned night heron	<i>Nycticorax nycticorax</i>	Wetland, wet shrubland
White-faced ibis	<i>Plegadis chihi</i>	Marsh
Bald eagle	<i>Haliaeetus leucocephalus</i>	Riparian forest, deciduous forest
Northern harrier	<i>Circus cyaneus</i>	Grassland, marsh
Red-shouldered hawk	<i>Buteo lineatus</i>	Riparian forest
Broad-winged hawk	<i>Buteo platypterus</i>	Deciduous forest
Swainson's hawk	<i>Buteo swainsoni</i>	Savanna, open woodland
King rail	<i>Rallus elegans</i>	Wetland
Common gallinule	<i>Gallinula chloropus</i>	Wetland
^A Piping plover	<i>Charadrius melodus</i>	Wetland
Upland sandpiper	<i>Bartramia longicauda</i>	Grassland
Wilson's snipe	<i>Gallinago delicata</i>	Hemi-marsh
American woodcock	<i>Scolopax minor</i>	Deciduous forest, open woodland, riparian forest
Wilson's phalarope	<i>Phalaropus tricolor</i>	Wetland, grassland
Franklin's gull	<i>Larus pipixcan</i>	Wetland
Least tern	<i>Sterna antillarum</i>	Wetland
Black tern	<i>Chlidonias niger</i>	Wetland
Forster's tern	<i>Sterna forsteri</i>	Wetland
Yellow-billed cuckoo	<i>Coccyzus americanus</i>	Deciduous forest, shrubland, open woodland
Black-billed cuckoo	<i>Coccyzus erythrophthalmus</i>	Riparian and deciduous forests, open woodland, shrubland
Barn owl	<i>Tyto alba</i>	Savanna
Eastern screech owl*	<i>Otus asio</i>	Riparian forest/woodland, savanna
Burrowing owl	<i>Speotyto cunicularia</i>	Grassland
Long-eared owl*	<i>Asio otus</i>	Open woodland, savanna, deciduous forest
Short-eared owl	<i>Asio flammeus</i>	Grassland

Common Name	Scientific Name	Habitat
Common nighthawk	<i>Chordeiles minor</i>	Grassland, savanna
Chuck-will's-widow	<i>Caprimulgus carolinensis</i>	Open woodland, savanna
Eastern whip-poor-will	<i>Caprimulgus vociferus</i>	Deciduous forest, open woodland
Chimney swift	<i>Chaetura pelagica</i>	Older growth forests, woodlands, savanna, towns
Belted kingfisher	<i>Ceryle alcyon</i>	Wetlands, riparian, with cutbanks
Red-headed woodpecker	<i>Melanerpes erythrocephalus</i>	Savanna, open woodland, deciduous forest
Northern flicker	<i>Colaptes auratus</i>	Savanna, open woodland
American kestrel	<i>Falco sparverius</i>	Savanna, open woodland
Peregrine falcon	<i>Falco peregrinus</i>	Riparian forest, deciduous forest
Eastern wood-pewee	<i>Contopus virens</i>	Forest/woodland, savanna
Acadian flycatcher	<i>Empidonax virens</i>	Deciduous forest, riparian forest
Say's phoebe	<i>Sayornis saya</i>	Grassland
Eastern kingbird	<i>Tyrannus tyrannus</i>	Grassland-shrub, edges
Loggerhead shrike	<i>Lanius ludovicianus</i>	Savanna, shrubland
Bell's vireo	<i>Vireo bellii</i>	Shrubland, savanna
Horned lark	<i>Eremophila alpestris</i>	Sparse grassland, agricultural land
Purple martin	<i>Progne subis</i>	Riparian forests, towns, and wetlands
Bank swallow	<i>Riparia riparia</i>	Rivers and streams with cutbanks
Sedge wren	<i>Cistothorus platensis</i>	Grassland, wetland
Bewick's wren	<i>Thryomanes bewickii</i>	Open woodland, shrubland
Veery	<i>Catharus fuscescens</i>	Riparian and deciduous forest
Wood thrush	<i>Hylocichla mustelina</i>	Deciduous and riparian forest
Brown thrasher	<i>Toxostoma rufum</i>	Shrubby grassland
Worm-eating warbler	<i>Helmitheros vermivorus</i>	Deciduous forest
Golden winged warbler	<i>Vermivora chrysoptera</i>	Shrubby open woodland
Prothonotary warbler	<i>Prothonotaria citrea</i>	Riparian forest
Kentucky warbler	<i>Oporornis formosus</i>	Deciduous and riparian forest
Common yellowthroat	<i>Geothlypis trichas</i>	Shrubland and grassland
Cerulean warbler	<i>Dendroica cerulea</i>	Deciduous forest
Field sparrow	<i>Spizella pusilla</i>	Shrubland, grassland
Grasshopper sparrow	<i>Ammodramus savannarum</i>	Grassland
Henslow's sparrow	<i>Ammodramus henslowii</i>	Grassland
Dickcissel	<i>Spiza americana</i>	Grassland
Bobolink	<i>Dolichonyx oryzivorus</i>	Grassland
Eastern meadowlark	<i>Sturnella magna</i>	Grassland, savanna
Western meadowlark	<i>Sturnella neglecta</i>	Grassland
Baltimore oriole	<i>Icterus glabula</i>	Open woodland, savanna

Target migratory bird species:

Common Name	Scientific Name	Habitat
Greater scaup	<i>Aythya marila</i>	Lakes, rivers, wetlands
Common loon	<i>Gavia immer</i>	Clear, large, open water lakes
Little blue heron	<i>Egretta caerulea</i>	Marsh

Common Name	Scientific Name	Habitat
Yellow rail	<i>Coturnicops noveboracensis</i>	Wetland, grassland
*Black rail	<i>Laterallus jamaicensis</i>	Marsh
Whooping crane	<i>Grus americana</i>	Wetland, grassland
Black-bellied plover	<i>Pluvialis squatarola</i>	Wetland
American golden-plover	<i>Pluvialis dominica</i>	Wetland
Lesser yellowlegs	<i>Tringa flavipes</i>	Wetland
Whimbrel	<i>Numenius phaeopus</i>	Wetland
Long-billed curlew	<i>Numenius americanus</i>	Grassland-wetland
Hudsonian godwit	<i>Limosa haemastica</i>	Wetland
Marbled godwit	<i>Limosa fedoa</i>	Wetland
Ruddy turnstone	<i>Arenaria interpres</i>	Grassland-wetland
^A Red knot	<i>Calidris canutus</i>	Wetland
Sanderling	<i>Calidris alba</i>	Wetland
Semipalmated sandpiper	<i>Calidris pusilla</i>	Wetland
White-rumped sandpiper	<i>Calidris fuscicollis</i>	Wetland
Pectoral sandpiper	<i>Calidris melanotos</i>	Wetland and grassland
Stilt sandpiper	<i>Micropalama himantopus</i>	Wetland
Buff-breasted sandpiper	<i>Tryngites subruficollis</i>	Wetland, short grassland
Short-billed dowitcher	<i>Limnodromus griseus</i>	Wetland
Long-billed dowitcher	<i>Limnodromus scolopaceus</i>	Marsh
Caspian tern	<i>Sterna caspia</i>	Wetland
Olive-sided flycatcher	<i>Contopus cooperi</i>	Coniferous forests, openings and edges
Sprague's pipit	<i>Anthus spragueii</i>	Grassland
Bohemian waxwing	<i>Bombycilla garrulus</i>	Coniferous or mixed forest
Smith's longspur	<i>Calcarius pictus</i>	Grassland
Bay-breasted warbler	<i>Dendroica castanea</i>	Forest/woodland
Canada warbler	<i>Wilsonia canadensis</i>	Deciduous forest
American tree sparrow*	<i>Spizella arborea</i>	Open woodland
Le Conte's sparrow	<i>Ammodramus leconteii</i>	Grassland
Harris's sparrow	<i>Zonotrichia querula</i>	Pastures, hedgerows
White-winged crossbill	<i>Loxia leucoptera</i>	Coniferous forest

Additional Methods for Special Locations

The following are additional techniques which could be implemented at certain sites *in addition* to the core methods described above. These could be used in areas where there are known populations of species of concern or when supplemental funding has been acquired for a given area. However, the basic core protocol must still be followed to allow for comparison of all sites, both across the state of Iowa and also for a regional comparison, provided that other states or areas are following the same protocol.

Nocturnal Broadcast Surveys

Since the target of the nocturnal broadcast calling surveys (i.e. owls) have home ranges much larger than the area of the hexagonal sampling site (e.g. burrowing owl: 64-139 ha, in Gervais et al. 2003), a larger area encompassing the sampling plot will be utilized for these surveys. Up to a 300-ha block around the center point should be used. Within this block, sampling points should be chosen ahead of time with the aid of aerial photos. During daylight hours, these sites should be located and flagged (along with necessary trails) with reflective tape. Each block should contain 3 to 10 broadcast

stations. Some of the stations can be along roads, but other should be away from roads. Hilltops may work best for this technique and care should be taken to broadcast across drainage areas as opposed to along drainages. For safety reasons, at least 2 technicians should always be together to complete this survey. Sites should be surveyed at least twice per season at times that do not interfere with breeding, as this technique could result in nest abandonment if done too often. Surveys should not be conducted more often than twice in one month. An additional problem may be that birds become habituated to the survey (and fail to respond) if done too often. It is advised to time the 'season' of this survey to that which would result in the best response from the 5 species of owls of greatest conservation need.

Nocturnal broadcast surveys begin 30 minutes after sunset and end at midnight (although some species may be responsive 4 hours prior to sunrise, so need to determine best timing for owls of interest here). Surveys are not done during bad weather conditions. Published literature suggests best results occur on moonlit (bright) nights. The recording should contain calls of the target species for that area beginning with the smallest species and ending with the largest species. Calls will be played on a portable player and if needed, amplified with additional speakers such that calls are 100-110 dB at 1 m in front of the technician holding the speaker.

When the technicians arrive at the survey point, 2 minutes of 'silence' are first observed where all calls are written down. Then, each call is broadcast 3 times with 30 seconds of silence between calls, with an additional 30 seconds between species calls (this can be set-up ahead of time with the recording). Observers should pause the recording when necessary for species ID. One observer moves (quietly) around (up to 50 m) in order to increase detection probability. Both observers listen and watch for birds. After all calls have been played, observers watch and listen for 5 more minutes, using a 1,000,000 candle-watt spotlight to search for additional birds. In addition to the owls, the technique may work for American woodcock, Whip-poor-wills, Henslow's sparrows, rails and other marsh birds. Species to be included on the tape are expected to vary by county.

Data collected should include: survey route/site description, call station number, UTM coordinates, and directions to station (these can be recorded when stations are identified and flagged during daylight). Also collected are data concerning: site identification number, call station number, time, temperature, windspeed, precipitation, cloud cover, moon phase and visibility, bird identification, sex (if possible), time of detection, response of detection (in regards to species playing on recording), and bird location.

The data collected from the nocturnal calling surveys should be evaluated immediately to determine if increased stations or numbers of surveys are needed. Two potential problems with increasing the number of surveys is that the birds may (1) habituate to the calls or (2) abandon territories if surveyed more than twice per month.

The nocturnal calling survey technicians should work in groups of at least 2, as this work will be done late at night, after hours for most businesses. These technicians should also carry a cell phone, GPS unit, maps, and first aid kit, in addition to flashlights or headlamps and possibly a hard hat if working in a forested or rocky area. These crews should also have a sign in/sign out system so that someone is aware of their locations and status. It is advisable to have a plan for emergencies established by the beginning of each field season with information as to who to contact, where to go, and directions to the areas that could be read to a 911 operator if needed.

This plan could be on a laminated piece of paper attached to the clipboard.

Automated Recordings

Use ARU (automated recording units) instead of technicians to record bird calls.

Nest Searching

If a nest happens to be found, please make a note and photograph the nest.

Chapter Twelve:

Butterfly Monitoring Protocol

Butterfly Monitoring

Butterfly Transects

The primary butterfly survey methods used in Iowa entails transect walking, following Pollard and Yates (1993). This transect should be 5 m in width and although the transect lengths may vary due to habitat features, the length should total to approximately 400 m. The transects are expected to pass through several different habitat classifications. The transect dissecting the sampling hexagon is 400 m in length. This middle transect should be considered the butterfly transect as it should cross through the designated habitat type. Should additional transect distance be necessary (i.e. the middle transect will not work for the butterfly transect) then 1 (or 2) other 200-m transects should be established instead. If possible, these transects are also in a north-south direction and can be either the east or west side of the primary transect at a spacing of at least 173.2 m between transects. For ease of effort, these extra transects connect the poles used in the bird point counts, stations #3 & 4 on the east side, and #6 & 7 on the west side. If either or both of the extra transects are to be used to replace part of the primary transect (due to a lake being in the primary transect, for example), a decision should be made and recorded as to which transect will be used so that future crews survey the same area. This decision will be made with the MSIM leadership team. The different transect location should be entered as its own site in the database with specific text detailing the exact placement of the transect in the comments.

The primary transect can be divided into habitat sections and, in a standard plot, is the dividing line of the permanent hexagonal plot. Should a transect cross a paved or major road, this should be treated as a break between sections (Pollard and Yates 1993). The primary transect is 400 m in length. Transects should be flagged every 10 m to ensure the same correct path is followed by various observers and observer is in the correct location. It is wise to label flags with distances from the start of the transect to aid in data collection. The transect should be established with the bamboo poles and flagging when the site is first set up in April to ensure it is ready to go by May. Zero meters should be at the northern most end of the transect with 400 m being at the southern-most end, meaning the flagging would be numbered 1 to 40 moving from north to south along the transect, with 1 being at 0 m.

Due to various disturbances (ie. active crop field or hay field or crew presence is minimal on a property), transects may be completed without the use of flagging, under the direction of the MSIM leadership team. These transects can be completed using a handheld GPS or a phone with the transect line laid out. When using the GPS, a technician will monitor the direction and distance they have traveled from their starting point to the end point. The digital map of the phone will provide a straight transect line, as well as labeled transect sections. With both methods, it should be noted that transect lines will not be exact as both the GPS and Phone have error associated with them and the technician must be proficient in following a line on a GPS while collecting data.

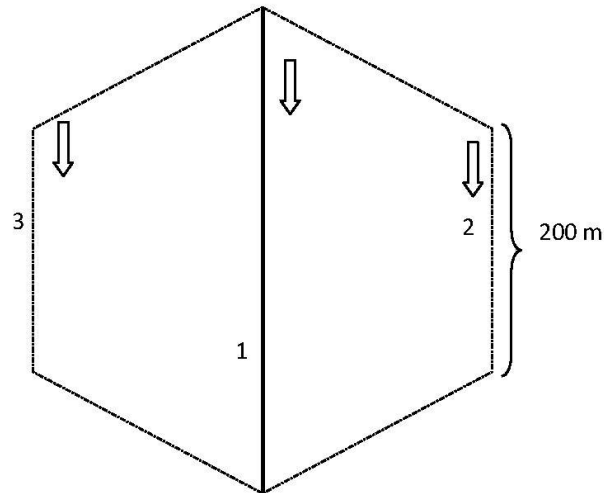
Butterfly Visual Encounter Surveys:

In addition to the transect surveys, a visual encounter survey should be done on the property on at least 2 visits for the butterflies. These surveys can be conducted anywhere on the property that appears to be the best habitat for butterflies, especially skippers and hairstreaks. The 'good habitat' encounter surveys should be 30 minutes in length, conducted in July, and have only one observer. This information will be used to compile species lists for the property and perhaps for occupancy analyses. The purpose of the additional effort is to document butterflies associated with a property in general, not necessarily the habitat the property was chosen to represent. Make sure that the appropriate location name or number is recorded on data sheet. The name for this 'good habitat' should be listed on the aerial photo in the map book created under the Landscape Characteristics protocol (Chapter 3) and should also be entered as a new site within the given property in the database if it is not already delineated as such.

Survey Methods

The primary transect follows the same path as the small mammal trap center line and connects the bird point count stations 2 and 5 while passing through bird point count station 1. Care should be taken to avoid attempting to walk the butterfly transect while mammal trapping is ongoing. The observer walks the transect between the bird point count station locations while searching an area 2.5 m on each side (for a total width of 5 m). All butterflies seen within the 5 m

width and to a distance of approximately 5 m in front of the observer are recorded. Any butterflies seen outside this area or 'box' (5 m wide by 5 m length by 5 m height) can be recorded as incidentals, however, if the butterfly moves into the box (or doesn't move as the observer moves forward such that the butterfly is then inside the box) it should be included as part of the transect data.



The observer should maintain a steady pace, unless a butterfly must be captured in order to be correctly identified to species. Individual butterflies should be counted only once. If the observer is unsure whether an individual is new or not, it should be treated as a new individual. Literature suggests that one will spend approximately 5 minutes per every 50 m with additional time being needed to record data and identify species (Ries et al. 2001). Use a stopwatch if needed to record the amount of time spent, and always stop the time count when capturing or identifying an individual.

Currently, the butterfly surveys are conducted between May 20 and through August 31 when weather is appropriate. A cold start to the summer season will result in the delay of the beginning of the butterfly surveys. If weather conditions are appropriate, the IWAP Butterfly Subcommittee will allow the data collection to begin in early May. All transect searches will be conducted no earlier than 10 am and end by 6:30 pm on any given day. The temperature should be between 21° and 35° C (70-95°F) with winds less than 16 km/hr (~10 mph), when possible. Most surveys should be conducted on sunny weather days. Transects should be visited on 4 different occasions, each separated by at least 2 weeks such that each site is visited at least once in each month of June and August, with 2 visits occurring in July. No human activity should occur in the survey area for at least 30 minutes before the surveys are conducted.

Preservation of Voucher Specimens

Some species, especially skippers, will need to have voucher specimens collected for identification in the lab. Traditional chemicals used to preserve insects have been found to be hazardous to human health. Therefore the best method to preserve butterflies will be to collect them in glassine envelopes in the field, freeze them (at least overnight) in the envelope, pin them, and then re-freeze them for several days. They will not need to be stored in the freezer, but will need to be stored in a sealed Insect Drawer (see BioQuip.com catalogue).

Photos should be taken of butterflies as often as possible to ensure correct identification. These photos should be uploaded into the MSIM Photodatabase.

Habitat & Plant Composition Data Collection

See Chapters 20 and 21. This information will be recorded under those protocols. Any milkweed seen within the transect on any of the surveys should be noted on the datasheet and recorded in the comment field of the survey when entered into the database.

Equipment List

- Compass
- Flagging, flags & tall bamboo stakes

Stopwatch/Phone

Digital camera with macro lens

Butterfly forceps

Glassine envelopes

Pinning kit

Butterfly net

Hand lens

Field guides

Zip-lock baggies

Standard field kit: Clip board, weather kestrel, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets, nail polish or spray paint.

Staff & Training

Training is recommended and should include 1) field guide use and identification, 2) discussion of defining species characteristics, 3) field practice with an experienced observer, and 4) proficiency testing. Technicians will also need training on habitat data collection.

Data Quality & Management

This protocol will be difficult to rate for quality:

- Examination of data will not reveal missed detections or misidentifications.
 - Misidentifications could be checked by either the use of photos, or by the field supervisor working periodically with each technician.
 - Photos should be uploaded to the MSIN Photodatabase and the IWAP Butterfly Subcommittee Chair should be asked for verification.
- Butterflies collected in the field will be double checked in the lab. See Additional Methods for Special Locations for information on collecting and preserving butterfly specimens.
 - Skipper identification is difficult in the field or with photographs. These species may need to have voucher specimens collected.
- All photographs should be reviewed by at least 2 additional people to verify species identifications.

At the end of each survey, each observer should review data sheets to ensure all information present. At the end of the week, the field crew leader should review the collected data sheets.

Data Analysis

The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of area occupied using program R. For more information, see chapter 5 (Data Analysis). The data collected with the transect technique will be used to compute abundance indices when possible.

Safety Issues & Considerations

The butterfly technicians will be working alone and therefore should carry a reliable cell phone, GPS unit, maps, and first aid kit. The crew should maintain a sign in/sign out method to ensure everyone returned from the field as well as to know exactly where each crew member is assigned to work every day.

Target Species

The following list of target species represents the 51 species of greatest conservation need listed in the 3rd Edition (2015 revision) for the Iowa Wildlife Action Plan (Reeder and Clymer 2015). Distribution maps for these species in Iowa can be found in Nekola (1995). Appendix 1 contains a list of additional, more common, butterfly species which may be encountered during the monitoring efforts. An asterisks (*) indicate butterflies designated as Data Deficient and 'L' indicates butterflies listed under the Endangered Species Act.

Target butterfly species:

Common Name	Scientific Name	Habitat
Pipevine swallowtail	<i>Battus philenor</i>	Forest, open fields, & roadsides
Zebra swallowtail	<i>Eurytides marcellus</i>	Woodland along rivers
Spicebush swallowtail	<i>Papilio troilus</i>	Woodlands
Olympia marble	<i>Euchlow olympia</i>	Open woods, river bluffs, poor soils, & grasslands
Harvester	<i>Feniseca tarquinius</i>	Woodland and stream
Purplish copper	<i>Lycaena helloides</i>	Moist or disturbed areas
Acadian hairstreak	<i>Satyrrium acadica</i>	Riparian & oldfield
Edward's hairstreak	<i>Satyrrium edwardsii</i>	Woodlands, clearings, & areas of poor soil
Hickory hairstreak	<i>Satyrrium caryaevorum</i>	Forest
Striped hairstreak	<i>Satyrrium liparops</i>	Forest openings and edges, prairie streamsides
White M hairstreak*	<i>Parrhasius m-album</i>	Woodland, savanna
Henry's elfin	<i>Callophrys henrici</i>	Woodland
Reakirt's blue	<i>Echinargus (Hemiargus) isola</i>	Native prairie
Silvery blue	<i>Glaucopsyche lygdamus</i>	Open fields & woodland openings
Melissa blue	<i>Plebejus (Lycaeides) melissa</i>	Xeric prairie and gravel ridges
Aphrodite fritillary	<i>Speyeria Aphrodite</i>	High quality prairie, wetlands, and fens
Regal fritillary	<i>Speyeria idalia</i>	Prairie & open grassland
Silver-bordered fritillary	<i>Boloria selene</i>	Fens, wet prairie, and meadows
Gorgone checkerspot	<i>Chlosyne gorgone</i>	Oldfield roadsides, pastures, vacant lots, and native prairie
Baltimore checkerspot	<i>Euphydryas phaeton</i>	Wetlands
Ozark Baltimore checkerspot*	<i>Euphydryas phaeton ozarkae</i>	Wetlands, fens, bogs, woodlands
Compton Tortoiseshell	<i>Nymphalis vaualbum (l-album)</i>	Large tracts of forest
Common ringlet	<i>Coenonympha tullia</i>	Prairie & marsh edge
Eyed brown	<i>Satyrodes eurydice</i>	Fens, wet prairies, and marshes
Monarch	<i>Danaus plexippus</i>	Open habitat and disturbed areas
Southern cloudywing	<i>Thorybes bathyllus</i>	Xeric prairie
Hayhurst's scallopwing*	<i>Staphylus hayhurstii</i>	Floodplain forest, Loess Hills forests
Dreamy duskywing*	<i>Erynnis icelus</i>	Woodland or edge
Sleepy duskywing	<i>Erynnis brizo</i>	Oak barrens, sand or shale soils
Juvenal's duskywing	<i>Erynnis juvenalis</i>	Oak forests
Mottled duskywing	<i>Erynnis martialis</i>	Xeric prairie
Columbine duskywing	<i>Erynnis lucilius</i>	Rocky wooded ravines
¹ Powesheik skipperling	<i>Oarisma powesheik</i>	High-quality tallgrass prairie
Ottoo skipper	<i>Hesperia ottoe</i>	Mid- and tall grass, high-quality prairie
Leonard's skipper	<i>Hesperia leonardus</i>	Open grassy areas
Dakota skipper ^L	<i>Hesperia dacotae</i>	Prairie
Crossline skipper	<i>Polites origines</i>	Xeric prairie
Long dash	<i>Polites mystic</i>	Xeric prairie in northwest, fens and wet prairies in northeast

Common Name	Scientific Name	Habitat
Northern broken-dash	<i>Wallengrenia egeremet</i>	Fens, xeric prairie, forest/woodlands
Little glassywing	<i>Pompeius verna</i>	Woodland edge
Arogos skipper	<i>Atrytone arogos</i>	Prairies & grasslands
Byssus skipper	<i>Problema byssus</i>	Tallgrass prairie
Mulberry wing	<i>Poanes massasoit</i>	Wetland fens
Broad-winged skipper	<i>Poanes viator</i>	Wetland fens
Dion skipper	<i>Euphyes dion</i>	Sedge wetlands
Black dash	<i>Euphyes conspicua</i>	Fens, wet prairies, marshes
Two-spotted skipper	<i>Euphyes bimacla</i>	Sedge meadows & marshes
Dusted skipper	<i>Atrytonopsis hianna</i>	Bluestem grasslands & oldfields
Pepper and salt skipper	<i>Amblyscirtes hegon</i>	Edge of woods & grass waterways
Common roadside skipper*	<i>Amblyscirtes vialis</i>	High quality, xeric prairie
Swarthy skipper	<i>Nastra lherminier</i>	

Additional Methods for Special Locations

Mark-Recapture

This technique would involve walking the transect several times during the same day or each day for several days in a row. All butterflies (or all butterflies of a target species) would be captured using a butterfly net and given a mark on the wing using either a permanent marker or a small dab of paint.

Suggested Field Guides

Glassberg, J. 1999. Butterflies through Binoculars: The East. Oxford University Press. New York, NY.

Heitzman, JR, and JE Heitzman. 1987. Butterflies and Moths of Missouri. Missouri Department of Conservation. Jefferson City, MO.

Marrone, G. 2002. A Field Guide to Butterflies of South Dakota. South Dakota Department of Game, Fish, and Parks. Pierre, SD.

Scott, JA. 1992. Butterflies of North America: A Natural History and Field Guide. Stanford University Press. Stanford, CA.
(This one should be left in the lab or office).

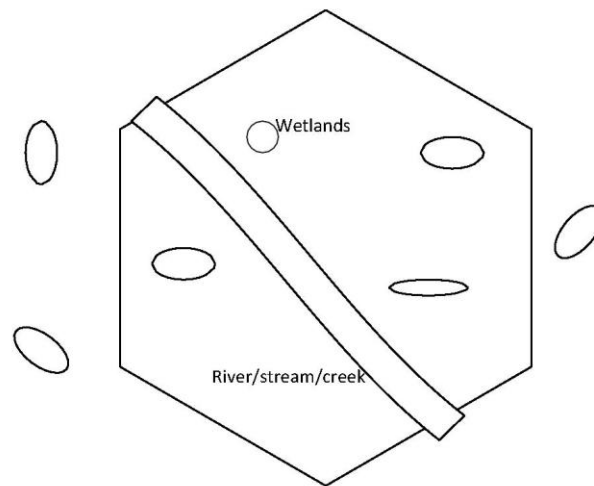
Chapter Thirteen:

Damselfly and Dragonfly Monitoring Protocol

Dragonfly & Damselfly Monitoring

Exuviae (the remains of the exoskeletons left behind when the dragonfly or damselfly has molted) are the most important indicators of resident populations of odonates. They are the most important because the presence of exuviae indicate healthy, reproducing populations. These exoskeletons can be collected without impacting odonate populations and identified at a later date in the lab. However, this protocol will search for adult dragonflies and damselflies due to the fact that the adults are much easier to find and identify.

Timed visual encounter surveys (VES) will be the primary method used in this protocol. VES is inexpensive, easy to implement, and efficient over diverse habitats (Manley et al. 2004). Additional benefits of VES include low site disturbance, low animal mortality, ease of implementation in terrestrial or aquatic environments, and other animals can be detected at the same time. Appropriate habitats within the property should be surveyed. This area may extend beyond the hexagon boundary, but will allow for a large variety of habitat types to be searched.



All relevant wetlands and also surrounding uplands should be searched during the timed VES. Odonates are known to fly into the surrounding uplands, primarily to forage, and therefore a smaller amount of time should be spent in these areas by the observer. If a hexagonal sampling plot has few wetlands, increase the search area to include wetlands within the 1-km² (~250 ac) area around the center point. Stay within the property boundaries.

Survey Methods

Field Methods

All wetland habitats should be searched for adult odonates. Each site should be visited at least 6 times per year, twice between mid-April through mid-June, twice between mid-June through mid-August, and twice between mid-August through mid-October. The [Iowa Odonata Survey](#) website (last accessed May 13, 2025) has time-of-year activity calendars for adult odonates in addition to records of odonates by county. This information can be used to assist in choosing when to conduct site visits in each county for the species of greatest conservation need.

Each site visit should be for a minimum of 4 search-hours per visit. Therefore if 2 technicians are searching the same hexagonal plot, each should search for 2 hours. If 4 technicians are searching the plot, only one hour apiece is needed as long as assigned survey areas have no overlap, meaning that the observers have taken different areas to survey and are not surveying the same ground twice. Presence of all adult species is recorded for each site (e.g. farm pond A, small creek B, prairie pond E, etc.). In addition to species presence, be sure to record the number of individuals seen. Remember that this number is per site, not the entire property. It is possible that an observer would detect a species in more than one habitat creating a higher final density of these animals for the entire property, but it is important to create a record of relative abundance indices based on the smaller locations in addition to the overall property

abundance. However, we tally the number of individuals seen, attempting to make every effort NOT to count the same individual more than once.

Similarly to the other VESs conducted with this monitoring design, searches should be conducted at varying times of the day. Do not always return to the site between 9 and 11 am, for example, vary the visits to cover morning, afternoon, and evening times depending on the species being targeted during that search. Morning, noon, and afternoon visits are best.

Species Vouchers

It may be necessary to collect a voucher specimen of adult odonates for later identification or proof of identification. To collect adult odonates, they should be placed individually into glassine envelopes that are then dipped into acetone (completely covering the envelope) for at least 10 seconds, some species like larger dragonflies may need ~5 minutes. Most specimens can be left in the glassine envelopes and acetone for a longer period of time (>24hrs) to dry and preserve for later identification. Before storing an individual in acetone, straighten the body and wings to ensure the specimen is in good shape. Be sure that the container holding the acetone is marked with “poison” as acetone can be absorbed by plastic - once it has been used with acetone, the container should never again be used for food storage.

Individuals with pruinescence (grey, white, or light blue pigment on body) should not be dried with acetone as it will change these colors. These species should be freezer killed and then dried by a light bulb.

Post killing, odonates should be positioned in the desired manner (either pinned and flat as a standard insect collection or flat on its side with the wings over the abdomen and placed back into the envelope) for drying. If in the envelope, it can be returned back to the acetone for 24 hours, otherwise it can be placed into direct sunlight to dry (NCOS 2002).

Equipment List

- Digital camera with macro lens
- Butterfly net
- Hand lens
- Binoculars
- Compass
- Plastic containers for collecting exuviae
- Glassine envelopes for collecting adults
- Acetone and container for killing adults
- Standard field kit: Clip board, pencils, weather kestrel, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets, nail polish or spray paint.

Staff & Training

Training is recommended and should include 1) field guide use and identification, 2) discussion of defining species characteristics, 3) field practice with an experienced observer, and 4) proficiency testing.

Data Quality & Management

Female dragonflies and damselflies are difficult to identify as they are more subtly colored than the males. Rosche (2002) suggests the best time to identify the female is while she is attached to the male during mating.

This protocol will be difficult to rate for quality:

- Examination of data will not reveal missed detections or misidentifications.
 - Misidentifications could be checked by either the use of photos, or by the field supervisor working periodically with each technician.
- All photographs should be reviewed by at least 2 additional people to verify species identifications.
- Some identifications will require the collection and examination of a specimen.

At the end of each survey, each observer should review data sheets to ensure all information present. At the end of the week, the field crew leader should review the collected data sheets as well.

Data Analysis

The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of area occupied using program R. For more information, see chapter 5 (Data Analysis). The data collected with this technique will be used to compute abundance indices when possible.

Safety Issues & Considerations

The odonate technicians should carry a reliable cell phone, GPS unit, maps, and first aid kit. The crew should maintain a sign in/sign out method to ensure everyone returned from the field as well as to know exactly where each crew member is assigned to work every day.

Target Species

The following list of target species represents the 30 species of greatest conservation need listed in the 3rd Edition (2015 revision) for the Iowa Wildlife Action Plan (Reeder and Clymer 2015). Distribution maps for these species can be found at the [Iowa Odonata Survey](#) website. Appendix 1 contains a list of additional, more common, odonate species which may be encountered during the monitoring efforts. An astrisks (*) indicate odonates designated as Data Deficient.

Target dragonflies & damselflies species:

Common Name	Scientific Name	Habitat
Spotted Spreadwing	<i>Lestes congener</i>	Edge of pools, marsh
Amber-winged spreadwing	<i>Lestes eurinus</i>	Fishless shallow ponds and wetlands
Sweetflag spreadwing	<i>Lestes forcipatus</i>	Marsh, pond edge
Paiute dancer	<i>Argia alberta</i>	Small streams, road ditches
Springwater dancer	<i>Argia plana</i>	Small, shallow streams with canopy cover and clay substrate
*Prairie bluet	<i>Coenagrion angulatum</i>	Lakes, ponds
*Taiga bluet	<i>Coenagrion resolutum</i>	Ponds & wetlands
*Boreal bluet	<i>Enallagma boreale</i>	Marsh
*Alkali bluet	<i>Enallagma clausum</i>	Pond edges without vegetation
*Western forktail	<i>Ischnura perparva</i>	Heavily vegetated ponds, lakes, and slow flow streams with mud substrate
Sedge sprite	<i>Nehalennia irene</i>	Ponds & sedge meadows
*Canada darner	<i>Aeshna canadensis</i>	Marsh, pond edge
*Variable darner	<i>Aeshna interrupta</i>	Lakes, ponds, streams
Midland clubtail	<i>Gomphus fraternus</i>	Creeks & rivers with rock & mud
Sulphur-tipped clubtail	<i>Gomphus militaris</i>	Artificial ponds, lakes
*Rapids clubtail	<i>Gomphus quadricolor</i>	Rocky creeks
Rusty snaketail	<i>Ophiogomphus rupinsulensis</i>	Sandy, rocky creeks
*Pale snaketail	<i>Ophiogomphus severus</i>	Privers & streams with fast flow & cobble substrate
Sioux snaketail	<i>Ophiogomphus smithi</i>	Sand-bottomed creeks
*Westfall's snaketail	<i>Ophiogomphus westfalli</i>	Clear forest streams with strong riffles & cobble substrate
*Brimstone clubtail	<i>Stylurus intricatus</i>	Sandy streams
Elusive clubtail	<i>Stylurus notatus</i>	Creeks & rivers with sandy substrate
*Arrow clubtail	<i>Stylurus spiniceps</i>	Rivers with sandy substrate
Stream cruiser	<i>Didymops transversa</i>	Medium to large streams & rivers
Royal river cruiser	<i>Macromia taeniolata</i>	Lakes, rivers
Slender baskettail	<i>Epithera costalis</i>	Lakes, ponds, and backwaters

Common Name	Scientific Name	Habitat
Smoky shadowdragon	<i>Neurocordulia molesta</i>	Large rivers
*Stygian shadowdragon	<i>Neurocordulia yamaskanensis</i>	Mississippi River
Plains emerald	<i>Somatochlora ensigera</i>	Prairie streams
Carolina saddlebags	<i>Tramea Carolina</i>	Marsh

Additional Methods for Special Locations

Exuviae Search

Additional searches for discarded exuviae can be done to verify the use of a wetland for reproduction. To find exuviae, a thorough search should be made of riparian vegetation, emergent plants, dead wood, and abiotic riparian structure (such as banks and graveled ground) during each sampling visit (Chovanec and Waringer 2001).

All exuviae should be collected in plastic containers for later identification in the lab using a dissection microscope. Plastic containers are best because they are rigid enough to prevent the exoskeleton from being crushed, but will not break like glass might. Old film containers, plastic tackle boxes, or plastic craft (bead) boxes are all potential exuviae transporters.

The exuviae collected in the field need to be identified to species in the lab using field guides or keys and a dissecting scope.

Larval Odonates

Any larval odonates that were collected during the aquatic invertebrate sampling can be identified at this time as well.

Chapter Fourteen:

Bumble Bee Monitoring Protocol

Bumble Bee Surveys

Bumble Bee Transects

The primary bumble bee survey method used for MSIM entails transect walking, following U.S. Fish and Wildlife Service (2019) “Recovery Monitoring (Bombus Community) Survey Protocol”. This transect should be 2m in width and although the transect lengths may vary due to habitat features, the length should total to approximately 400m. The transect is expected to pass through several different habitat classifications. This transect, when possible, should dissect the hexagon to cross through the designated habitat type. The same butterfly transect line should be used for the basis of the bumble bee transects. Transect surveys should be completed by only one observer per survey.

Should additional transect distance be necessary (i.e. the middle transect will not work for the butterfly transect) then 1 (or 2) other 200-m transects should be established instead. If possible, these transects are also in a north-south direction and can be either the east or west side of the primary transect at a spacing of at least 173.2 m between transects. For ease of effort, these extra transects connect the poles used in the bird point counts, stations #3 & 4 on the east side, and #6 & 7 on the west side. If either or both of the extra transects are to be used to replace part of the primary transect (due to a lake being in the primary transect, for example), a decision should be made and recorded as to which transect will be used so that future crews survey the same area. This decision will be made with the MSIM leadership team. The different transect location should be entered as its own site in the database with specific text detailing the exact placement of the transect in the comments.

The primary transect can be divided into habitat sections and is the dividing line of the permanent hexagonal plot. Should a transect cross a paved or major road, this should be treated as a break between sections (Pollard and Yates 1993). The primary transect is 400 m in length. Transects should be flagged to ensure that the observer is in the correct area. Transects should be flagged every 10 m to ensure the same correct path is followed by various observers. It is wise to label flags with distances from the start of the transect to aid in data collection. The transect should be established with the bamboo poles and flagging when the site is first set up in April to ensure it is ready to go by May. Zero meters should be at the northern most end of the transect with 400 m being at the southern-most end, meaning the flagging would be numbered 1 to 40 moving from north to south along the transect, with 1 being at 0 m.

Due to various disturbances (i.e. active crop field or hay field or crew presence is minimal on a property), and advances in technology, transects may be completed without the use of flagging, under the direction of the MSIM leadership team. These transects can be completed using a handheld GPS or a GPS enabled phone with the transect line/segments laid out. When using the GPS, a technician will monitor the direction and distance they have traveled from their starting point to the end point. The digital map on the phone will provide a straight transect line, as well as labeled transect sections. With both methods, it should be noted that transect lines will not be exact as both the GPS and Phone have error associated with them.

Bumble Bee VES

In addition to the transect surveys, a visual encounter survey should be done on the property on at least 2 visits for bumble bees. These surveys can be conducted anywhere on the property that appear to be the best habitat for bumble bees, such as flowering prairie/grassland patches or anywhere with flowering forbs. The ‘good habitat’ encounter surveys should be 30 minutes in length, conducted once in July and once in August, and only have one observer. These surveys must be completed at least 10 days apart and must follow the weather standards of the bumble bee transect.

This information will be used to compile species lists for the property and possible for occupancy analysis. The purpose of the additional effort is to document bumble bees associated with a property in general, not necessarily the habitat the property was chosen to represent. Make sure that the appropriate location name or number is recorded on data sheet. The name for this ‘good habitat’ should be listed on the aerial photo in the map book created under the Landscape Characteristics protocol (Chapter 3) and should also be entered as a new site within the given property in the database if it is not already delineated as such. Therefore, should the site not already be named in your GPS and Map

Books, please ensure you have recorded an appropriate UTM (Zone 15) with the correct location and send that information to the appropriate supervisor to be entered as a location in the database.

Survey Methods

The primary transect follows the same path as the small mammal trap center line and butterfly transect and connects the bird point count stations 2 and 5 while passing through bird point count station 1.

Care should be taken to avoid attempting to walk the transect while mammal trapping is ongoing. The observer walks the transect between the bird point count station locations while searching an area 1 m on each side (for a total width of 2 m). All bumble bees seen within the 2 m width and to a distance of approximately 2 m in front of the observer are recorded. Any bumble bees seen outside this area or 'box' (2 m wide by 2 m length by 2 m height) can be recorded as incidentals, however, if the bumble bee moves on its own accord into the box (or doesn't move as the observer moves forward such that the bumble bee is then inside the box) it should be included as part of the transect data.

The observer should maintain a steady pace, unless a bumble bee must be captured. Individual bumble bees should be counted only once. If the observer is unsure whether an individual bumble bee is new or not, it should be treated as a new individual. Literature suggests that one will spend approximately 15 minutes every 100 m with additional time being needed to record data and identify species (USFWS 2019). Use a stopwatch if needed to record the amount of time spent, and always stop the time count when capturing (ie. chasing or sneaking up on the bee for a photo) or identifying individuals.

Bumble bee transects are conducted once a month between May and September when weather is appropriate. The temperature should be between 60-90°F with winds <15-20 mph, when possible. Most surveys should be conducted on sunny weather days, and surveys cannot be conducted in the rain. Although visits occur only once a month, the surveys should be at least 10 days apart. No human activity should occur in the survey area for 30 minutes before the surveys are conducted.

Capturing & Handling Specimens

On some properties, when possible, an observer should catch all bumble bees seen to properly ID and photograph each specimen. Try and capture one bee at a time to ensure you get an accurate count of each species seen on the transect. If needed, you may estimate numbers of each species, but this is only recommended for commonly observed species. The following resources can be used to help learn the technique to capturing and transferring the bumble bees out of the net:

- How to Net a Bumble Bee: <https://youtu.be/mVOi-jRpw2E> & <https://www.youtube.com/watch?v=SwYbv5bySPQ>
- How to transfer a Bumble Bee: <https://youtu.be/TjaOpZWNF4M>, Videos last accessed; 4/14/2025

Due to logistics, MSIM does not chill the bees before processing. Instead, MSIM will use a photo tube. Our photo tubes are a modified marking tube for honey bees that has window insulation plastic to create a clear, elastic photo window. Once a bumble bee is inside the photo tube, take photos from as many angles as possible or take a video of the bee moving around in the tube to capture high quality still photos. Take care to not crush the bumble bee with the plunger. Getting photos of the side profile, abdomen and thorax, face, underside of abdomen, tip of abdomen, and hind leg are all beneficial in obtaining a correct ID. Do not keep bee in photo tube for more than a few minutes to keep them from overheating.

However, swinging a net at a bumble bee in a High or Low Rusty Patch Bumble Bee (*Bombus affinis*) zone (U.S. Fish & Wildlife, 2025) is strictly prohibited. Surveys conducted in these areas will be photograph only. This means that only visual sightings and photographs of any bumble bee species must be done from a short distance away, ensuring that the bumble bee is not being disturbed. If a Rusty Patch Bumble Bee is captured outside a high or low potential zone, safely and quickly photograph the individual and release it unharmed. Then **IMMEDIATELY** report the finding to the MSIM leadership team. In the future, a net can not be used during bumble bee surveys at this property.

The MSIM Leadership Team will provide you with maps so you know which properties are within HPZ/LPZ areas.

However, you can also find that information on the [Rusty Patched Bumble Bee Map](#) website, last accessed 4/10/2025.

Photo Procedure

On properties that you cannot swing a net, consider these guidelines to capture photos that can verify bumble bee ID. Get as close as possible without disturbing the bumble bee. Move slowly and deliberately to avoid startling or disturbing the bee. Be patient and wait for the right opportunity to get a good shot. Take continual shots or a video to be able to select the best photos to ID the bumble bee. Make sure to provide a spacer photo in between unique individuals. When photos are taken from farther away, crop the photo to put the focus on the bumble bee before you upload the photo to the photo database.

Habitat & Plant Composition Data Collection

See Chapters 20 and 21. This information will be recorded under those protocols. Additionally, nectaring plants will be recorded during the surveys and noted in the comment field of the survey when entered into the database.

Equipment List

Compass
Flagging, flags & tall bamboo stakes or access to a digital transect
Stopwatch/Phone
Butterfly net
Photo tube: modified honey bee marking tube & window plastic insulation
Field guides
Digital camera/Phone
Standard field kit: Clip board, weather kestrel, pencils, ruler, Sharpie markers, hand sanitizer, & data sheets

Staff & Training

Training is recommended and should include 1) field guide use and identification, 2) discussion of defining species characteristics, 3) discussion and review of Rusty Patch Bumble Bee specific protocols 4) field practice with an experienced observer, and 5) proficiency testing. Technicians will also need training on habitat data collection.

Data Quality & Management

This protocol will be difficult to rate for quality:

- Examination of data will not reveal missed detections or misidentifications.
- Misidentifications could be checked by either the use of digital cameras, or by the field supervisor working periodically with each technician.
- Photos should be uploaded to the MSIM Photo Database and the IWAP Bee Subcommittee Chairs should be asked to check them for verification.
- At the end of each survey, each observer should review data sheets to ensure all information is present.

Data Analysis

The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of area occupied using program R. For more information, see chapter 5 (Data Analysis). The data collected with the transect technique will be used to compute abundance indices when possible.

Safety Issues & Considerations

The bumble bee technicians will be working alone and therefore should carry a reliable cell phone, GPS unit, maps, and first aid kit. The crew should maintain a sign in/sign out method to ensure everyone returned from the field as well as to know exactly where each crew member is assigned to work every day. Those with a known bee allergy, should not take part in the survey. If a crew member has a known bee allergy, all crew member should know where an epi pen is and how to use it, regardless if the allergic crew member is not completing these surveys.

Target Species (coming later in 2025)

The following list of target species represents the 13 species of greatest conservation need listed in the 4th Edition (2025

revision) for the Iowa Wildlife Action Plan. Appendix 1 contains a list of additional, more common, bumble bee species which may be encountered during the monitoring efforts. An astrisks (*) indicate bumble bee species designated as Data Deficient and 'L' indicates bumble bee species listed under the Endangered Species Act.

Target bumble bee species:

Common Name	Scientific Name	Habitat
Rusty Patched Bumble Bee	<i>Bombus affinis</i>	
Black and Gold Bumble Bee	<i>Bombus auricomus</i>	
Two-spotted Bumble Bee	<i>Bombus bimaculatus</i>	
Ashton's Cuckoo Bumble Bee	<i>Bombus bohemicus</i>	
Northern Amber Bumble Bee	<i>Bombus borealis</i>	
Lemon Cuckoo Bumble Bee	<i>Bombus citrinus</i>	
Yellow Bumble Bee	<i>Bombus fervidus</i>	
Southern Plains Bumble Bee	<i>Bombus fraternus</i>	
Brown-belted Bumble Bee	<i>Bombus griseocollis</i>	
Common Eastern Bumble Bee	<i>Bombus impatiens</i>	
American Bumble Bee	<i>Bombus pensylvanicus</i>	
Tri-colored Bumble Bee	<i>Bombus ternarius</i>	
Half-black Bumble Bee	<i>Bombus vagans</i>	
Variable Cuckoo Bumble Bee	<i>Bombus variabilis</i>	
Red-belted Bumble Bee	<i>Bombus rufocinctus</i>	
Frigid Bumble Bee	<i>Bombus frigidus</i>	
Morrison Bumble Bee	<i>Bombus morrisoni</i>	
Confusing Bumble Bee	<i>Bombus perplexus</i>	
Yellow-banded Bumble Bee	<i>Bombus terricola</i>	

Chapter Fifteen: Terrestrial Snail Monitoring Protocol

This protocol is no longer implemented.

There are no protocols for these species in the USFS MSIM Program. However, the US Fish and Wildlife Service has developed a protocol for monitoring the Iowa Pleistocene snail (Henry et al. 2003) which has been adapted below to find terrestrial snails of other species in additional habitats. The Iowa WAP (Zohrer et al. 2005) has designated 8 terrestrial snails as species of greatest conservation concern. The Plan also states that there is no comprehensive list of terrestrial snail species occurrence in Iowa.

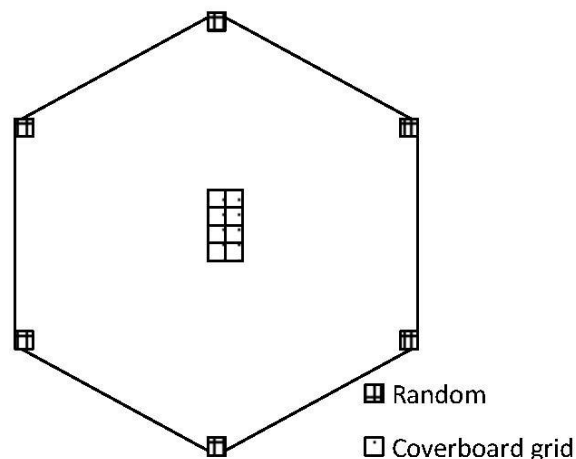
Following 1-2 years of attempting to implement this protocol and failing to be able to correctly identify snail species, this protocol has not been used since. We have no plans to fully implement the protocol in the near future.

Snail Monitoring

While the 8 species of greatest conservation need are all associated with Algific slopes, Iowa needs additional information as to the other terrestrial snails that may occur within the state in other habitats. Therefore, this protocol could be implemented at every permanent sampling location.

Coverboards are the primary method that will be used to monitor terrestrial snails.

These boards are smaller than those used in the Amphibian & Reptile protocol, although all snails and herpetofauna encountered under either size of board should be recorded. For snails, two different coverboard arrangements will be used at each 26-acre (10.5 ha) site. A grid of 8 boards will be used near the center point of the hexagon. Remember that the hexagonal sampling plot is centered on the primary habitat classification for that site. This grid of coverboards should ensure that the given habitat type is adequately sampled. In addition, 6 coverboards will be placed within the hexagon such that 1 board occurs at each hexagon point.



Survey Methods

All coverboards should be 8 x 24 inches (20.3 x 70 cm) and made of either cardboard or wood (basswood or oak species of wood). It is important that the wood not be treated with chemicals. Alternatively, corrugated cardboard may be better in some habitats. The coverboards need to be able to remain moist (Anderson 2004, Henry et al. 2003). Materials that dry quickly should not be used. Each of the 6 boards placed at the bird point count locations (so 1 board per location) should be marked in a grid pattern as that in Figure 2. Each block is 2 x 2 inches (5 x 5 cm) in size. These individual coverboards are important in two respects. The first is in monitoring additional habitats associated with a sampling area, provided that the hexagonal points fall in habitats other than that used for the site classification. The second reason for using the additional coverboards is for the assessment of spatial aggregation (Henry et al. 2003).

Should snail populations be declining, it is conceivable that this could manifest in an increased aggregation of the remaining individuals in suitable habitat (Henry et al. 2003).

Grid pattern on each of the coverboards placed at each hexagonal point. Write the numbers below on the coverboard itself before deploying to the field.

1,4	2,4	3,4	4,4	5,4	6,4	7,4	8,4	9,4	10,4	11,4	12,4
1,3	2,3	3,3	4,3	5,3	6,3	7,3	8,3	9,3	10,3	11,3	12,3
1,2	2,2	3,2	4,2	5,2	6,2	7,2	8,2	9,2	10,2	11,2	12,2
1,1	2,1	3,1	4,1	5,1	6,1	7,1	8,1	9,1	10,1	11,1	12,1

By recording the location of the snails on the grid, additional data analysis can be conducted with regard to distance traveled. This is very little additional work (recording location on a grid) for potentially large information gain.

The grid of coverboards, placed near the center point of the hexagonal sampling plot, should be numbered as above except that these numbers will range from 1,1 to 24,16. All coverboards should be soaked in water before being deployed - preferably in water on the sampling property (creek, pond, etc.). Periodically the soil underneath the coverboard should be soaked with water to maintain the moisture level.

The design of this protocol calls for these coverboards to be checked every time the site is visited by any given technician. This should result in at least 19 checks between April and October (e.g. 9 bird point count visits, 4 small mammal trapping associated visits, and 6 amphibian and reptile visual encounter surveys = 19 occasions to also check snail boards). A data sheet for snails should be filled out at each of these 19 coverboard checking occasions.

Snails may live for 3 or more years. Since they are also not believed to travel long distances (although no data has been published on this, a FWS study indicates that if the Iowa Pleistocene snail moved constantly in a straight line it would disperse 14.7 m in a year (Henry et al. 2003)), it may be prudent to permanently mark snails.

To mark snails, colored, numbered bee tags can be glued onto the shell with superglue.

Henry et al. (2003) recommend that the numbers 6 and 9 be avoided and that juvenile snails less than 5 mm in length be marked with paint. Shell diameter and height should be measured to the nearest 0.5 mm and the height and width of the shell opening should be measured as well. The number of whorls of the shell should also be recorded.

Environmental Data Collection

Environmental variables such as air and soil temperature and other weather conditions should be recorded at the time of the survey on the snail monitoring data sheet.

Equipment List

- 14 Coverboards - 8 x 24 inches (20.3 x 70 cm) and of oak, basswood wood, or corrugated cardboard. These should already have the grid drawn on them.
- Water to maintain moisture under boards
- Bee tags and superglue, paint for juvenile shells
- Calipers or ruler
- Hand lens
- Plastic baggies
- Paper towels

Air and soil thermometers
 Field guides
 Dissecting scope at lab or office
 Standard field kit: Clip board, pencils, ruler, weather kestrel, small scissors, Sharpie markers, compass, hand sanitizer, & data sheets.

Staff & Training

Two weeks of training is recommended and should include 1) field guide use and identification, 2) trips to University museums to discuss defining species characteristics, 3) field practice with an experienced observer, and 4) proficiency testing.

Data Quality & Management

Snail species are extremely difficult to identify and this will be difficult to rate for quality unless snails are collected and sent for identification. All snail shells that are found should be collected for species confirmation. Other than species identification this protocol should be straightforward to implement. Many species would need to be collected alive and transported to the lab to be identified with the aid of a dissecting scope.

At the end of each trapping day, each observer should review data sheets to ensure all information present. At the end of the week, the field crew leader should review the collected data sheets as well.

Data Analysis

The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of area occupied using program R. For more information, see chapter 5 (Data Analysis). Given the distance between the coverboards (about 200 m), the 6 outer boards and the center group of boards could be analyzed as 7 different areas because snails are thought to be capable of moving less than 15 m in a year (Henry et al. 2003).

If the boards are visited each day for 4-5 days, population size estimates and survival probabilities can be computed for each of the 7 areas in the hexagon as well, depending upon the number of recaptures found on the boards.

Safety Issues & Considerations

Proper hygiene should be followed after handling snails.

Target Species

Target snail species:

Common Name	Scientific Name	Habitat
Iowa Pleistocene snail	<i>Discus macclintocki</i>	Algific slopes
Minnesota Pleistocene snail	<i>Novasuccinea n. Sp. minnesota a</i>	Moderate slopes
Iowa Pleistocene succinea	<i>Novasuccinea n. Sp. minnesota b</i>	Moderate slopes
Hubricht's vertigo	<i>Vertigo hubrichti</i>	Algific slopes
Bluff vertigo	<i>Vertigo meramecensis</i>	Limestone or dolomite cliffs & outcrops

Additional Methods for Special Locations

Time Constrained Searching

The time constrained search method has not been as successful in detecting terrestrial snails as the coverboard method (Henry et al. 2003 and unpublished references therein).

However, this represents a more traditional method. To do this, the observer searches through litter and under rocks and logs to find terrestrial snails. An additional spin on this method is to collect the litter layer and bring it into the lab

for sorting and species identification (Kappes 2005). This method is more destructive to both the habitat and the snail population and is not recommended. However, it should be noted that the protocol to be implemented will only allow for the detection of cellulosic (wood) eating snails, while timed VESs should also detect other snail species.

Suggested Field Guides

Leonard, AB. 1959. Handbook of Gastropods in Kansas. The State Printing Plant. Topeka, Kansas.

Grid pattern on the coverboards placed near the center of the hexagon. Dark lines indicate coverboard edges.

1,16	2,16	3,16	4,16	5,16	6,16	7,16	8,16	9,16	10,16	11,16	12,16	13,16	14,16	15,16	16,16	17,16	18,16	19,16	20,16	21,16	22,16	23,16	24,16
1,15	2,15	3,15	4,15	5,15	6,15	7,15	8,15	9,15	10,15	11,15	12,15	13,15	14,15	15,15	16,15	17,15	18,15	19,15	20,15	21,15	22,15	23,15	24,15
1,14	2,14	3,14	4,14	5,14	6,14	7,14	8,14	9,14	10,14	11,14	12,14	13,14	14,14	15,14	16,14	17,14	18,14	19,14	20,14	21,14	22,14	23,14	24,14
1,13	2,13	3,13	4,13	5,13	6,13	7,13	8,13	9,13	10,13	11,13	12,13	13,13	14,13	15,13	16,13	17,13	18,13	19,13	20,13	21,13	22,13	23,13	24,13
1,12	2,12	3,12	4,12	5,12	6,12	7,12	8,12	9,12	10,12	11,12	12,12	13,12	14,12	15,12	16,12	17,12	18,12	19,12	20,12	21,12	22,12	23,12	24,12
1,11	2,11	3,11	4,11	5,11	6,11	7,11	8,11	9,11	10,11	11,11	12,11	13,11	14,11	15,11	16,11	17,11	18,11	19,11	20,11	21,11	22,11	23,11	24,11
1,10	2,10	3,10	4,10	5,10	6,10	7,10	8,10	9,10	10,10	11,10	12,10	13,10	14,10	15,10	16,10	17,10	18,10	19,10	20,10	21,10	22,10	23,10	24,10
1,9	2,9	3,9	4,9	5,9	6,9	7,9	8,9	9,9	10,9	11,9	12,9	13,9	14,9	15,9	16,9	17,9	18,9	19,9	20,9	21,9	22,9	23,9	24,9
1,8	2,8	3,8	4,8	5,8	6,8	7,8	8,8	9,8	10,8	11,8	12,8	13,8	14,8	15,8	16,8	17,8	18,8	19,8	20,8	21,8	22,8	23,8	24,8
1,7	2,7	3,7	4,7	5,7	6,7	7,7	8,7	9,7	10,7	11,7	12,7	13,7	14,7	15,7	16,7	17,7	18,7	19,7	20,7	21,7	22,7	23,7	24,7
1,6	2,6	3,6	4,6	5,6	6,6	7,6	8,6	9,6	10,6	11,6	12,6	13,6	14,6	15,6	16,6	17,6	18,6	19,6	20,6	21,6	22,6	23,6	24,6
1,5	2,5	3,5	4,5	5,5	6,5	7,5	8,5	9,5	10,5	11,5	12,5	13,5	14,5	15,5	16,5	17,5	18,5	19,5	20,5	21,5	22,5	23,5	24,5
1,4	2,4	3,4	4,4	5,4	6,4	7,4	8,4	9,4	10,4	11,4	12,4	13,4	14,4	15,4	16,4	17,4	18,4	19,4	20,4	21,4	22,4	23,4	24,4
1,3	2,3	3,3	4,3	5,3	6,3	7,3	8,3	9,3	10,3	11,3	12,3	13,3	14,3	15,3	16,3	17,3	18,3	19,3	20,3	21,3	22,3	23,3	24,3
1,2	2,2	3,2	4,2	5,2	6,2	7,2	8,2	9,2	10,2	11,2	12,2	13,2	14,2	15,2	16,2	17,2	18,2	19,2	20,2	21,2	22,2	23,2	24,2
1,1	2,1	3,1	4,1	5,1	6,1	7,1	8,1	9,1	10,1	11,1	12,1	13,1	14,1	15,1	16,1	17,1	18,1	19,1	20,1	21,1	22,1	23,1	24,1

Chapter Sixteen:

Fish Monitoring - Wadeable Streams & Rivers

The Fisheries Bureau of the Iowa DNR has been monitoring fish for many years and has protocols for different wetland habitats. The following is an adaptation of the “Biological Sampling Procedures for Wadeable Streams and Rivers in Iowa” (Iowa DNR 2001). Few changes have been made to the original protocol.

Fish Monitoring in Wadeable Streams & Rivers

This protocol is completely based upon the “Biological Sampling Procedures for Wadeable Streams and Rivers in Iowa” (Iowa DNR 2001) protocol first drafted in 1994. In addition to recording fish species, information could also be collected on benthic macroinvertebrates. A few modifications are suggested in this section, mostly in regard to the length of area to be sampled. The design includes electrofishing to determine fish species and numbers, in addition to collecting benthic macroinvertebrates and habitat data.

Please check with a Program Manager to see if a wadeable stream is known to contain Topeka Shiners or to be Critical Habitat for this Federally Endangered species. If it is, do not sample these areas unless accompanied by appropriate Iowa DNR or U.S.F.W.S. staff. If Topeka Shiners are discovered in areas previously undocumented, immediately call the Iowa DNR Threatened and Endangered Species Coordinator.

Within the permanent sampling plot, any wadeable stream or river should be searched for all fish species using this protocol. In some of these plots a water habitat will be the focal point, meaning the hexagon will be centered on a stream, river, lake, creek, etc. In these plots, it is anticipated that a stream reach of up to 400 m or more will need to be sampled.

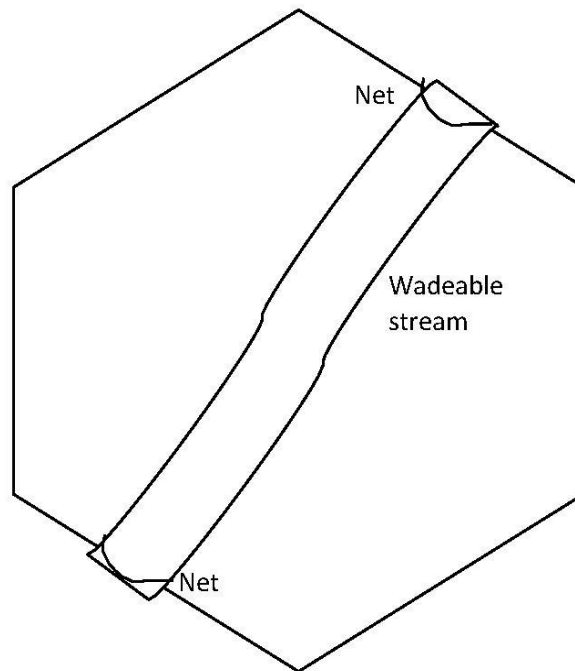
Regardless of the amount of stream occurring within the plot, a 150-m reach is the minimum that should be sampled. So, if only 50 m of suitable habitat is found within the plot, then 50 to 100 m beyond each of the 2 boundaries should be surveyed as well. Five hundred meters should be the maximum stream reach surveyed due to time considerations.

Survey Methods

Sampling in wadeable streams and rivers should occur between June 15 and September 30 (15 weeks and 2 days). In general, sampling will occur during daylight hours for active sampling gears.

Stream flow levels should be similar to base flow conditions. Sampling should be halted when the stream flow is elevated or there are high turbidity levels; when the stream flow is extremely low; or when there has been a minor runoff event within the last week. A runoff event could disrupt the aquatic community. Surveys are also halted during inclement weather (extreme wind, lightning, or rain).

The Iowa DNR wadeable streams protocol also suggests that no sampling should be done within one year after a major flood event or within one year of a severe drought. For the purposes of this monitoring program, however, community changes associated with these events also provide important information. Therefore, these two events are not considered valid reasons to disrupt the sampling regime. It should be noted on the data sheets or in the database, however, if and which of these 2 events had occurred and the date(s) of occurrence.



The Iowa DNR Fisheries Bureau wadeable streams protocol further clarifies that within each sampling reach, there should be 2 distinguishable pool/riffle sequences or 2 well defined channel bends. If neither of these is present, then there are specifications as to the length which should be surveyed. These include that waters ≤ 40 feet (12.2 m) in width should be surveyed to a length 30x the width, and waters >40 feet (12.2 m) in width should be surveyed 20x the mean width. For simplicity, this protocol advocates sampling 30x the width of the stream regardless of other considerations.

Ideally, this will result in a distance of between 300 & 400 m being surveyed.

The first step in the sampling protocol is to collect information from the GIS database as to the location of roads, trail, and other disturbances near the sampling area. Notes should also be made as to the best (apparent) location for entering the water. See Chapter 3 (Landscape Characteristics) for further information. Sampling each reach is expected to take 8 hours or less.

Sampling may only stretch over 2 days if stream conditions do not change overnight.

Data should be collected in the following sequence:

1. Measure stream width, delineate sampling reach, and place block nets (when possible).
2. Collect water samples for physicochemical water quality parameters. We don't do this
3. Collect semi-quantitative benthic macroinvertebrate samples. We don't do this
4. Collect qualitative, multi-habitat benthic macroinvertebrate sample.
5. Conduct fish sampling.
6. Complete habitat measurements.

Water Sample Collection

Water samples should be taken from the stream or river with the use of clean, glass jars that are labeled with a Sharpie marker. Water samples should be stored following recommendations outlined by the University of Iowa Hygienics Laboratory. Water samples are not currently being taken.

Benthic Macroinvertebrate Sampling

These data are qualitative and semi-quantitative, providing a list of macroinvertebrate species as well as an abundance index to the taxa observed. These techniques will not allow for the estimation of density or biomass. For the semi-quantitative data, triplicate samples should be made of either 1) rock substrates in riffle or shallow run habitat, or 2)

multi-plate, artificial substrates deployed in moderately swift run habitat. Although this benthic macroinvertebrate sampling is ideal, Iowa MSIM is not currently collecting these data.

To do this, a modified-Hess sampler, a Surber sampler, or modified Hester-Dendy (multi-plate artificial) substrates, is used, depending upon the habitat characteristics of the stream being monitored. If it is necessary to use the multi-plate artificial substrate device, this must first be deployed for 4-6 weeks to allow for colonization before data can be collected. The Iowa DNR routinely deploy these substrates during reconnaissance visits to the site or during sampling of nearby sites in order to minimize travel costs.

The modified-Hess sampler is an open-ended, mesh enclosed cylinder. Photos of this can be seen in Iowa DNR (2001). The following is copied verbatim from the Iowa DNR (2001) sampling protocol, pages 6-14: The upstream side is a mesh window that allows water to flow through the sampler while keeping all drifting macroinvertebrates out of the sampler. The downstream side of the cylinder has a funnel-shaped mesh collection bag and collection container for capturing macroinvertebrates dislodged as substrates inside the sampler are agitated. The modified-Hess sampler is most effective in shallow riffles and runs (<1.5 feet or 45.7 cm) with abundant rock substrates. This sampling device performs well in streams where there is a mixture of substrate particle sizes and the sampler can be penetrated 2-4 inches (5-10 cm) into the stream bottom.

Please refer to Appendix 4 (Macroinvertebrate Sampling) for additional information.

Benthic macroinvertebrate surveys

- Modified-Hess sampler or Surber sampler, or 4 Modified Hester-Dendy artificial substrate Samplers
- Collection jars
- Jar labels
- 10% formalin with Borax solution

Fish Community Sampling - Electrofishing

For small streams (average base-flow widths of less than 15 feet or 4.6 m) a single backpack unit is sufficient. In wider streams, it may be necessary to use 2 backpack units simultaneously.

For other streams which may be too deep or wide to cover with backpack units, a towboat electrofishing unit (with a generator, electrical control box, retractable electrodes, and a live well) is used.

Both the downstream and upstream ends of the sampling area should be blocked using 3/16" block nets. If block nets are not feasible, the use of habitat changes such as a top of a riffle can be used as an end of the segment. Beginning at the downstream starting point, a single pass is made upstream to capture all fish in the water. Sample all habitats thoroughly by methodically sweeping the anode from side to side. All stunned fish are captured in 3/16" dip nets and transferred into buckets or tanks until processed. Fish should be processed periodically slightly downstream of where your sampling segment will continue upstream once processing is complete. These additional stops to process fish increase survival of individuals in the buckets or tanks. Additionally, bubblers should be added to the buckets or tanks in order to add oxygen and help sustain capture fish until processing.

Additional data collected include the type of equipment used to stun the fish, the beginning and ending times for the use of the backpack shocker, and stream reach length and average width.

Environmental Data Collection

Environmental data collected the day of sampling should include: surface water temperature, ambient air temperature, flow level, secchi disk reading (in tenths of feet), conductivity (u/mhos), weather conditions, sampling effort (in minutes), and any relevant comments. In addition, be sure to record the number of people in the crew and their names, the name of the site, and sketch a map of the area sampled.

Equipment List

- GPS unit
- Water collection jars

Binoculars
Dip nets
Block nets
Twine for repairs to blocknets and seine nets
Backpack electrofishing units
Extra batteries for Backpack units
Battery powered bubblers for buckets
Buckets or holding tanks
Non-breathable chest waders Inflatable life preservers
Plastic calipers
Standard field kit: Clip board, pencils, weather kestrel, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets.
Field guides
Rubber gloves/electrician gloves

Staff & Training

Training is recommended and should include 1) field guide use and identification, 2) trips to University museums to discuss defining species characteristics, 3) field practice with an experienced observer, 4) safely using the sampling equipment, 5) proficiency testing, and 6) habitat data collection. Ideally, we would have 1 technician who is knowledgeable about fish identification and sampling methodology to travel between crews to assist with sampling efforts. This designated technician should review duties and safety precautions with the sampling crew before each survey.

Data Quality & Management

Electroshocking and seining data can be affected by:

- Incorrect use of equipment: Should be checked periodically by supervisor.
- Observer handling care: Fish should not be left in holding buckets any longer than necessary. Mortalities can be assessed by examining the data, and should be <1%.
- Error in species ID: Difficult to monitor, therefore, could switch observers between crews or collect voucher specimen/photographs.

At the end of each sampling day, field crews should review data sheets to ensure all information is present. At the end of the week, the field crew leader should review the data sheets for ID, escape and mortality rates, and legibility.

Data Analysis

The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of area occupied using program R. For more information, see chapter 5 (Data Analysis). This is the only protocol where sites are visited only once per year. Both of the other 2 fisheries protocols (rivers and lakes) visit each site 3 times per year.

Following the methods are outlined in the Iowa DNR (2001) protocol. The data collected allow the estimate of the following community parameters of the fish sample:

- Species composition (i.e., the number of fish of each species as a percentage of the total number of captured fish)
- Fish species relative abundance (i.e., catch per unit effort)
- Proportion of fish with external abnormalities.
- The methods employed do not provide quantitative information suitable for fish population density or biomass estimates.

Safety Issues & Considerations

As with all other protocols, basic hygiene, including washing hands prior to eating or face touching should be followed by all personnel.

Electrofishing can be dangerous. All personnel need to be trained in the use of this equipment. Working in wadeable streams is also physically challenging. Working in aquatic situations can be dangerous. Technicians should be cautious of slippery substrates and be aware of the speed of the river flow. Sampling should be suspended during inclement weather, including heavy rain or lightning storms. If a person is swept off their feet when wearing chest waders, it is possible that the air trapped in the bottom of the waders will force the person to travel down the channel upside down with their head below water. Therefore, it is recommended that chest waders have release snaps in the front of the bib to allow the technician to escape in that situation. It would also be advisable to wear an inflatable life jacket underneath the bib of the chest waders.

Care should be taken in order to lessen the probability of spreading an infectious agent, such as a fungus or virus, between wetlands. One way to reduce the chance of spreading an infectious agent between wetlands is to allow the waders and equipment to dry for 3-4 days between sites. This may be impractical given the short time frame available for aquatic surveying in Iowa. It may be best to rinse the waders, gloves, and other equipment with a solution of water and bleach. Spray bottles are provided for this purpose.

Target Species

The following list of fish species represents the 80 species of greatest conservation need listed in the 3rd edition (2015 revision) for the Iowa Wildlife Action Plan (Reeder and Clymer. 2015) and may be encountered during a survey. Distribution maps for these species can be found in "Iowa Fish & Fishing" (Harlan et al. 1987) Appendix 1 contains a list of all fish species known to occur in Iowa which may also be encountered during the monitoring efforts. An astrisks (*) indicated fish designated as Data Deficient.

Target species:

Common Name	Scientific Name	Habitat
Chestnut lamprey	<i>Ichthyomyzon castaneus</i>	Mississippi and Chariton rivers
Northern brook lamprey	<i>Ichthyomyzon fossor</i>	Northeast 1/4
Silver lamprey	<i>Ichthyomyzon unicuspis</i>	Mississippi River
American brook lamprey	<i>Lampetra appendix</i>	Northeast 1/4
Lake sturgeon	<i>Acipenser fulvescens</i>	Mississippi River
Pallid sturgeon	<i>Scaphirhynchus albus</i>	Missouri River
Shovelnose sturgeon	<i>Scaphirhynchus platyrhynchus</i>	Mississippi and Missouri Rivers
Paddlefish	<i>Polyodon spathula</i>	Mississippi, Missouri, Des Moines, Iowa, Cedar, and Skunk rivers
American eel	<i>Anguilla rostrata</i>	Mississippi and Missouri Rivers & larger tributaries
Skipjack herring	<i>Alosa chrysochloris</i>	Mississippi and Missouri Rivers
Largescale stoneroller	<i>Campostoma oligolepsis</i>	NE 2/3
Gravel chub	<i>Erimystax x-punctatus</i>	Large rivers and streams
Western silvery minnow	<i>Hybognathus agryritis</i>	Missouri drainage
Mississippi silvery minnow	<i>Hybognathus nuchalis</i>	Mississippi drainage
Plains minnow	<i>Hybognathus placitus</i>	Missouri drainage
Pallid shiner	<i>Hybopsis amnis</i>	Upper Mississippi River
Redfin shiner	<i>Lythurus umbratilis</i>	Northeast 1/4
Shoal chub	<i>Macrhybopsis hyostomus</i>	Large, interior rivers, statewide
Sturgeon chub	<i>Macrhybopsis gelida</i>	Eastern ½ Missouri River
Sicklefin chub	<i>Macrhybopsis meeki</i>	Missouri River
Pearl dace	<i>Margariscus margarita</i>	Worth county

Common Name	Scientific Name	Habitat
Golden shiner	<i>Notemigonus crysoleucas</i>	
Pugnose shiner	<i>Notropis anogenus</i>	West Lake Okojobi
River shiner	<i>Notropis blennius</i>	
Silverband shiner	<i>Notropis shumardii</i>	Mississippi & Missouri(?) River
Ghost shiner	<i>Notropis buchanani</i>	Mississippi River
Blacknose shiner	<i>Notropis heterolepis</i>	NW
Ozark minnow	<i>Notropis nubilus</i>	NE ¼
Carmine shiner	<i>Notropis percobromus</i>	NE ¼
Weed shiner	<i>Notropis texanus</i>	Cedar & Mississippi Rivers
Topeka shiner	<i>Notropis topeka</i>	W ¾
Mimic shiner*	<i>Notropis volucellus</i>	
Channel shiner*	<i>Notropis wickliffi</i>	Iowa River
Pugnose minnow	<i>Opsopoeodus emiliae</i>	Mississippi River
Suckermouth minnow	<i>Phenacobius mirabilis</i>	Upper Des Moines basin
Southern redbelly dace	<i>Phoxinus erythrogaster</i>	NE 1/3, NW ¼
Flathead chub	<i>Platygobio gracillis</i>	Missouri drainage
Longnose dace	<i>Rhinichthys cataractae</i>	NE corner
Blue sucker	<i>Cycleptus elongates</i>	Mississippi and Missouri Rivers & larger tributaries
Lake chubsucker*	<i>Erimyzon succetta</i>	
Black buffalo	<i>Ictiobus niger</i>	Mississippi River & large tributaries
Spotted sucker	<i>Minytrema melanops</i>	Mississippi River
Silver redhorse	<i>Moxostoma anisurum</i>	
River redhorse	<i>Moxostoma carinatum</i>	Upper pools of Mississippi
Black redhorse	<i>Moxostoma duquesnei</i>	Turkey & upper Iowa river drainages
Brown bullhead	<i>Ameiurus nebulosus</i>	N 1/3
Blue catfish	<i>Ictalurus furcatus</i>	Lower Mississippi & Missouri Rivers
Slender madtom	<i>Noturus exilis</i>	Mississippi River tributaries
Tadpole madtom	<i>Noturus gyrinus</i>	Statewide
Freckled madtom	<i>Noturus gyrinus</i>	Mississippi River & large tributaries
Grass (Redfin) pickerel	<i>Esox americanus</i>	Missouri River & tributaries
Northern pike	<i>Esox lucius</i>	
Central mudminnow	<i>Umbra limi</i>	N 1/3
Brook trout	<i>Salvelinus fontinalis</i>	NE corner
Trout perch	<i>Percopsis omiscomycus</i>	NW ¼; Upper Mississippi River, Grand & Chariton Rivers
Pirate perch	<i>Aphredoderus sayanus</i>	Mississippi River & large tributaries
Burbot	<i>Lota lota</i>	Missouri River, Mississippi River & tributaries
Brook silverside	<i>Labidesthes sicculus</i>	East
Banded killifish	<i>Fundulus diaphanous</i>	Natural lakes in NW; Missouri River
Starhead topminnow	<i>Fundulus dispar</i>	L. Sioux, Iowa Rivers
Blackstripe topminnow	<i>Fundulus notatus</i>	E 1/3
Plains topminnow	<i>Fundulus sciadicus</i>	Rock River Basin
Mottled sculpin	<i>Cottus bairdi</i>	Lower Bear Creek

Common Name	Scientific Name	Habitat
Slimy sculpin	<i>Cottus cognatus</i>	NE corner
Rock bass	<i>Ambloplites rupestris</i>	
Longear sunfish [†]	<i>Lepomis megalotis</i>	
Northern sunfish [†]	<i>Lepomis peltastes</i>	
Western sand darter	<i>Annicrypta clara</i>	Mississippi River
Crystal darter	<i>Crystallaria asprella</i>	Mississippi & Turkey Rivers
Mud darter	<i>Etheostoma asprigene</i>	Mississippi River & tributaries
Rainbow darter	<i>Etheostoma caeruleum</i>	Cedar River basin
Bluntnose darter*	<i>Etheostoma chlorosomum</i>	Mississippi River
Iowa darter	<i>Etheostoma exile</i>	Northern ½
Least darter	<i>Etheostoma microperca</i>	Maquoketa, tributary to Otter Creek
Orangethroat darter*	<i>Etheostoma spectabile</i>	SE ¼
Orangethroat darter*	<i>Etheostoma spectabile</i>	SE ¼
Banded darter	<i>Etheostoma zonale</i>	NE ¼
Northern logperch	<i>Percina caprodes</i>	Mississippi drainage, Clear Lake
Blackside darter	<i>Percina maculate</i>	Mississippi River
Slenderhead darter	<i>Percina phoxocephala</i>	Mississippi drainage
River darter	<i>Percina shumardi</i>	Mississippi River

[†]Until the publication of Page et al. (2013), the Northern Sunfish was called the Longear Sunfish (*L. megalotis*). But, in Page et al. (2013), the name of the form of the Longear Sunfish known to occur in Iowa was changed to Northern Sunfish (*L. peltastes*). In 2014, sunfish were documented at the Fairport Hatchery and these were preliminarily determined to be *L. megalotis*. If this identification stands with genetic analyses, *L. megalotis* would be a new species to Iowa.

Additional Methods for Special Locations

Minnow traps

Minnow traps may be an effective way to find additional fish. These are used as part of the Amphibian protocol for capturing tadpoles. Minnow traps should be deployed in water at least deep enough to cover the trap opening but with an empty plastic bottle or other floatation device to ensure part of the trap stays above water to allow non-gilled captures to breathe. Traps should be checked daily and left in the water for 3 to 5 days. For more information on this protocol see Chapter 10 (Amphibian & Reptile Monitoring).

Seining

Seining may be the most efficient method to sample small fishes (e.g. redbfin shiner *Lythrurus umbratilis*). However, recent research in northwest Iowa appears to indicate that seining does not add additional information when electrofishing is also used (Clay Pierce, personal communication). The seine should be of 3/16 inch mesh size, and have floats attached at the top and weights attached at the bottom. For most wadeable streams and rivers in Iowa a haul or bag seine should be sufficient. If not performed correctly, fish could escape from under the net. If available, the same equipment could be used in wadeable streams as in the larger systems, but in the wadeable streams, the trawling net would be drawn through the water by hand (Herzog et al. 2005). The mesh size on the inner trawling net used in the larger systems is also 3/16 inch (4.76-mm).

Two technicians should pull the seine from a downstream to upstream direction, taking care that the net stays on the bottom of the channel bed. The seine should be removed from the water every 50 m. Fish should be removed from the net and can be processed by another technician as the seine technicians continue upstream, or they can be placed in a holding bucket until processing.

The entire reach should be sampled with the electroshock technique moving from downstream to the upstream blocking net. This same area should also be sampled with between 1 and 3 seine hauls (Quist et al. 2003).

Make sure the fish in the holding buckets or tanks have fresh water to limit mortality. At pre-determined stopping points (which can be blocked by additional nets prior to beginning the sampling), identify and count the fish. If fish are to be marked at that property, mark the fish and record the mark. Release all fish.

Collect information on all captured fish, regardless of size (i.e. those less than 1 inch in size should also be identified if possible, and counted). In addition, examine all collected fish for external abnormalities [skeletal deformities, eroding fins, lesions, and tumors (DELTs)]. Record this information on the data sheet. The DELT coding procedures have been adapted from the Ohio EPA fish sampling procedures (OEPA 1989). These guidelines are listed in the appendix 4.

For any un-identifiable species, a voucher may be collected by preserving 1 or more specimen in 10% formalin.

Chapter Seventeen:

Fish Monitoring - Lakes

The Fisheries Bureau of the Iowa DNR has been monitoring fish for many years and has protocols for different wetland habitats. The following is an adaptation of the “Statewide Biological Sampling Plan” which was co-written by J Larscheid and L Mitzner with input from M Conover, D Bonneau, K Hill, J Hudson, S Grummer, M Flammang, J Wahl, L Miller, M McGhee, S Waters, and D McWilliams. This protocol has never been implemented for Iowa MSIM and may not reflect current Fisheries Bureau methodology standards.

Fish Monitoring in Lakes

Within the permanent sampling plot, any non-wadeable pond or lake should be searched for all fish species using this protocol. In some of these plots a water habitat will be the focal point. In these plots, it is anticipated that a large water body will need to be sampled. In other plots, it may be that only a small water body will need to be surveyed. Regardless of primary habitat classification, some wetlands on the property may need to be surveyed using this protocol depending on the size and type of the wetland. For example, a large lake within 500 m of the center point chosen using the protocol in chapter 3 (Landscape Characteristics) in the forested habitat class would still be surveyed using this protocol. Water bodies that are shallow enough to be surveyed using a back-pack shocker should be examined following the protocol in Chapter 15 (Fish Monitoring in Wade-able Streams). The protocol described in the current chapter is for deeper water bodies.

Survey Methods

Sampling in lakes and deeper ponds will occur between September and October to allow for cooler surface waters so fish are more likely to be found using the techniques. In general, sampling will occur between 8 am and 5 pm. By electroshocking only during these hours, surveys will be standardized to allow comparisons on a capture-per-unit-effort basis. Trends as to fish abundance are usually evaluated based upon the number of fish sampled per minute of actual shocking time. Lake and pond water bodies should be visited 3 times during these 2 months.

Electrofishing

DC electrofishing boats will be the primary sampling tool on ponds and lakes. Each DC shocking boat will have 16, ½ inch droppers. Dropper exposure will be based on the measured conductivity (umhos) such that increasing conductivity will result in decreasing dropper exposure as outlined in Reynolds (1996). Electrofishing is most effective in shallow water and selects species associated with shoreline or shallow water habitats. Once sampling locations have been chosen, they should be georeferenced and diagramed on a map to ensure that future sampling occurs in the same area. Within the water body, areas should be chosen (and mapped for future data collection) for searches that contain a variety of structure and habitat types. Bays, points, stumps, aquatic vegetation, and the faces of dams work well for black bass, bluegill, crappie, and several other species depending on the lake being surveyed.

The total amount of time spent shocking (meaning the amount of time that electricity is sent into the water, not including times when the current is stopped), will vary with the size of the water body as follows:

Lake or pond size	Effort in minutes
<100 acres (40.5 ha)	30-90
100-500 acres (40.5-202 ha)	60-120
>500 acres (202 ha)	>90

Shocking runs are to be conducted in 15-minute segments (Pearson 1993, NYSBF 1989) and divided into at least 3 runs of similar distances so that variability can be calculated. This will allow a minimum of 3 runs using 45 minutes per lake. The track taken should be recorded using a GPS unit.

Dip-nets with a small mesh size (3/16 inch (4.76 mm) or smaller) should be used.

Trawling

Recent work from Missouri has indicated that a trawling device will be effective for catching small bodied fish in a variety of habitats (Herzog et al. 2005). This method entails using a modified two-seam balloon trawl, also called a Missouri trawl. As of October 2006, Missouri Department of Conservation staff (who designed the system) was advocating [Innovative Net Systems](#) for the supplier of the trawl (David Herzog, personal communication). The company has several designs, but MDC recommends either the Missouri trawl or the Armadillo-Herzog (AH) trawl. The primary difference in the 2 trawls appears to be that the AH trawl is made of more durable materials (and is therefore more expensive).

The trawls should be pulled through the water moving downstream. The trawl should just barely move faster than the current. It can be pulled by 2 people in shallow water or by a boat. If pulled by a boat, it should be attached to the front of the boat and the boat should move backwards downstream at a speed slightly greater than that of the current. Be sure to GPS the locations of each haul's start and stop (or, alternatively to record the track taken as the boat moves. Each haul should take between 3 and 5 minutes before the net is pulled aboard and emptied into the holding buckets. These data will be quantified by time as in fish captured per unit of time.

Fish Handling

All fish captured with either of the above methods will be placed into holding tanks or buckets. Make sure the fish in the holding buckets or tanks have fresh water and an air bubbler to limit mortality. These data should be collected (and identified as such on the data sheet) for each electrofishing run and net-haul. At pre-determined stopping points, identify and count the fish. If fish are to be marked at that site, mark the fish and record the mark. Release all fish.

Collect information on captured fish, regardless of size (i.e. those less than 1 inch in size should also be identified and counted). In addition, examine all collected fish for external abnormalities [skeletal deformities, eroding fins, lesions, and tumors (DELTs)]. Record this information on the data sheet. The DELT coding procedures have been adapted from the Ohio EPA fish sampling procedures (OEPA 1989). These guidelines are listed in the Appendix at the end of this protocol. A minimum of 50 fish should be measured for each species captured.

Lengths should be measured to the nearest 1 mm. The rest of the captured fish must be counted to obtain valid catch per unit effort information in the data set. These counts should be grouped by length class. Ideally the first 10 fish in each length class will be measured for exact length with the remaining grouped together (e.g., 53, 54, 51, 52, 55, 53, 53, 57, 59, 53 and 120 additional fish in the 50-60 mm group).

For any un-identifiable species, a voucher may be collected by preserving specimen in 10% formalin.

Environmental Data Collection

Environmental data collected the day of sampling should include: surface water temperature, secchi disk reading (in tenths of feet), conductivity (uhmos), weather conditions, sampling effort (in minutes), and any relevant comments. In addition, be sure to record the number of people in the crew and their names, the name of the site, and sketch a map of the area sampled.

Equipment List

- Water collection jars
- Dip nets
- Chest waders
- Inflatable life preservers
- Plastic calipers
- Standard field kit: Clip board, pencils, kestrel weather station, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets.
- Field guides
- Rubber gloves
- DC Electroshocking boat
- Trawling equipment

Fish voucher collection materials (jars, formalin, etc)
Spray bottle with bleach solution to decontaminate equipment

Staff & Training

Training (beginning on August 15) is recommended and should include 1) field guide use and identification, 2) trips to University museums to discuss defining species characteristics, 3) field practice with an experienced observer, 4) safely using the sampling equipment, 5) proficiency testing, and 6) habitat data collection. The crew leader should review duties and safety precautions with the sampling crew before each survey.

Data Quality & Management

Electroshocking and trawling data can be affected by:

- Incorrect use of equipment: Should be checked periodically by supervisor.
- Observer handling care: Fish should not be left in holding buckets any longer than necessary. Mortalities can be monitored through data, and should be <1%.
- Error in species ID: Difficult to monitor, therefore, could switch observers between crews or collect voucher specimen.

At the end of each trapping day, field crews should review data sheets to ensure all information present. At the end of the week, the field crew leader should review the data sheets for identification, escape and mortality rates, and legibility. Be sure to keep data collected by different methods separate. Also be sure to keep the locations of the data collection labeled.

Data Analysis

The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of area occupied using program R. For more information, see chapter 5 (Data Analysis).

The data collected should allow the estimate of the following community parameters of the fish sample:

1. Species composition
2. Species relative abundance (i.e., the number of fish of each species as a percentage of the total number of captured fish)
3. Fish abundance (i.e., catch per unit effort)
4. Proportion of fish with external abnormalities.

The methods employed do not provide quantitative information suitable for fish population or biomass estimates.

Safety Issues & Considerations

As with all other protocols, basic hygiene, including washing hands prior to eating or face touching should be followed by all personnel.

Electro-fishing can be dangerous. All personnel need to be trained in the use of this equipment. Working in aquatic situations can be dangerous. Technicians should be cautious of slippery substrates and be aware of the speed of water flow. Sampling should be suspended during inclement weather, including heavy rain or lightning storms. If a person is swept into the water when wearing chest waders, it is possible that the air trapped in the bottom of the waders will force the person to travel downstream with their head below water. Therefore, it is recommended that chest waders have release snaps in the front of the bib to allow the technician to escape in that situation. It would also be advisable to wear an inflatable life jacket underneath the bib of the chest waders.

Care should be taken in order to lessen the probability of spreading an infectious agent, such as a fungus or virus, between wetlands. One way to reduce the chance of spreading an infectious agent between wetlands is to allow the waders to dry for 3-4 days between sites. This may be impractical given the short time frame available for fish surveying in Iowa. As an alternative, it may be best to rinse the waders and equipment with a solution of hot water and bleach. Spray bottles are provided for this decontamination step.

Target Species

The list of fish species represents is in Chapter 15 (Fish Monitoring in Wadeable Streams).

Appendix 1 contains a list of additional, more common, species which may also be encountered during the monitoring efforts.

Additional Methods for Special Locations

Fyke Nets

Fyke nets are passive gear that sample fish by entrapment. Fyke nets tend to be selective for cover seeking, mobile species (Neilson and Johnson 1983, McWilliams et al. 1974). Nets used in this procedure should be standardized by size to ensure continuity across areas. All sampling will be conducted using 2 ft x 4 ft (60.96 cm x 121.92 cm) frames with 7 hoops of 2 ft (60.92 cm) diameters enclosed with ¾ inch (1.91 cm) bar mesh netting for larger fish or 3/16 inch (4.79 mm) mesh for smaller fish.

Fyke nets are typically deployed in shoreline habitats where the water is about 4 feet (1.22 m) deep at the frame. Sampling sites should be geo-referenced and mapped to ensure the same areas are sampled through time.

The number of nets set should vary with the size of the water body as follows:

Waterbody size	Effort (nets/night)
<100 acres (40.5 ha)	3-15
100-500 acres (40.5-202 ha)	5-20
>500 acres (202 ha)	7-28

Typically, nets are set for just one night, meaning that up to 28 net sets may be needed per wetland. Fyke nets are set overnight and emptied each day. The time of setting and raising should be recorded.

Age Growth

In some situations it may be necessary to collect tissue for age-growth calculations. Most likely, this will be rare for the MSIM program and will only be done at the request of a scientist willing to do the lab work and analysis. All aging structures/tissues collected should be placed into scale envelopes on which the following information has been recorded: site name, sampling gear used, date of sampling, species, length, weight, and any comments. At the end of each day the scale envelopes should be spread out and allowed to dry completely. This is especially important for spines which can go rancid quickly if not allowed to dry.

Spring Sampling

It should be left to the biologist's discretion to decide if supplemental sampling for fish should be conducted in the spring for certain water bodies.

Minnow Traps

Minnow traps may be an effective way to find additional fish. They are used as part of the amphibian protocol for capturing tadpoles. Minnow traps should be deployed in water at least deep enough to cover the trap opening but with an empty plastic bottle or other floatation device to ensure part of the trap stays above water to allow non-gilled captures to breathe. Traps should be checked daily and left in the water for 3 to 5 days. Fish captured in these (& other Aquatic traps used in the Amphibian and Reptile protocol) should be recorded as 'incidentals' in the MSIM database.

Chapter Eighteen

Fish Monitoring - Large Rivers

The Fisheries Bureau of the Iowa DNR has been monitoring fish for many years and has protocols for different wetland habitats. The following is an adaptation of both the USGS Long Term Resource Monitoring Program of Pool 13 of the Mississippi River, following the Long-Term Resource Monitoring Procedures: Fish Monitoring protocol (LTRMP, Gutreuter et al. 1995), and the Great River Ecosystems Field Operations Manual, Environmental Monitoring and Assessment Program (EMAP, Angradi et al. DRAFT 2005). The reader should refer to both of the above documents for more in depth information. This protocol has never been implemented for Iowa MSIM and may not reflect current Fisheries Bureau methodology standards.

Fish Monitoring in Large Rivers

Within all permanent sampling plots, all non-wadeable rivers should be searched for all fish species using this protocol. In some of these plots the river will be the primary habitat classification and this will be the primary protocol followed. This protocol is based upon the "LTRMP" (Gutreuter et al. 1995) protocol and the "EMAP" (Angradi et al. 2005) protocol. In addition to recording fish species, information is also collected on benthic invertebrates and habitat variables. A few modifications are suggested in this section, mostly in regard to the area to be sampled. The design includes electro-shocking to determine fish species and numbers in addition to collecting benthic invertebrates and habitat data. Water bodies that are shallow enough to be searched using a backpack shocker should be examined following the protocol described in Chapter 15 (Fish Monitoring in Wadeable Streams). The protocol described in this chapter is for deeper water habitats.

Please check with a Program Manager to see if a river is known to contain Topeka Shiners or to be Critical Habitat for this Federally Endangered species. If it is, do not sample these areas unless accompanied by appropriate Iowa DNR or U.S.F.W.S. staff. If Topeka Shiners are discovered in areas previously undocumented, immediately call the Iowa DNR Threatened and Endangered Species Coordinator. One individual may be collected from such sites for the purpose of vouchering this species with the prior approval of the Iowa DNR T&E Coordinator.

Survey Methods

Sampling in large rivers should occur between July 1 and September 30 (13 weeks and 1 day) (Angradi et al. 2005). Ideally, three visits per site would follow Gutreuter et al. (1995) with 3 visits, one each during: June 15-July 30, August 1-September 15, and September 16-October 30. Following the LRTMP protocol for timing, Gutreuter et al. (1995) will allow for a longer sampling time and perhaps, a more even sampling effort. This time-frame will allow for the fish to be relatively active, feasible weather conditions, and stable water flow. In general, sampling will occur between 8 am and 5 pm.

If Secchi depth is <15 cm, then sampling should probably be halted although this is left to the discretion of the crew leader. Surveys are also halted during inclement weather (extreme wind, lightning, or rain). Electrofishing should be conducted first in order to avoid disturbing the fish from their habitats with the other data collection.

Prior to implementing this protocol, collect information from the GIS data base as to the location of roads, trail, and other disturbances near the sampling area (see Chapter 3, Landscape Characteristics). Notes should also be made as to the best (apparent) location for entering the water. GPS coordinates should be loaded into the GPS unit to facilitate finding the correct locations in the river to begin each sampling run. Sampling within each area is expected to take 8 hours or less.

Data should be collected in the following sequence:

1. Conduct fish sampling.
2. Collect water samples for physicochemical water quality parameters.
3. Measure water temperature, velocity, water depth, Secchi transparency, conductivity.
4. Collect semi-quantitative benthic macroinvertebrate samples.
5. Collect qualitative, multi-habitat benthic macroinvertebrate sample.

6. Complete habitat measurements.

Fish Community Sampling Electroshocking

As a minimum, a 500 m run typically takes 30 minutes (excluding processing the fish), therefore, it is expected that completing the fish sampling will take at least over 90 minutes of time simply for the electroshocking and ignoring the fish identification and data recording. This will vary depending on habitat cover.

A standard electrofishing boat should be sufficient for the sampling. See Nielsen and Johnson (1983) for more information. Electrofishing may begin as early as 1 hour after sunrise (Gutreuter et al. 1995). The transect runs should be roughly mapped out in advance with advice from an Iowa DNR fisheries bureau biologist. Record GPS locations for at least each start and end point of each run or record the path using GPS. Fish should be processed after each run and the data should be labeled accordingly by run number. "The path of the boat should be analogous to the motion of a person using a metal detector: a side-to-side path with complete lateral coverage and a slow forward pace" (Angardi et al. 2005). Be sure to thoroughly traverse areas of snags, piers, and other cover.

All stunned fish are captured in 1/8" or 3/16" mesh landing nets and transferred into buckets or tanks filled with water until processed. The holding tank should be at least 300 L in volume. An aerator should be used to maintain oxygen in the tank. Fish should be processed immediately following each run (see fish handling below). If fish are processed during the run, e.g. due to excessive stress, then these individuals should be released behind the boat into deeper water to ensure they are not recaptured.

Additional data collected include the type of equipment used to stun the fish, the beginning and ending times for the use of the electro-shocker, and stream reach length and average width.

Trawling

In addition to electrofishing, seining or trawling will be used to collect additional data with each visit. Recent work from Missouri has indicated that a trawling device will be effective for collecting small bodied fish in a variety of habitats (Herzog et al. 2005). This method entails using a modified two-seam balloon trawl, also called a Missouri (or Armadillo) trawl. As of October 2006, Missouri Department of Conservation staff (who designed the system) was advocating [Innovative Net Systems](#) for the supplier of the trawl (David Herzog, personal communication). The company has several designs available, but MDC recommends either the Missouri trawl or the Armadillo-Herzog (AH) trawl. The primary difference between the 2 appears to be the durability of materials in the more expensive AH trawl.

A trawl net can be used on the same runs as electroshocking to collect additional fish.

Alternatively, additional areas can be chosen to use the trawling system. The traditional trawl net should be placed off the back of the boat (see Nielsen and Johnson 1983 for additional information). The trawl should move in a downstream direction (Gutreuter 1995). The trawl should just barely move faster than the current. The Missouri trawl can be pulled by 2 people in shallow water as a seine or by a boat. The Missouri trawl net would be placed off the front of the boat and the boat would be moved backwards going downstream at a speed slightly greater than that of the current. Be sure to GPS the location of each haul's start and stop or record the track taken as the boat moves. Each haul should last for 3 to 5 minutes before the net is pulled aboard and emptied into the holding tanks. These data will be quantified by time, as in fish per unit time.

Fish Handling

Collect information on all captured fish, regardless of size (i.e. those less than 1 inch in size should also be identified if possible, and counted) or method of capture. Make sure fish in holding tanks have fresh water to limit mortality. These data should be collected (and identified as such on each data sheet) for each of the methods used. At pre-determined stopping points, identify and count the fish. Measure and mark the fish if applicable. Then release the fish at areas where they are unlikely to be resampled.

In addition, examine all collected fish for external abnormalities (skeletal deformities, eroding fins, lesions, and tumors (DELTS)). Record this information on the data sheet. The DELT coding procedures have been adapted from the Ohio EPA

fish sampling procedures (OEPA 1989). These guidelines are listed in Appendix 4. A minimum of 50 fish should be measured for each species captured. Lengths should be measured to the nearest 1 mm. The rest of the captured fish should be counted to obtain valid catch per unit effort information.

Some samples will be preserved for vouchers or later identification. Fish chosen for preservation should be placed into 10% formalin solution or 70% ethanol solution.

Equipment List

Water collection jars
Dip nets
Electroshocking boat and associated equipment
Trawling equipment
Inflatable life preserver
Plastic calipers or measuring board
Standard field kit: Clip board, kestrel weather station, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets.
Field guides
Rubber gloves, fish-handling gloves
Holding tank
GPS Unit
Clear tape to cover labels
State and federal permits
Coolers with ice sealed in bags
Voucher jars with formalin
Benthic macroinvertebrate surveys: Modified-Hess sampler or Surber sampler, or 4 Modified Hester-Dendy artificial substrate
Samplers
Collection jars
Jar labels
10% formalin with Borax solution or 70% Enthanol

Staff & Training

Training should include 1) field guide use and identification, 2) trips to university museums to discuss defining species characteristics, 3) field practice with an experienced observer, 4) safety using the sampling equipment, 5) habitat data collection, 6) boat training, and 7) proficiency testing. The crew leader should review duties and safety precautions with the sampling crew before each survey.

Data Quality & Management

Electroshocking and seining data can be affected by:

- Incorrect use of equipment: Should be checked periodically by supervisor.
- Observer handling care: Fish should not be left in holding buckets any longer than necessary. Mortalities can be monitored through data, and should be <1%.
- Error in species ID: Difficult to monitor, therefore, could switch observers between crews or collect voucher specimens.

At the end of each trapping day, field crews should review data sheets to ensure all information present. At the end of the week, the field crew leader should review the data sheets for ID, escape and mortality rates, and legibility. Be sure to keep data collected by different methods separate. Also be sure to label each data sheet with the location of the surveys conducted.

Data Analysis

The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of areas occupied and detection probabilities using program PRESENCE or program MARK. For

additional information on the PAO techniques, see Chapter 5 (Data Analysis).

The data collected allow the estimate of the following community parameters of the fish sample:

1. Species composition
2. Species relative abundance (i.e., the number of fish of each species as a percentage of the total number of captured fish)
3. Fish abundance (i.e., catch per unit effort)
4. Proportion of fish with external abnormalities.

The methods employed do not provide quantitative information suitable for fish population or biomass estimates.

Safety Issues & Considerations

As with all other protocols, basic hygiene, including washing hands prior to eating or face touching should be followed by all personnel.

Electro-fishing can be dangerous. All personnel need to be trained in the use of this equipment. Working in rivers is also challenging and crews should have safe-boating training. Working in aquatic situations can be dangerous. Technicians should be cautious of slippery substrates and be aware of the speed of the river flow. Sampling should be suspended during inclement weather, including heavy rain or lightning storms. All crew members should wear an inflatable life jacket underneath the bib of the chest waders.

Each boat should have a personal floatation device for each person on board, a cell phone, tools, first aid kit, engine oil, sunblock, insect repellent, and a tow line.

Care should be taken in order to lessen the probability of spreading an infectious agent, such as a fungus or virus, between wetlands. One way to reduce the chance of spreading an infectious agent between wetlands is to allow the equipment to dry for 3-4 days between sites. This may be impractical given the short time frame available for aquatic surveying in Iowa. As an alternative, it may be best to rinse all equipment with a solution of water and bleach. Spray bottles are provided for this purpose.

Target Species

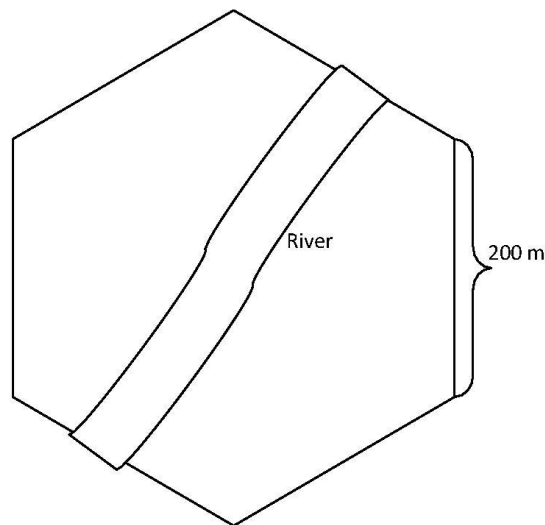
The list of fish species represents is in Chapter 15 (Fish Monitoring in Wadeable Streams).

Appendix 1 contains a list of additional, more common, species which may also be encountered during the monitoring efforts.

Chapter Nineteen: Mussel Monitoring Protocol

Mussel Monitoring

Mussels are dependent upon host fish for dispersal and therefore areas to be searched for mussels will be restricted to those to which fish have access, as documented by the appropriate fish survey which will be conducted on each permanent sampling plot at a time earlier in the year to the timeframe recommended for mussel surveys. However, as it is possible that fish may pass through some wetlands without inhabiting them, fish presence is not a requirement for mussel surveys.

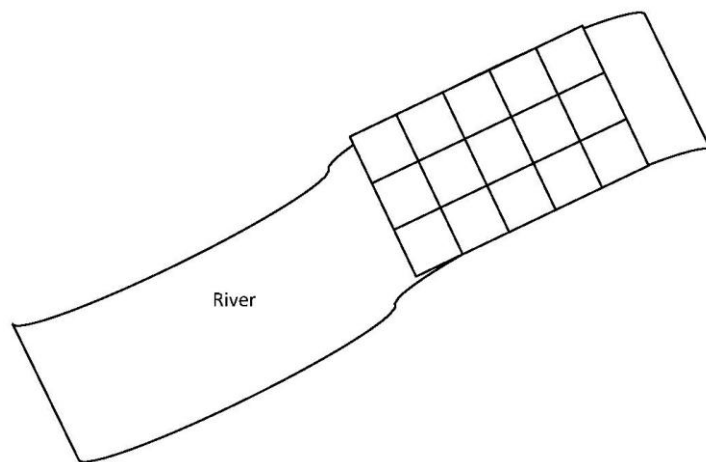


Survey Methods

Surveys are to be conducted between the beginning of August and the end of September (8 weeks and 5 days). This time frame will allow high flow and cold temperature waters to be avoided.

A sampling grid should be established using the side of the channel as one axis and a line perpendicular to the stream bank crossing the channel as the other axis. This grid should be established in both deep waters requiring scuba diving (>3 feet deep) and wadeable waters <3 feet deep. Established quadrats will be 1 m². Establishing this grid may be done with measuring equipment in the field or by using current spatial data and GIS software. When using measuring equipment to establish the grid in the field, it may be best to establish this grid by extending a surveyors tape or a line marked in 1-m increments along the stream side as a reference guide. Marking 1-m increments with marking poles or surveyor's flags establishes one axis of the grid, and the marking tape or marked line can then be stretched across the water body perpendicular to the channel to give a reference for the specific location of each quadrat. The line can be held by two surveyors (one on each bank) at the marked locations or held across with poles (e.g. rebar) driven into the bank. GPS the location of the starting end of the grid, along with the starting points of the longitudinal transects.

Due to the size or characteristics (e.g.-steep banks) of some water bodies it may be more efficient to establish the grid using current spatial data on the extent of water in the water body and GIS software. Information on the extent of the water body can be digitized from current aerial imagery or collected with a GPS by a technician in the days leading up to a planned survey. A point grid with 1 m spacing can then be placed within the outlined area, and then the start points and subsequent quadrats to be searched can be assigned based on this point grid. Handheld GPS units are used to locate these points during the survey, thus horizontal accuracy of the GPS is important and the most accurate GPS available should be used.



Example grid. Grid should actually be stretched the length of the channel within the hexagonal grid.

A total of 100 m of channel length should be searched. This 100 m can be divided into two sections of 50 m each. The location of these 2 sections of the same water body within (or nearby) the permanent hexagonal sampling plot should be placed such that as diverse of habitats as possible are surveyed. Once the starting points have been established, each technician should spend 15-20 minutes randomly searching the entire area for as many species as possible. This search should include noting individuals/shells on the shore, as well as visual or tactile searches in the water. If there are fewer than 3 technicians in the water, then increase the amount of time each spends randomly searching so that at least 1 hour of total time is spent in the random search. One person should remain on shore to record the species as they are called out. If the area being surveyed for the random timed search will also be part of quantitative sampling any mussels found should be placed back where they were found so as not to bias the quantitative surveys.

Once the random timed search has been completed, begin at the furthest downstream location for the quadrat sampling. Depending on stream width, 50 to 80 quadrats should be sampled at each location following a systematic design. The following formula is adapted from Strayer and Smith (2003) and can be used to determine the number of quadrats that should be 'skipped' between those that are searched:

$$q = \frac{L \times W}{n/k}$$

Where q is the number between quadrats, L is the length of the area to be searched, W is the width of the area to be searched, n is the number of quadrats to be searched, and k is the number of starts (e.g. the number of technicians searching). So, for example, if a river is 10 m wide, 50 m in length, and 25 (50 quadrats divided by 2 stream sections) quadrats should be searched by 3 technicians, then the spacing between quadrats should be equal to 60 quadrats. So, if 3 technicians (**X**, **Y**, and **Z**) randomly choose 4, 12, and 25 as starting locations, then the following table would illustrate the quadrats each would search given the above length and width measurements.

↓ Riverwidth					→ Riverlength										
1	11	21	31	41	51	61	71	81	91	101	111	121	131	141	151
2	Y	22	32	42	52	62	Y	82	92	102	112	122	Y	142	152
3	13	23	33	43	53	63	73	83	93	103	113	123	133	143	153
X	14	24	34	44	54	X	74	84	94	104	114	X	134	144	154
5	15	Z	35	45	55	65	75	Z	95	105	115	125	135	Z	155
6	16	26	36	46	56	66	76	86	96	106	116	126	136	146	156
7	17	27	37	47	57	67	77	86	97	107	117	127	137	147	157
8	18	28	38	48	58	68	78	88	98	108	118	128	138	148	158
9	19	29	39	49	59	69	79	89	99	109	119	129	139	149	159
10	20	30	40	50	60	70	80	90	100	110	120	130	140	150	160

↓ Riverwidth					→ Riverlength									
161	171	181	191	201	211	221	231	241	251	261	271	281	291	301
162	172	182	192	202	212	222	232	242	252	262	272	282	292	302
163	173	183	Y	203	213	223	233	243	Y	263	273	283	293	303
164	174	X	194	204	214	224	234	X	254	264	274	284	294	X
165	175	185	195	Z	215	225	235	245	255	Z	275	285	295	305
166	176	186	196	206	216	226	236	246	256	266	276	286	296	306
167	177	187	197	207	217	227	237	247	257	267	277	287	297	307
168	178	188	198	208	218	228	238	248	258	268	278	288	298	308
169	179	189	199	209	219	229	239	249	259	269	279	289	299	309
170	180	190	200	210	220	230	240	250	260	270	280	290	300	310

↓ Riverwidth					→ Riverlength									
311	321	331	341	351	361	371	381	391	401	411	421	431	441	451
312	322	332	342	352	362	372	382	392	402	412	422	432	442	452
Y	323	333	343	353	363	Y	383	393	403	413	423	Y	443	453
314	324	334	344	354	X	374	384	394	404	414	X	434	444	454
315	Z	335	345	355	365	375	Z	395	405	415	425	435	Z	455
316	326	336	346	356	366	376	386	396	406	416	426	436	446	456
317	327	337	347	357	367	377	387	397	407	417	427	437	447	457
318	328	338	348	358	368	378	388	398	408	418	428	438	448	458
319	329	339	349	359	369	379	389	399	409	419	429	439	449	459
320	330	340	350	360	370	380	390	400	410	420	430	440	450	460

↓ Riverwidth, → Riverlength

461	471	481	491
462	472	482	492
463	473	483	Y
464	474	X	494
465	475	485	495
466	476	486	496
467	477	487	497
468	478	488	498
469	479	489	499
470	480	490	500

Each technician should randomly choose a starting quadrat within the first 5 m from the starting downstream location. The technician may not always move in a straight line, depending upon how straight the channel bed is. As another example, a stretch that is *an average* of 4 m wide and 50 m in length, for 25 quadrats and 3 technicians, (and assuming the 3 technicians (X, Y, and Z) again randomly choose 2, 12, and 15 as starting locations, would result in every 24th quadrat being searched as depicted below:

↓ Riverwidth					→ Riverlength									
1	5	9	13	17	21	25	29	33	37	41	45	49	53	57
X	6	10	14	18	22	X	30	34	38	42	46	X	54	58
3	7	11	Y	19	23	27	31	35	Y	43	47	51	55	59
4	8	Z	16	20	24	28	32	Z	40	44	48	52	56	Z

↓ Riverwidth					→ Riverlength									
61	65	69	73	77	81	85	89	93	97	101	105	109	113	117
62	66	70	X	78	82	86	90	94	X	102	106	110	114	118
Y	67	71	75	79	83	Y	91	95	99	103	107	Y	115	119
64	68	72	76	80	Z	88	92	96	100	104	Z	112	116	120

↓ Riverwidth										→ Riverlength				
										161	166	171		
121	125	129	133	137	141	145	149	153	157	162	167	172	176	Z
X	126	130	134	138	142	X	150	154	158	163	168	173	177	181
123	127	131	Y	139	143	147	151	155	Y	164	169	174	178	182
124	128	Z	136	140	144	148	152	Z	160	165	X	175	179	Y

↓ Riverwidth, → Riverlength				
	188	193	198	Z
184	189	X	200	205
185	190	195	201	206
186	191	196	202	Y
187	192	197	203	208

If the above example were a straight channel, then only 200 quadrats would be available for sampling. With the extra width in some sections, the number of quadrats increases to 208, with a resulting 2 extra quadrats (27 as opposed to 25) being available for the survey. If time permits, these quadrats should be searched in addition to the first 25.

It may be easiest to use ropes, delineated in 1 m increments, stretched across the river.

These ropes should be held in place with rebar and spaced at 1 m increments as well. The technician on shore (along with one of the technicians in the water) can move the ropes to keep ahead of the quadrat searchers. Eight to 10 ropes may be needed depending on the width of the stream. Do not leave ropes unattended. Alternatively, it may be necessary to use meter tapes instead of ropes, especially in rivers with excessive meandering.

To search each chosen 1 m² quadrat, the technicians use their hands to feel for mussels along the surface of the channel bed. The quadrat should also be excavated to a depth of 10 cm (or deeper if mussels are found that deep) using a small hand trowel or possibly a shovel in some situations. This step is important to remove larger cobble that may impede the search. However, areas with known records of Federally listed species (Higgin's Eye Pearlymussel, Sheepnose Mussel, Spectaclecase Mussel, Salamander Mussel, or Scaleshell Mussel) are to be searched ONLY by hand as a shovel or dredge could inadvertently harm mussels. If necessary, use a sieve to sort through the substrate to search for mussels.

In water over 3 feet deep, it would be necessary to SCUBA dive to collect the mussels. The same transect-grid should be established along with the same methods of selecting the 1 m² quadrats to be searched and excavated. The only difference being that dive equipment is needed to collect the mussels. Weighted lead lines or a person guiding from the water surface will be needed in order to maintain proper spacing between quadrats. In Iowa, water in river channels moves very fast. It is critical that only fast river qualified/certified divers be used in these situations. It is probably that GPS will be needed to find the location for the quadrats in this situation.

SCUBA surveys would only be undertaken by staff whom already have the proper certification. These surveys have not been undertaken by MSIM technicians and we have no plans to implement them due to safety concerns. Iowa has been working on a mussel re-introduction program. Information on this program should be read by the technicians and notes should be made on their data sheets as to the possibility of re-introductions in the area to be examined (USFWS 2004, MCT 2003).

Mussel Handling

All mussels should be kept wet in a dive bag until measurements have been completed and the mussels can be replaced at the site from which they were discovered. Mussels should be removed from the water for the shortest amount of time possible to minimize disturbance and mortalities.

In addition to species identification, the length of each mussel should be recorded. This length is measured from the posterior to the anterior margin of the shell (the longest length of the shell). Also, in some species, it should be possible to tell the sex of the mussel by examining the shell. Mussel shells are differently shaped between the sexes. The sex of each individual should also be recorded, if possible.

When placing mussels back into the water, it is best to lay them lightly on their side so they can orient themselves in the proper direction. If however, a trained IWAP Mussel Subcommittee expert member accompanies the field crew, he or she may place the mussel on its foot in the substrate such that the siphon end is up and pointing upstream. Placing the mussel upside down in the substrate can lead to the death of the mussel, hence the directive to place the mussel on its side, so that it may right itself.

If possible, mark each mussel with a bee tag and dental adhesive. It is not necessary to mark the mussels, as the quadrats are excavated and the site is visited only once per year.

However, marking is not time consuming, or particularly expensive. Mussels are capable of moving 20 m upstream or >40 m downstream, so estimating survival between years using mark-recapture and searching the same quadrats may be difficult, but not impossible.

The coordinates of the ends of the overall grid should be recorded using a GPS unit so that the locations can be found for later surveys. In addition, record the location of each excavated quadrat.

Environmental Data Collection

Standard weather conditions should be recorded on the datasheet.

Equipment List

- Plastic calipers
- Inflatable life jackets
- Knee pads
- Small spade for excavation
- Mesh dive bags
- Buckets
- Gloves (E.g. dish washing gloves)
- Delineated ropes
- Rebar
- Flagging tape
- Sieve
- Bleach
- Spray Bottles
- Standard field kit: Clip board, weather kestrel, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & rite-in-rain data sheets.

Staff & Training

A crew of 4 people will allow one person to stay on land as the data recorder as the other technicians call out the information from the water.

Mussel identification must be learned with hands on experience. The best starting place will be a museum collection or a short course with a malacologist. Training (beginning in mid-July) is recommended and should include 1) field guide use and identification, 2) discussions of defining species characteristics, 3) field practice with an experienced observer, 4) proficiency testing, and 5) habitat data collection.

SCUBA divers would be needed to conduct surveys in water >3 feet deep. In fast flowing water (most of the rivers of Iowa) these certified SCUBA divers should be qualified to handle conditions in fast flowing water. This will not be done by MSIM technicians.

Data Quality & Management

Voucher Specimens

Shells of mussels may be collected and catalogued at a willing museum. No live mussels should be collected without written permission from the Iowa DNR endangered species coordinator. For individuals difficult to identify in the field (and also *in lieu* of collecting living organisms), digital photo vouchers must be made. To photograph a mussel for use as a voucher, take pictures of both sides of the mussel after it has been cleaned as much as possible (i.e. wipe off mud and algae). Also take a photograph of the beak - the raised part of the dorsal margin of the shell. This structure is also called the umbo. This photograph should be taken looking straight onto the beak, so, for example, hold the mussel so that each side is touching one of your knees.

Data Analysis

By using the quadrat design, density of mussel species will be able to be computed. Since the sex of each individual will be recorded, inferences as to sex ratios can be made as well. The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of area occupied using program R. For more information, see chapter 5 (Data Analysis). Data collected under this protocol could also be used to examine recruitment, size class distribution, and habitat preferences, depending on the number of mussels found.

Safety Issues & Considerations

As with all other protocols, basic hygiene, including washing hands prior to eating or face touching should be followed by all personnel. In searching through sediments by hand, technicians are at risk for injury due to broken glass and sharp rocks scattered along the channel bed. Iowa water is often murky with low visibility. Therefore it is advised that technicians wear gloves, for example the yellow dishwashing gloves or oyster shucking gloves available at grocery stores, to protect their hands. All technicians should have current tetanus shots before beginning work.

Working in aquatic situations can be dangerous. Technicians should be cautious of slippery substrates and be aware of the speed of the river flow. Sampling should be suspended during inclement weather, including heavy rain or lightning storms. If a person is swept off their feet when wearing chest waders, it is possible that the air trapped in the bottom of the waders will force the person to travel down the channel upside down with their head below water. Therefore, it is recommended that chest waders have release snaps in the front of the bib to allow the technician to escape in that situation. It would also be advisable to wear an inflatable life jacket underneath the bib of the chest waders.

Care should be taken to decrease the probability of spreading an infectious agent, such as a fungus or virus, between wetlands. An additional concern is the potential spread of zebra mussels, an exotic species. One way to reduce the chance of spreading an infectious agent between wetlands is to allow the equipment to dry for 3-4 days between sites. This may be impractical given the short time frame available for mussel surveying in Iowa. As an alternative, it may be best to rinse the waders and all equipment with a solution of water and bleach (Miller and Payne 1998).

Spray bottles and bleach solutions are provided for this purpose.

Additional Methods for Special Locations

The USGS is currently monitoring Mississippi River Pools 8 & 13. This survey covers sections of meandering, channelized, and pool classes within each Pool as part of a Long-Term Resource Monitoring project.

Target Species

The following list of target species represents the species of greatest conservation need as chosen by the Steering committee for the Iowa Wildlife Action Plan (Zohrer et al. 2005). Limited distribution maps for these species can be found in Cummings and Mayer (1992) with additional information provided in Arbuckle (2000). Appendix 1 contains a list of additional, more common, mussel species which may be encountered during the monitoring efforts.

Target mussel species:

Common Name	Scientific Name	Habitat
Elktoe	<i>Alasmodonta marginata</i>	NE ¾ Iowa
Slippershell	<i>Alasmodonta viridis</i>	East Iowa
Flat floater	<i>Anodonta suborbiculata</i>	Mississippi River
Cylinder	<i>Anodontoides ferussacianus</i>	North central Iowa
Rock pocketbook	<i>Arcidens confragosus</i>	Mississippi River
*Spectacle case	<i>Cumberlandia monodonta</i>	Mississippi River
Purple pimpleback	<i>Cyclonaias tuberculata</i>	SE Iowa
Butterfly	<i>Ellipsaria lineolata</i>	Mississippi & Cedar Rivers
Spike	<i>Elliptio dilatata</i>	NE ¾ Iowa
Ebony shell	<i>Fusconaia ebena</i>	Mississippi River
Ozark pigtoe	<i>Fusconaia ozarkensis</i>	
*Higgin's eye pearlymussel	<i>Lampsilis higginsii</i>	Mississippi River & tributaries
Yellow sandshell	<i>Lampsilis teres anodontoides</i>	NE 2/3 Iowa
Slough sandshell	<i>Lampsilis teres teres</i>	NE 2/3 Iowa
Creek heelsplitter	<i>Lasmigona compressa</i>	NE 2/3 Iowa
Fluted shell	<i>Lasmigona costata</i>	NE ¾ Iowa
Pondmussel	<i>Ligumia subrostrata</i>	Des Moines & Iowa Rivers
Hickorynut	<i>Obovaria olivaria</i>	Mississippi River
*Bullhead (Sheepnose)	<i>Plethobasus cyphus</i>	Mississippi & Des Moines Rivers
Round pigtoe	<i>Pleurobema sintoxia</i>	NE ¾ Iowa
Monkeyface	<i>Quadrula metanerva</i>	NE 2/3 Iowa
Wartyback	<i>Quadrula nodulata</i>	Mississippi River
Strange floater (Squawfoot)	<i>Strophitus undulates</i>	NE ¾ Iowa
Lilliput	<i>Toxoplasma parvus</i>	NE 2/3 Iowa
Pistolgrip	<i>Tritogonia verrucosa</i>	Mississippi, Iowa, & Des Moines Rivers
Fawnsfoot	<i>Truncilla donaciformis</i>	East Iowa
Pondhorn	<i>Unio merus tetrasmus</i>	South central Iowa
Paper pondshell	<i>Utterbackia imbecillis</i>	NE ¾ Iowa
Ellipse	<i>Venustaconcha ellipsiformis</i>	East 2/3 Iowa

*Federally listed species.

Chapter Twenty: Crayfish Monitoring Protocol

Crayfish Monitoring

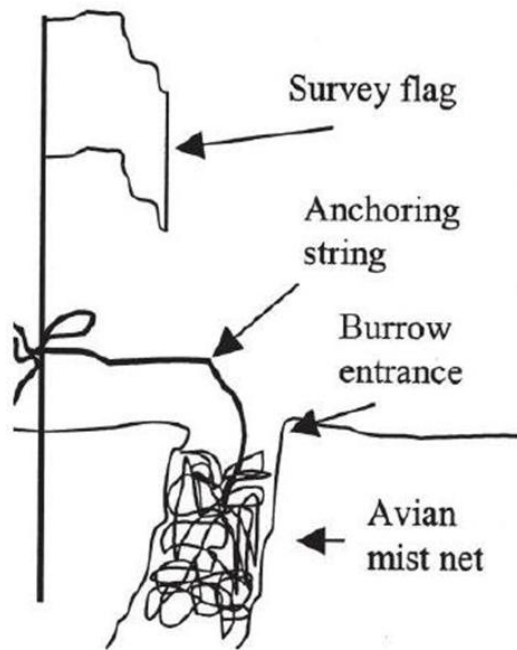
Two trapping methods will be implemented for surveying and monitoring crayfish populations: minnow traps and the burrowing crayfish net (Welch and Eversole 2006). Crayfish are classified into three groups based upon the degree to which they utilize burrows (Hobbs 1989). Primary burrowers spend the majority of their life cycle in burrows emerging only to mate or find food. Secondary burrowers will occupy burrows during part of their life cycle, but will emerge when the mouth of the burrow is flooded or for other purposes. Tertiary burrowers are found primarily in the water column and only utilize burrows during a drought to avoid desiccation (Hobbs 1989). Minnow traps are commonly used for capturing crayfish and will catch individuals of all three groups; however, they are used to target secondary and tertiary burrowers because these species spend more time in open water. The burrowing crayfish net is a method for targeting primary burrowing crayfish. This is one of a few methods for capturing burrowing crayfish and is preferred over burrow excavation because it does not destroy the burrow or burrow complex.

Survey Methods

Two different types of traps will be utilized for surveying crayfish populations: minnow traps (Dorn et al. 2005) and burrowing crayfish nets (Welch and Eversole 2006). Minnow traps are a common sampling methods for crayfish and, overall, provide comparable estimates of population density to other sampling methods (Capelli and Magnuson 1983). In addition, they are easy to place and require less effort than other active sampling methods. The burrowing crayfish net is one of several methods identified for capturing primary and secondary burrowing crayfish. The nets are simple to create and implement and yield a higher capture efficiency than other methods (e.g. Norrocky burrowing crayfish trap [Norrocky 1984]). Trapping will occur at each property on a schedule similar to that used for amphibian and reptile aquatic trapping (see Chapter Ten Amphibian and Reptile Monitoring Protocol for additional information). Aquatic habitats suitable for crayfish and crayfish burrow complexes will be identified and marked on each property for subsequent sampling. Traps will be placed in water bodies and burrows for three days (two nights) and checked daily. Trapping will be done once during spring, summer, and fall survey seasons.

Six minnow traps will be set in 1-2 water bodies randomly selected for that year. At least one of the waterbodies will be sampled every season and every year that the property is surveyed. If possible, multiple traps will be placed in a single water body with some traps being placed near shore and others being placed away from shore in open-water habitats. Traps will be placed on the bottom of the water body with the trap opening submerged in water with the trap opening submerged in water, anchored with a bamboo stake or other small post, and marked with flagging for identification. A pocket of air will be created at the top (commonly with an empty soda bottle) of the trap to allow for respiration of amphibians and reptiles caught in the traps. The funnel entrance of the trap should be ~3.5 cm to facilitate capture of all sizes of crayfish (Somers and Stechey 1986). The effective sampling area of a single minnow trap is 0.01 ha (Acosta and Perry 2000). Therefore, minnow traps should be placed at least 6 m apart to avoid competition of traps for individual crayfish. The number of traps placed in a single waterbody will vary depending upon the size of the water body but minimum of two traps should be placed in each water body. If multiple water bodies are identified on a single property, a minimum of two traps should be placed in each sampled water body. Although Iowa MSIM does not bait minnow traps, traps can be baited with fish (i.e. sardines, carp, or other raw fish). On occasion crayfish may be captured in other aquatic traps used for amphibians and reptiles, which are baited with fish (Somers and Stechey 1986).

Burrowing crayfish nets are constructed from discarded avian mist nets and measure approximately 20 x 150 cm (Welch and Eversole 2006). Mesh size of the net can vary, but we will utilize nets with a mesh size of 38 mm. After nets are cut to the appropriate size, they are then folded over cross-wise several times until they measure 20 x 20 cm. The last fold of the net is then tied with an anchor string to a wire marking flag near the entrance of the burrow. A maximum of 20 burrowing crayfish nets will be deployed per property (if more than one crayfish colony exists). Nets will be deployed in burrows that appear active (see below for more information on locating colonies and burrows).



Schematic of the burrowing crayfish net from Welch and Eversole (2006).

Crayfish burrows and burrow colonies are located by searching the shorelines of aquatic habitats and adjacent upland habitats present on the property. Burrows and burrow colonies will be marked with marking flags and marked in a GPS unit for subsequent sampling. Burrowing crayfish nets will be deployed in active burrows; those burrows that have a distinctive “chimney” (pile of excavated substrate above the burrow entrance) and evidence of recent excavation.

All individual crayfish will be removed from the traps and identified to species. In addition, sex of each crayfish will be determined. Crayfish are sexed by examining the first two sets of swimmerets (small leg-like structures on the ventral side of the abdomen behind the walking legs); they are fully developed and prong-like on males and generally undeveloped on females. Females will also have an opening between the last two pairs of legs where they receive and store sperm. Crayfish cannot be identified in the field, so species should be grouped into similar visual groups (ie. Unknown A, Unknown B). From each group identified, 1-2 male specimens should be collected and preserved in 10% formalin or 70% ethanol solution. These specimens will be identified to species under a dissecting scope using the first two sets of swimmerets. The carapace length will be measured to the nearest 0.1 mm using a dial caliper for collected specimen.

Although not currently used, each individual crayfish can be marked when first captured to determine capture efficiency and recapture rates. Crayfish can be marked by punching holes in the telson and uropods, a method outlined by Guan (1997). This is a semi-permanent marking method and allows for 1,350 unique mark combinations. If needed, we can combine this marking methods with one that involves clipping pleura on the abdominal segments of the crayfish to allow for 10,800 unique mark combinations. Holes will be punched in the telson and uropods of crayfish using a needle (0.5-0.8 mm diameter). A new needle will be used to mark each crayfish to prevent potential spread of disease between individuals. Previously marked crayfish that are recaptured will be recorded.

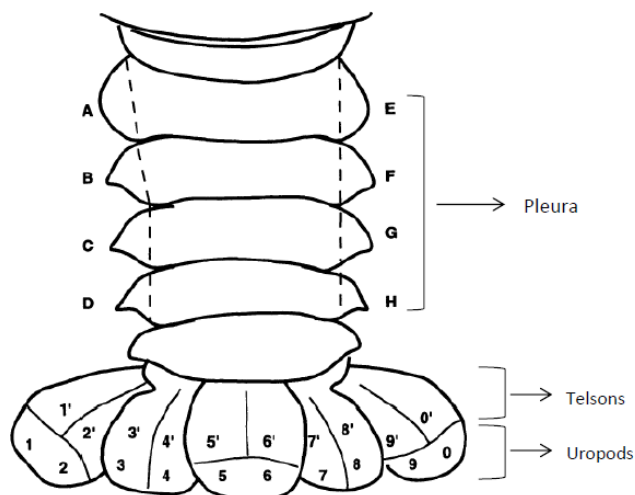


Diagram of the telsons, uropods, and pleura used for individually marking crayfish (Guan 1997).

Environmental Data Collection

Before checking traps each day, weather conditions will be recorded that may affect the activity levels of crayfish and thus, affect capture rate. The weather conditions include: ambient temperature (°F), wind speed (miles/hour), and water temperature (°F). These measurements are recorded on the datasheet.

Equipment List

- Weather kestrel temperature and wind gauge
- Minnow traps (Gee Minnow trap 9" x 17.5", ¼" mesh)
- Trap bait (Carp or sardines)
- Burrowing crayfish nets
- Bamboo stakes
- Marking flags
- Minnow net (for holding individual crayfish)
- Needles and styrofoam pad (for marking crayfish)
- Dial calipers
- Jar with 10% formalin or 70% ethanol solution (for preserving crayfish)
- Field guide
- Hip boots
- Digital camera
- GPS unit
- Standard field kit: Clip board, weather kestrel, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & rite-in-rain data sheets.

Target Species

Crayfish have been included in the 2015 Revision of the IWAP (Reeder & Clymer 2015).

All native crayfish have been designated as Data Deficient (*).

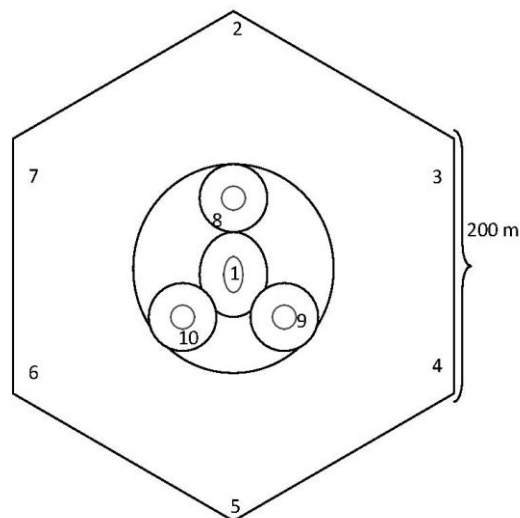
Common Name	Scientific Name	Habitat
Devil Crayfish*	<i>Cambarus diogenes</i>	Streams & creeks
Calico Crayfish*	<i>Orconectes immunis</i>	Sloughs & floodplains
Golden Crayfish*	<i>Orconectes luteus</i>	Rivers, streams, creeks
Northern Clearwater Crayfish*	<i>Orconectes propinquus</i>	Cool water streams
Virile Crayfish*	<i>Orconectes virilis</i>	Rivers, streams, and ponds lacking predatory fish
White River Crayfish*	<i>Procambarus acutus</i>	
Prairie Crayfish*	<i>Procambarus gracilis</i>	Grasslands

Chapter Twenty-one: Terrestrial Plant Species & Habitat Classification Monitoring

The principal motivation for collecting information on habitat and vegetative characteristics is to correlate faunal species presence or absence back to the properties. One goal of the Iowa statewide monitoring program is to collect data that can be compared to data collected from other places. Data comparisons are most appropriate when the information has been collected in a similar manner. To that end, the following protocol has been designed based upon the USFS Forestry Inventory and Analysis (FIA) protocols. Some changes have been made to both protocols over the last several years but there are still enough similarities to be able to make comparisons with the data sets.

Habitat Classification & Monitoring

Within each of the permanent hexagonal sampling sites, a total of 10 possible vegetation plots per property are established as the diagram below depicts. Eight of the plots will be sampled each year (BPC1-7, plus 1 additional plot). If time permits, all 10 plots should be sampled. By having the additional 3 plots in the center of the hexagon, this should result in sampling more of the representative habitat on which the hexagon is centered.



Nested plots centered on middle of hexagonal sampling area, see diagrams below for additional information.

To find with GIS, the computations for finding the center of plots labeled 8, 9, and 10 (one of which will be sampled on each property) in diagram below are:

Point 8 = X (UTM from BPC1) + 0; and Y (UTM of BPC1) + 36
 Point 9 = X (UTM from BPC1) + 31; and Y (UTM of BPC1) - 18
 Point 10 = X (UTM from BPC1) - 31; and Y (UTM of BPC1) - 18

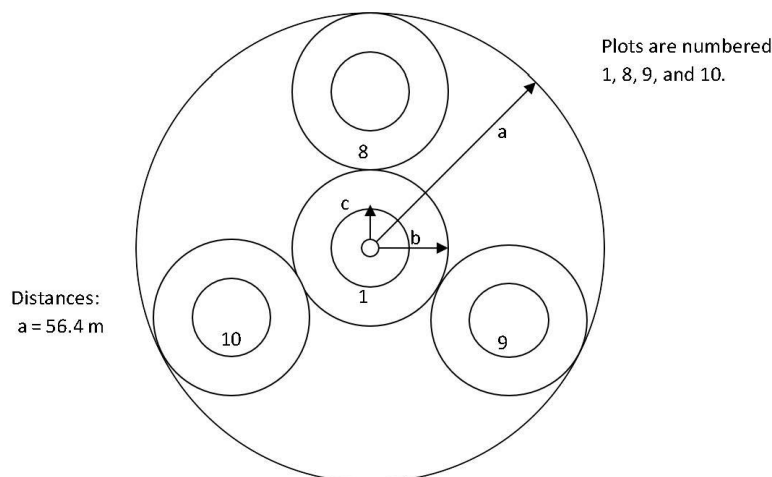
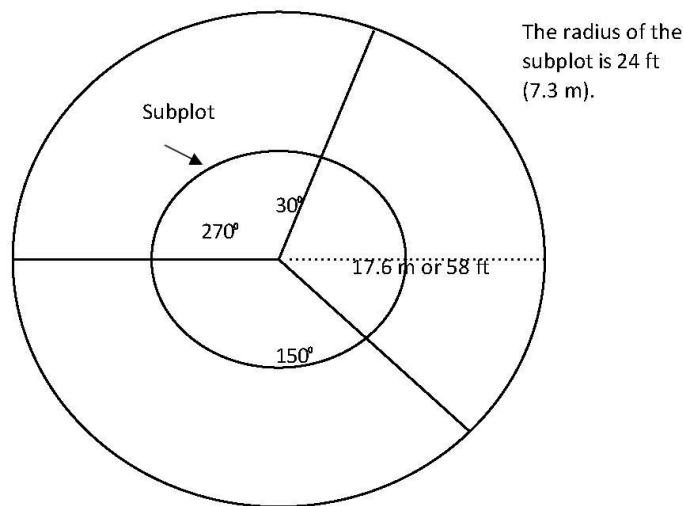
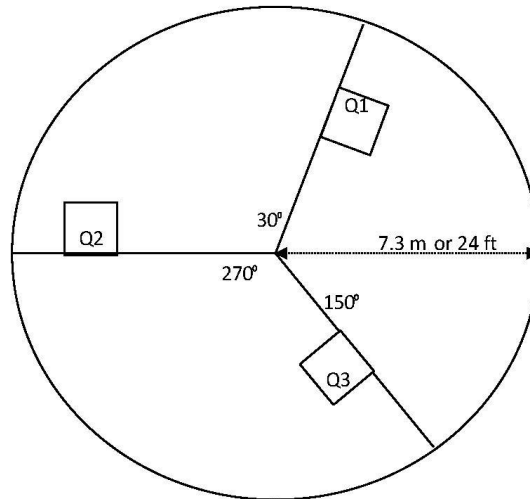


Diagram of nested plots centered around the center of the hexagonal sampling plot.



Within & around any given plot - woody debris transects, choose 1 per any given plot.



Q1, Q2, & Q3 are located 4.572 m (or 15 ft) from the center of each subplot at the angles indicated. Q1, Q2, & Q3 are all 1 m² quadrats.

Diagram of the interior of any one of the subplots associated with the larger plant composition monitoring area.

Survey Methods

This section of the monitoring plan is more important for characterizing the habitat available to species noted in the permanent sampling sites than it is for comparing the area to other places within the US. Therefore, this protocol could be changed significantly from the USFS MSIM or FIA protocols. Ideally 2 technicians will complete the surveys at each plot.

The following are the definition of terms used throughout this section.

- **Hex:** Area encompassing BPC 1-7. These locations are most often placed in a hexagonal pattern with BPC1 in the center.
- **Plot:** Area in which 1 vegetation survey is conducted (17.6m radius around BPCs 1-7 and 1 additional point).
- **Subplot:** Smaller sampling area within the plot (7.3m radius around BPCs 1-7 and 1 additional point).
- **Quadrats:** 1-m² areas within the subplot in which specific measurements (Robel pole height and plant species composition) are taken at. These quadrats are always placed 4.5m away from the center pole in the azimuths of 30°, 150° and 270°.

Plot Layout

The first plot is centered directly over the middle of the hexagon (around bird point count station #1). Each additional bird point count station (#2-7) will have a terrestrial habitat plot centered around it. The center of subplots 8-10 are located 36.4 m (120 ft) from the center of plot 1 at angles of 120°, 240°, and 360°, respectively. At least 8 plots will be surveyed each year (BPC #1-7, and 1 additional plot [8, 9, or 10]). If time allows all 10 plots should be surveyed.

Each plot has a subplot (7.3 m radius) that contains 3 potential 1 m² quadrats located 4.562 m (15 ft) from the center of the plot. Two of these quadrats will be randomly chosen at each plot to be sampled for plant species composition. The other quadrat location will be used to collect Robel pole (vertical vegetation) measurements. When time allows, additional plant species composition surveys can be done at the third quadrat location. When possible, marking the location of each quadrat with GPS should be done to facilitate future measurements in the same location. Within each hex, at least 16 of the 30 potential quadrats will be measured for plant composition.

Within each of the plots, 1 woody debris transect (extending 17.6 m or 58 ft) should be randomly established in 1 of the 3 pre-determined aspects (30°, 150°, 270°). Each transect is first marked in a straight line (using a hip chain, a surveyors tape, or a previously measured and marked rope) as it is critical not to bias the measurements by moving the line to include (or exclude) logs. Along this same transect, ground cover and litter depth will be measured.

Ground Truthing of Data Obtained from GIS

The data collected in Chapter 3 (Landscape Characteristics) needs to be ground-truthed and would be appropriate to ground-truth as part of this protocol.

Timing

Plots should be visited at least once in the summer (mid-June through August).

Subplot Measurements

(within each 0.017 ha (7.3 m or 24 ft) subplot)

Litter Depth

At 3 locations (2.5, 5, & 7.5 m) along one of the transects, litter depth is measured and recorded. Litter depth can be measured using a meter stick by ensuring the meter stick is touching the soil and marking the number of cm above the soil dead vegetation occurs. Care should be taken to ensure areas that have been disturbed by the animal trapping and searching efforts is avoided as this is likely to cause trampling. Please record the direction of the randomly selected transect (30°, 150°, or 270°).

General Plant List

Within each subplot (with a radius of 7.3 m or 24 feet), one technician spends 5 minutes searching for as many different plant species as possible. This search is timed exactly and does not include time spent with species identification (it is beneficial to have a well-trained botanist or at least a knowledgeable enthusiast). Another technician records the data as it is being voiced, each crew should have a system to keep unknown plant species identified. This could be done through photos, temporary plant collections, or pressing plant specimen. Make sure unknown plants can be easily matched between the individuals “collected” and those on the datasheet.

Plant Species Composition/Quadrat

The 1-m² frame is positioned on the ground at the correct quadrat location [4.5 m from the center of the plot, in 2 of 3 transects directions (30°, 150°, 270°)]. The frame should be level with the ground - to achieve this, one or two sides of the frame may need to be propped up. If the area is heavily vegetated, it may be necessary to carefully thread the frame down through the vegetation as best as possible. Should the area be on a hillside, the technician should stay downslope of the frame to avoid accidentally stepping into the quadrat.

Technicians should estimate the percent of cover of each vascular plant species that are within each quadrat. Plants that are living and plants that have died within the given year should be included. Total species composition could exceed 100% if canopies of different species overlap, covering the same ground cover, between 0 and 6 ft above the ground

surface. All 'trace' plants are recorded as <1%. Other categorical percentage classes are: 1-5%, 6-25%, 26-50%, 50-75%, 75-99%, and 100%. If bare soil, water, or rock is located within the quadrat, this should be noted on the data sheet with a percent class cover. Non-plants can be entered into the quadrat section of the database as "Null species", the estimated percent class, and in the comment box, add what it really was (e.g. water, rock, etc.). All matter within the quadrat should be included in the summation to at least 100%.

Also, within each quadrat, a 'trampling code' is assigned to quantify damage by humans or wildlife. These trampling codes are of 1 = 0-10% of quadrat trampled; 2 = 10-50% of quadrat trampled; and 3 = 50-100% of quadrat trampled.

If possible, unknown plants should be collected off the measured plot for later identification. Suggested labels for each unknown plant are located in Appendix 1. Unknown plants should be photo documented or pressed before leaving the property to ensure that all of the unknowns are there (not accidentally dropped in the field).

Canopy Cover

To provide information about canopy cover over the plot, a visual estimate (yes or no) is made 7.3m from the center point in the 4 cardinal directions. At each location, a technician will look straight up. If they see any canopy that direction is marked as "Yes". If no canopy is seen, then that direction is marked "No".

Visual Obstruction Measures

A Robel Pole is used to estimate the amount of vertical obstruction (i.e. vegetative structure). The pole should be placed in the center of the 3rd 1-m² quadrat location [4.5 m from the center of the plot, in the 3rd transect direction not used for quadrats (30°, 150°, 270°)]. Place the pole in the middle of the plot and move to the end of the attached rope (approximately 4 m). Squat down until the eye is 1 m off the ground and record the lowest number visible on the pole. Be careful not to walk through this area prior to taking the measurements. If additional quadrats are used for plant composition, Robel pole reading should be done prior to the plant composition measurement that would be using the same location. Additionally, the Robel pole and quadrat may need to be placed on opposite sides of the transect to ensure that vegetation is not trampled for either measurement.

Plot Measurements

(within each 0.1-ha [17.6 m or 58 ft radius] plot)

Woody Debris Transects

For every log greater than 7.7 cm (3 in) in diameter that touches the transect line, the following information is recorded: diameter at the small end, diameter at the large end, length to the nearest 0.5 m, and decay class.

Log Decay Class Table (adapted from Manley et al., 2004).

Decay class	Structural Integrity	Texture
1	Fresh, intact log	No rot
2	Sound	Mostly intact, but partly soft
3	Piece supports its own weight	Large pieces, but 'crunches'
4	Does not support weight, but maintains shape	Small pieces, can push a metal rod through it
5	None, crumbled and spread out on ground	Soft and powdery

Tree and Snag Measurement

Within the plot, all tree and snags ≥12.5 cm (5 in) in diameter, record the following information: species, diameter at breast height (cm), and, if a snag, decay class. Decay class is used for snags based upon the following classification table.

Snag Decay Class Table (adapted from Manley et al., 2004).

Decay class	Limbs & branches	Top of snag	Bark remaining
1	All present	Intact	100%
2	Few limbs, no fine branches	May be broken or intact	Varies

Decay class	Limbs & branches	Top of snag	Bark remaining
3	Limb stubs	Broken	Varies
4	Few or no stubs	Broken	Varies
5	None	Broken	<20%

Ground Cover Percent

As a comparison for the plant composition subplot estimates, every 5 m along the woody debris transect, the ground cover percent will be estimated within a 1-m² frame. Seven ground cover classifications (herbaceous plant, grass, shrub, tree, rock, litter, and bare soil) should be used and the percentage of ground that is within a 1-m² plot centered at the point on the transect corresponding to 5, 10, and 15 m from the center point of the subplot. Make sure to not trample the vegetation prior to taking this measurement by walking through or placing equipment inside the frame.

Suggested Order of Measurements:

*This order represents the most efficient way to conduct the survey, but it is not required to complete the survey in this manner.

1. Woody Debris transect [Plot; randomly select direction (30°, 150°, 270°)].
2. Ground Cover Percent [Plot; same direction as woody debris transect]
3. Litter Depth [Subplot; same direction as woody debris transect]
4. Plant Composition/Quadrats [Subplot; 1 on woody debris transect, 1 in another randomly selected direction (30°, 150°, 270°)].
5. Vertical Vegetation [Subplot; direction that has not been used (30°, 150°, 270°)].
6. Canopy Cover [Subplot; 4 directions (0°, 90°, 180°, 240°)].
7. Tree and Snag Measurement [Plot; all trees/snag >12.5 m DBH]
8. General Plant List [Subplot; 1 person search for 5 minutes]

Equipment List

Unknown/collected plant labels
 1 m² quadrat sampling frame
 Hand lens
 Field guides & species lists
 Stopwatch
 Hip chain, surveyors tape, or previously measured and marked rope
 Robel pole
 Meter stick
 Masking tape
 Standard field kit: Clip board, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets.

Staff & Training

Training should include 1) visits to herbarium collections - learn to identify common species and learn the correct way to press plants, 2) field trips to practice identification skills in the field, and 3) practice surveys with supervisor to ensure proper procedures are followed.

Data Quality & Management

Technicians need to understand that the correct identification of plant species is critical and the importance of following the data collection protocol exactly.

Potential sources for error in this protocol include the timing of the surveys (i.e. plants are easier to identify when blooming), returning to each site for at least 2 visits should reduce the variation associated with timing. Errors associated with the technician (diligence, species ID, etc.) can be mitigated by having different observers do the repeat visits and with 'testing' technician plant knowledge. Another testing possibility would be if both technicians are plant knowledgeable, having both record information for one quadrat and then they immediately compare data to determine discrepancies.

Data Analysis

Plant species composition data will primarily be used as covariates to correlate to wildlife species presence or absence. The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of area occupied using program R. For more information, see chapter 5 (Data Analysis).

Safety Issues & Considerations

Typical field considerations should be followed. Proper hygiene (i.e. hand washing before meals, checking for ticks & other potential parasites) should be maintained. Technicians should look out for poison ivy and poison oak.

Additional Methods for Special Locations

Press Plants

Unknown plants can be pressed using a plant press and if needed identified using a dissecting microscope, to ensure all plants found are identified to the species level.

To Monitor Vegetation (as opposed to collected habitat data to correlate to faunal species)

If possible, additional visits in the spring (mid-April through mid-June) and fall (mid-August through mid-October) could be added for a total of at least 3 habitat visits per site.

Chapter Twenty-two: Aquatic Habitat Classification Protocol

The principal motivations for collecting information on habitat and vegetative characteristics is to correlate habitat characteristics to faunal presence and absence. One goal of the Iowa statewide monitoring program is to collect data that can be compared to data collected from other places. Data comparisons are most appropriate when the information has been collected in a similar manner. To that end, the following protocol has been designed based upon the USFS Forestry Inventory and Analysis (FIA) protocols, the USFS MSIM protocols, and the Iowa DNR fish habitat protocols.

Aquatic Habitat Classification & Monitoring

The following are guidelines for those permanent sampling plots which include rivers, streams, creeks, impoundments, backwaters, artificial lakes, natural lakes, and ponds. It is in the interest of the technicians to ensure that other teams associated with that wetland are or are not collecting the same data (i.e. ESD Wetlands Water Quality Monitoring Teams).

Each wetland for which data is collected with these techniques will be categorized into one of the 8 water habitat types: rivers, streams, creeks, impoundments, backwaters, lakes, shallow lakes, and ponds. See Chapter 2 (Plot Design) for definitions of each habitat type.

Survey Methods

Included in these measurements are data that can be determined from GIS coverages in the lab prior to field work (see Chapter 3, Landscape Characteristics). Measurements include amount of roads and other impacted soils adjacent to the water body, locations of and numbers of water bodies. This information will need to be ground-truthed in the field.

Lentic (Standing Water) Site Measurements

The area of the habitat will be estimated either from GIS and aerial photos (i.e. known values for Saylorville Lake, for example) or by estimating length and width in the field and pacing the circumference of the pond, impoundment, or lake with a GPS unit in hand periodically recording point locations.

The depth of the water is determined either from known data or, if no known data exists, in the field using a meter stick in shallower water. If the deepest section of the water body is not known, then 5 depths should be taken from around the area thought to be the deepest point.

Plot Establishment

In lakes, impoundments, and ponds, 30 plots should be established extending 3 m from the shoreline into the water. Each plot should be 0.25 m wide (so basically each plot is a 0.75 m² rectangle). Plots are spaced equally around the perimeter of the lake, pond, or impoundment. For backwater areas which are not too deep, one transect is established that runs across the longest section of the wetland. Both designs (the plots-from-the-edge and the transect through the backwater) follow those of the USFS MSIM program. Again, this distance (shoreline distance) needs to be known prior to data collection to allow 30 0.75 m² rectangular plots to be spaced evenly. It may be best to decide on exact locations based on GIS coverage to ensure proper spacing of pre-selected locations in the field.

Substrate Measurements

Within each 0.75 m² rectangular plot, record the maximum depth, the depth at the end of the plot furthest from the shore or transect, and the percent of substrate covered by the following 7 categories: bedrock, rubble (contains stones, boulders, and bedrock), cobble-gravel (2-300 mm in size), sand, mud (silt or clay), organic (muck or peat), and vegetation (emergent, submergent, and logs; Cowardin et al. 1979). Percent coverage should add to 100. The substrate measurements will be used as an index for the majority of breeding areas for aquatic species (e.g. emergent vegetation and dragonflies, submergent vegetation and rocks for amphibians, cobble-gravel and certain fish species) along the edge of the wetlands.

In areas too deep to see the bottom of the plot for the substrate covers, it may be necessary to rely on taking pole-prodding or dredge-samples to determine these values. If the substrate is within reach of a pole yet the water is too murky to see clearly, it may be possible to determine substrate class by using an aluminum pole (often 12 ft in length and 1 3/8 inches in diameter) or meter stick. By using the pole to test the substrate it may be possible to determine what the area is composed of (silt vs. bedrock). Take at least 2 'jabs' within each plot where it is impossible to see the bottom. Larger areas will need more 'jabs' with the pole. However, in some situations it may be impossible to determine substrate type by either sight or pole-prodding. A dredge may be necessary to determine substrate.

Lotic (Running Water) Measurements

For some areas, i.e. wadeable stream flowing through a site classified as 'savanna', a pre-determined, downstream point of the stream will be considered the starting point and the transect of the stream (which will be monitored for fish in addition to the measures collected under the aquatic habitat protocol) distance monitored will be 30 or 35 times the stream width at that point and moving upstream. This stream-length should not exceed 400 m, even for larger rivers. In any case, the stream reach should be the same for this protocol as for the fish and mussel protocols to ensure appropriate habitat measures are collected.

Plot Establishment

Along the 30 or 35 times the length of the stream/river/creek being characterized, eleven transects should be established extending across the water, from shore to shore. Each transect should be 0.25 m wide. The spacing between the plots is approximately 3 times the stream width. The first transect should be placed randomly within the first 5 m from the edge of the start of the stream length. Each additional plot-transect should be located at a spacing of 3 times the stream width past the last plot.

Plot Measurements

Within each transect-plot, the following should be recorded:

Channel type: Riffle, run, pool, or glide. *Wetted width:* Width of water.

Bankfull width: The area of the channel from one side to the other, including the crest or almost crest area, beyond which the water would flow out onto the floodplain.

Bankfull height: How deep the water would get before flooding, so measure the height of the lower of the 2 banks.

Incised height: The depth of the incision of the channel. The incised height is always greater than or equal to the bankfull height. Incised height is also defined as the distance to the first terrace above the bankfull height.

Again, measure the lower of the 2 terraces if they differ across the stream channel.

Stream discharge: The volume of water passing a point during a given time (m³/sec). *not currently measured for Iowa MSIM

Water temperature: *not currently measured for Iowa MSIM

Water pH: The amount of acidity in the water. *not currently measured for Iowa MSIM

Conductivity: The amount of ions (e.g. salts) dissolved in the water. *not currently measured for Iowa MSIM

For this protocol, a run shall be defined as having:

- Moderate gradient with substrate of small gravel and/or cobble;
- above average water velocities;
- average depth;
- low to moderate turbulence;
- channel controlled;
- and generally associated with downstream extent of riffles.

Riffle

- Relatively high gradient with substrate of large gravel and/or cobble;
- above average water velocities;
- below average depth;
- surface turbulence;
- channel is controlled (i.e. no backwater influence);
- shallow, turbulent stream segments with higher gradients than pools or glides (Nielson and Johnson 1983).

Glide:

- Relatively low gradient with substrate of small gravel and/or silt/sand;
- below average water velocity;
- below average depth;
- no turbulence;
- variable control of channel;
- generally associated with the tails of pools and the heads of riffles;
- moderately shallow stream channels with laminar flow, lacking pronounced turbulence (Nielson and Johnson 1983).

Pool:

- Relatively low gradient with substrate of fine materials;
- below average water velocity;
- below average turbulence;
- above average depth;
- section controlled;
- often associated with a bounded head crest (upstream break in the slope) and a bounded tail crest (downstream break in the slope - in other words some sort of break in the channel, at least a partial break) (Kerschner et al. 2004);
- deeper habitats with slower current velocities (Nielson and Johnson 1983)

The width (extending perpendicular to the channel) of riparian vegetation is recorded at every plot. Riparian vegetation would include wet meadows or woody vegetation (i.e. willows) on the streambank, hillside, or floodplain. In some areas, it may be possible to do this from GIS or infra-red photos, however this information would still need to be ground-truthed.

Substrate Measurements

From the bank into the water and 0.25 m wide, shoreline substrate is measured by recording the water depth and the percentage of each 0.25 m x 0.25 m covered by the six substrate types as well as emergent and submergent vegetation. The first plot should be placed ½ on shore and 3 additional plots should be placed at equal spacing to cross the channel, but only the first of the 4 plots should be on shore. Water Depths, if it is possible to cross the channel, depth should be measured by taking 10 depth measurements at equal spacing.

For all pools in the channel reach, maximum depth and surface area are recorded, as well as documentation of the 'cause' of the pool (i.e. channel blocked by tree, rock, or sedimentation).

Woody Debris

Each piece of woody debris in the channel that is at least 3 m in length, or at least 2/3 the width of the channel, and 10 cm in diameter, should have the length and width of the woody debris piece recorded. This may be impractical for larger rivers.

Aquatic Measurements of Lotic Habitat Sampling Plots

- Channel type (record for each plot): R=Riffle, P=pool, RU=run, G=glide Wetted width: Width of water.
- Bankfull width: The width of the channel from one side to the other, including the crest or almost crest area, beyond which the water would flow out onto the floodplain.
- Bankfull height: How deep the water would get before flooding, so measure the height of the lower of the 2 banks.
- Incised height: The depth of the incision of the channel. This is the distance to the first terrace. It will be equal to or greater than the bankfull height.
- Stream discharge: The volume of water passing a point during a given time (m³/sec). Water temperature
- Water pH: The amount of acidity in the water.

- Conductivity: The amount of ions (e.g. salts) dissolved in the water. Riparian vegetation width is the width of the vegetation within the floodplain
- At 4 locations into the channel from the shore edge, record the water depth at that point and determine the amount of that part of the plot (0.25*0.25m) in each of the following categories:
 - Bedrock
 - Boulders (>300 mm)
 - Cobble (75-300 mm in size)
 - Gravel (2-75 mm)
 - Sand
 - Silt (mud or clay, organic muck or peat)
 - Emergent vegetation
 - Submergent vegetation
- Percent coverage in these 8 boxes should add to 100.
- If it is possible to cross the channel, water depth should be measured at 10 equally spaced locations.

Equipment List

Appropriate sampling frames made from PVC pipes and ropes

Compass

Hip chain, surveyors tape, or a previously measured and marked rope

Meter stick

Notes and maps of GIS coverage

GPS Unit

Chest waders

Standard field kit: Clip board, pencils, ruler, Sharpie marker, hand sanitizer, & data sheets.

Staff & Training

Two weeks of training should include 1) field trips to practice skills in the field, and 2) practice surveys with supervisor to ensure proper procedures are followed.

Data Quality & Management

Technicians need to understand the importance of following the data collection protocol exactly. Potential sources for error in this protocol include the timing of the surveys, as all properties should be covered as succinctly as possible to the paired animal protocol (e.g. lentic and aquatic trapping; lotic and electro-fishing) to decrease variation caused by seasonal rainfall variation.

Data Analysis

Aquatic habitat data will primarily be used as covariates to correlate to wildlife species presence or absence.

Safety Issues & Considerations

Typical field considerations should be followed. Proper hygiene (i.e. hand washing before meals, checking for ticks & other potential parasites) should be maintained. Technicians should also take proper precautions around water, i.e. avoiding fast, deep flowing water.

Additional Methods for Special Locations

None.

Additional Reading

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Appendix 1: Tables of Iowa Wildlife

Species list have not been updated in this version (2025), please confirm with our resources as to the correct taxonomic information and listing status of each species.

Iowa Mammals (Bowles 1975, Kane et al. 2003)

Excluding bats which are in a separate chapter. These mammals represent 49 genera, 21 families, and 6 orders.

Order	Family	Scientific name	Common name
Marsupialia	Didelphidae	<i>Didelphis virginiana</i>	Virginia opossum
Insectivora	Soricidae	<i>Blarina brevicauda</i>	Northern short-tailed shrew
		<i>Blarina b. carolinensis</i>	**Southern short-tailed shrew
		<i>Blarina hylophaga</i>	*Elliot's short-tailed shrew
		<i>Cryptotis parva</i>	^B Least shrew
		<i>Sorex cinereus</i>	Masked shrew
		<i>Sorex haydeni</i>	*Hayden's shrew
		<i>Microsorex hoyi</i>	**Pygmy shrew
	Talpidae	<i>Scalopus aquaticus</i>	Eastern mole
Lagomorpha	Leporidae	<i>Lepus townsendii</i>	White-tailed jackrabbit
		<i>Sylvilagus floridanus</i>	Eastern cottontail
Rodentia	Sciuridae	@ <i>Cynomys ludovicianus</i>	Black-tailed prairie dog
		<i>Tamias striatus</i>	Eastern chipmunk
		<i>Glaucomys volans</i>	^C Southern flying squirrel
		<i>Marmota monax</i>	Woodchuck
		<i>Sciurus carolinensis</i>	Gray squirrel
		<i>Sciurus niger</i>	Fox squirrel
		<i>Spermophilus franklinii</i>	Franklin's ground squirrel
		<i>Spermophilus tridecemlineatus</i>	Thirteen-lined ground squirrel
		<i>Tamiasciurus hudsonicus</i>	Red squirrel
	Geomyidae	<i>Geomys bursarius</i>	Plains pocket gopher
	Heteromyidae	<i>Perognathus flavescens</i>	^A Plains pocket mouse
	Castoridae	<i>Castor canadensis</i>	Beaver
	Cricetidae	<i>Clethrionomys gapperi</i>	^{A**} Red-backed vole
		<i>Microtus ochrogaster</i>	Prairie vole
		<i>Microtus pennsylvanicus</i>	Meadow vole
		<i>Microtus pinetorum</i>	Woodland vole
		<i>Ondatra zibethicus</i>	Muskrat
		<i>Onychomys leucogaster</i>	Northern grasshopper mouse
		<i>Peromyscus leucopus</i>	White-footed mouse
		<i>Peromyscus maniculatus</i>	Deer mouse
		<i>Reithrodontomys megalotis</i>	Western harvest mouse
		<i>Sigmodon hispidus</i>	**Hispid cotton rat
		<i>Synaptomys cooperi</i>	^B Southern bog lemming
	Muridae	@ <i>Mus musculus</i>	House mouse
		@ <i>Rattus norvegicus</i>	Norway rat
	Zapodidae	<i>Zapus hudsonius</i>	Meadow jumping mouse
	Capromyidae	@ <i>Myocaster coypus</i>	Nutria

Order	Family	Scientific name	Common name
	Erethizontidae	<i>Erethizon dorsatum</i>	Porcupine
Carnivora	Canidae	<i>Canis latrans</i>	Coyote
		<i>Canis lupus</i>	**E Gray wolf
		<i>Urocyon cinereoargenteus</i>	Gray fox
		<i>Vulpes vulpes</i>	Red fox
	Ursidae	<i>Ursus americanus</i>	Black bear
	Procyonidae	<i>Procyon lotor</i>	Raccoon
	Mustelidae	<i>Gulo gulo</i>	**Wolverine
		<i>Lutra canadensis</i>	River otter
		<i>Martes pennanti</i>	Fisher
		<i>Mephitis mephitis</i>	Striped skunk
		<i>Mustela erminea</i>	Ermine
		<i>Mustela frenata</i> - 2 subspecies in Bowles: <i>M.f. primulina</i> & <i>m.f. spadix</i>	Long-tailed weasel
		<i>Mustela nivalis</i>	Least weasel
		<i>Mustela vison</i>	Mink
		<i>Spilogale putorius</i>	^A Spotted Skunk
		<i>Taxidea taxus</i>	Badger
	Felidae	<i>Felis concolor</i>	**Mountain lion
		<i>Lynx canadensis</i>	**Lynx
		<i>Lynx rufus</i>	Bobcat
Artiodactyla	Cervidae	<i>Cervus elaphus canadensis</i>	**Wapiti
		<i>Odocoileus hemionus</i>	**Mule deer
		<i>Odocoileus virginianus</i>	White-tailed deer
	Antilocapridae	<i>Antilocapra americana</i>	**Pronghorn
	Bovidae	<i>Bison bison</i>	**Bison

*Indicates species in GAP (Kane et al. 2003) but not in Bowles 1975.

**Indicates species in Bowles 1975, but not in GAP (Kane et al. 2003).

@Introduced species.

^AIowa endangered species.

^BIowa threatened species.

^CIowa species of special concern.

^EFederally endangered species.

Iowa Bats (Bowles 1975, Kane et al. 2003)

These animals represent 7 genera, 2 families, and 1 order.

Order	Family	Scientific name	Common name
Chiroptera	Vespertilionidae	<i>Myotis septentrionalis</i>	^D **Northern myotis
		<i>Myotis lucifugus</i>	Little brown bat
		<i>Myotis sodalis</i>	^A EIndiana bat
		<i>Lasionycteris noctivagans</i>	Silver-haired bat
		<i>Pipistrellus subflavus</i>	Eastern pipistrelle
		<i>Eptesicus fuscus</i>	Big brown bat
		<i>Lasiurus borealis</i>	Red bat
		<i>Lasiurus cinereus</i>	Hoary bat

Order	Family	Scientific name	Common name
		<i>Nycticeius humeralis</i>	Evening bat
	Molossidae	<i>Tadarida macrotis</i>	*Big free-tailed bat

*Species listed in Bowles (1975), but not in GAP (Kane et al. 2003).

**Listed as Keen's myotis in Bowles (1975).

^AIowa endangered species.

^BIowa threatened species.

^CIowa species of special concern.

^DFederally threatened species.

^EFederally endangered species.

Iowa Amphibians (Christiansen & Bailey 1991, Kane et al. 2003)

These animals represent 9 genera, 7 families, and 2 orders.

Order	Family	Scientific name	Common name
Caudata	Proteidae	<i>Necturus maculosus</i>	^B Mudpuppy
	Salamandridae	<i>Notophthalmus viridescens</i>	^B Central newt
	Ambystomatidae	<i>Ambystoma tigrinum</i>	Tiger salamander
		<i>Ambystoma texanum</i>	Smallmouth salamander
		<i>Ambystoma laterale</i>	^A Blue-spotted salamander
Anura	Ranidae	<i>Lithobates pipiens</i>	Northern leopard frog
		<i>Lithobates blairi</i>	Plains leopard frog
		<i>Lithobates utricularia</i>	Southern leopard frog
		<i>Lithobates palustris</i>	Pickerel frog
		<i>Lithobates areolata</i>	^{A**} Crawfish frog
		<i>Lithobates clamitans</i>	Green frog
		<i>Lithobates catesbeiana</i>	Bullfrog
		<i>Lithobates sylvatica</i>	*Wood frog
	Hylidae	<i>Hyla chrysoscelis</i>	Cope's gray treefrog
		<i>Hyla versicolor</i>	Gray treefrog
		<i>Pseudacris crucifer</i>	Spring peeper
		<i>Pseudacris triseriata</i>	Western chorus frog
		<i>Acris crepitans</i>	Northern cricket frog
	Bufonidae	<i>Bufo americanus</i>	American toad
		<i>Bufo woodhousii woodhousii</i>	Woodhouse's toad
		<i>Bufo woodhousii fowleri</i>	Fowler's toad
		<i>Bufo cognatus</i>	Great plains toad
	Pelobatidae	<i>Spea bombifrons</i>	Plains spadefoot toad

*This species is not represented in either reference, however, it has been reported in the Iowa Frog and Toad Volunteer Survey for several years, and is believed to have entered Iowa from surrounding states.

**Indicates species in Christiansen and Bailey (1991), but not in GAP (Kane et al. 2003).

^AIowa endangered species.

^BIowa threatened species.

^CIowa species of special concern.

Iowa Reptiles (Christiansen and Bailey 1990 & 1998, Kane et al. 2003)

These animals represent 31 genera, 9 families, and 2 orders.

Order	Family	Scientific name	Common name
Testudinata	Emydidae	<i>Terrapene ornata</i>	^B Ornate box turtle
		<i>Chrysenys picta</i>	Painted turtle

Order	Family	Scientific name	Common name
		<i>Trachemys scripta</i>	Red-eared turtle
		<i>Graptemys geographica</i>	Map turtle
		<i>Graptemys pseudogeographica</i>	False map turtle
		<i>Emydoidea blandingi</i>	^B Blanding's turtle
		<i>Clemmys insculpta</i>	^A Wood turtle
	Chelydridae	<i>Chelydra serpentina</i>	**Snapping turtle
		<i>Macrolemys temmincki</i>	**Alligator snapping turtle
	Kinosternidae	<i>Kinosternon flavescens</i>	^A Yellow mud turtle
		<i>Sternotherus odoratus</i>	^B Stinkpot
	Trionychidae	<i>Apalone spinifera</i>	Spiny softshell turtle
		<i>Apalone mutica</i>	Smooth softshell turtle
Squamata	Scincidae	<i>Eumeces septentrionalis</i>	Northern prairie skink
		<i>Eumeces fasciatus</i>	Five-lined skink
		<i>Eumeces obsoletus</i>	^A Great plains skink
	Anguidae	<i>Ophisaurus attenuatus</i>	^B Slender glass lizard
	Teiidae	<i>Cnemidophorus sexlineatus</i>	Six-lined racerunner
	Colubridae	<i>Nerodia sipedon</i>	Northern water snake
		<i>Nerodia rhombifera</i>	^B Diamondback water snake
		<i>Nerodia erythrogaster</i>	Yellowbelly water snake
		<i>Nerodia erythrogaster neglecta</i>	^{A D} Copperbelly water snake
		<i>Regina grahami</i>	Graham's crayfish snake
		<i>Storeria dekayi</i>	Brown snake
		<i>Storeria occipitomaculata</i>	Northern redbelly snake
		<i>Virginia valeriae</i>	Smooth earth snake
		<i>Tropidoclonion lineatum</i>	Northern lined snake
		<i>Thamnophis sirtalis</i>	Eastern garter snake
		<i>Thamnophis radix</i>	Plains garter snake
		<i>Thamnophis proximus</i>	Western ribbon snake
		<i>Carphophis amoenus</i>	^B Western worm snake
		<i>Opheodrys vernalis</i>	^C Smooth green snake
		<i>Opheodrys aestivus</i>	**Rough green snake
		<i>Diadophis punctatus</i>	Prairie ringneck snake
		<i>Coluber constrictor</i>	Racer
		<i>Lampropeltis triangulum</i>	Milk snake
		<i>Lampropeltis calligaster</i>	Prairie kingsnake
		<i>Lampropeltis getulus</i>	^B Speckled kingsnake
		<i>Pituophis melanoleucus</i>	^C Bullsnake
		<i>Elaphe vulpina</i>	Fox snake
		<i>Elaphe obsoleta</i>	Black rat snake
		<i>Heterodon platyrhinos</i>	Eastern hognose snake
		<i>Heterodon nasicus</i>	**A Western hognose snake
	Viperidae	<i>Sistrurus catenatus</i>	^A Massasauga
		<i>Sistrurus catenatus catenatus</i>	*Eastern massasauga
		<i>Crotalus horridus</i>	Timber rattlesnake
		<i>Crotalus viridis</i>	^A Prairie rattlesnake

Order	Family	Scientific name	Common name
		<i>Agkistrodon contortrix</i>	^A Copperhead

*Indicates species in GAP (Kane et al. 2003) but not in Christiansen and Bailey (1998).

**Indicates species in Christiansen and Bailey (1990 or 1998), but not in GAP (Kane et al. 2003).

@Introduced species.

^AIowa endangered species.

^BIowa threatened species.

^CIowa species of special concern.

^DNot in GAP or Christiansen and Bailey (1998), but on the T&E list.

Iowa Breeding Birds (Kent and Dinsmore 1996, Kane et al. 2003)

These animals represent 153 genera, 42 families, and 18 orders.

Order	Family	Scientific name	Common name
Gaviformes	Gaviidae	<i>Gavia immer</i>	Common loon
Podicipediformes	Podicipedidae	<i>Podilymbus podiceps</i>	Pied-billed grebe
		<i>Podiceps grisegena</i>	Red-necked grebe
		<i>Podiceps nigricollis</i>	Eared grebe
		<i>Aechmophorus occidentalis</i>	Western grebe
Pelecaniformes	Pelecanidae	<i>Pelecanus erythrorhynchos</i>	American white pelican
	Phalacrocoracidae	<i>Phalacrocorax auritus</i>	Double-crested cormorant
Ciconiiformes	Ardeidae	<i>Botaurus lentiginosus</i>	American bittern
		<i>Ixobrychus exilis</i>	Least bittern
		<i>Ardea herodias</i>	Great blue heron
		<i>Ardea albus</i>	Great egret
		<i>Egretta caerulea</i>	Little blue heron
		<i>Bubulcus ibis</i>	Cattle egret
		<i>Butorides virescens</i>	Green heron
		<i>Nycticorax nycticorax</i>	Black-crowned night heron
		<i>Nyctanassa violacea</i>	Yellow-crowned night heron
	Threskiornithidae	<i>Plegadis chihi</i>	White-faced ibis
Anseriformes	Anatidae	<i>Cygnus buccinator</i>	Trumpeter swan
		<i>Cygnus olor</i>	Mute swan
		<i>Branta canadensis</i>	Canada goose
		<i>Aix sponsa</i>	Wood duck
		<i>Anas crecca</i>	Green-winged teal
		<i>Anas rubripes</i>	American black duck
		<i>Anas platyrhynchos</i>	Mallard
		<i>Anas acuta</i>	Northern pintail
		<i>Anas discors</i>	Blue-winged teal
		<i>Anas clypeata</i>	Northern shoveler
		<i>Anas strepera</i>	Gadwall
		<i>Anas americana</i>	American wigeon
		<i>Aythya valisineria</i>	Canvasback
		<i>Aythya americana</i>	Redhead
		<i>Aythya collaris</i>	Ring-necked duck
		<i>Aythya affinis</i>	Lesser scaup
		<i>Bucephala albeola</i>	Bufflehead

Order	Family	Scientific name	Common name
		<i>Lophodytes cucullatus</i>	Hooded merganser
		<i>Oxyura jamaicensis</i>	Ruddy duck
Falconiformes	Cathartidae	<i>Cathartes aura</i>	Turkey vulture
	Accipitridae	<i>Pandion haliaetus</i>	Osprey
		<i>Elanoides forticatus</i>	Swallow-tailed kite
		<i>Ictinia mississippiensis</i>	Mississippi kite
		<i>Haliaeetus leucocephalus</i>	AT Bald eagle
		<i>Circus cyaneus</i>	A Northern harrier
		<i>Accipiter striatus</i>	Sharp-shinned hawk
		<i>Accipiter cooperii</i>	Cooper's hawk
		<i>Buteo lineatus</i>	A Red-shouldered hawk
		<i>Buteo platypterus</i>	Broad-winged hawk
		<i>Buteo swainsoni</i>	Swainson's hawk
		<i>Buteo jamaicensis</i>	Red-tailed hawk
	Falconidae	<i>Falco sparverius</i>	American kestrel
		<i>Falco columbarius</i>	Merlin
		<i>Falco peregrinus</i>	A Peregrine falcon
Galliformes	Phasianidae	<i>Perdix perdix</i>	Gray partridge
		<i>Phasianus colchicus</i>	Ring-necked pheasant
		<i>Bonasa umbellus</i>	Ruffed grouse
		<i>Tympanuchus cupido</i>	Greater prairie chicken
		<i>Tympanus phasianellus</i>	Sharp-tailed grouse
		<i>Meleagris gallopavo</i>	Wild turkey
	Odontophoridae	<i>Colinus virginianus</i>	Northern bobwhite
Gruiformes	Rallidae	<i>Rallus elegans</i>	A King rail
		<i>Rallus limicola</i>	Virginia rail
		<i>Porzana carolina</i>	Sora
		<i>Gallinula chloropus</i>	Common moorhen
		<i>Fulica americana</i>	American coot
	Gruidae	<i>Grus canadensis</i>	Sandhill crane
		<i>Grus americana</i>	Whooping crane
Charadriiformes	Charadriidae	<i>Charadrius melodus</i>	AT Piping plover
		<i>Charadrius vociferus</i>	Killdeer
	Scolopacidae	<i>Actitis macularia</i>	Spotted sandpiper
		<i>Bartramia longicauda</i>	Upland sandpiper
		<i>Numenius americanus</i>	Long-billed curlew
		<i>Limosa fedoa</i>	Marbled godwit
		<i>Gallinago gallinago</i>	Wilson's snipe
		<i>Scolopax minor</i>	American woodcock
		<i>Phalaropus tricolor</i>	Wilson's phalarope
	Laridae	<i>Larus pipixcan</i>	Franklin's gull
		<i>Larus delawarensis</i>	Ring-billed gull
		<i>Sterna forsteri</i>	C Forster's tern
		<i>Sterna antillarum</i>	AE Least tern
		<i>Chlidonias niger</i>	C Black tern

Order	Family	Scientific name	Common name
Columbiformes	Columbidae	<i>Columa livia</i>	Rock pigeon
		<i>Streptopelia decaocto</i>	Eurasian collared dove
		<i>Zenaida macroura</i>	Mourning dove
		<i>Ectopistes migratorius</i>	Passenger pigeon (extinct)
Psittaciformes	Psittacidae	<i>Conuropsis carolinensis</i>	Carolina parakeet (extinct)
Cuculiformes	Cuculidae	<i>Coccyzus erythrophthalmus</i>	Black-billed cuckoo
		<i>Coccyzus americanus</i>	Yellow-billed cuckoo
Strigiformes	Tytonidae	<i>Tyto alba</i>	A Barn owl
	Strigidae	<i>Otus asio</i>	Eastern screech-owl
		<i>Bubo virginianus</i>	Great horned owl
		<i>Speotyto cunicularia</i>	Burrowing owl
		<i>Strix varia</i>	Barred owl
		<i>Asio otus</i>	B Long-eared owl
		<i>Asio flammeus</i>	A Short-eared owl
Caprimulgiformes	Caprimulgidae	<i>Chordeiles minor</i>	Common nighthawk
		<i>Caprimulgus carolinensis</i>	Chuck-will's-widow
		<i>Caprimulgus vociferus</i>	Whip-poor-will
Apodiformes	Apodidae	<i>Chaetura pelagica</i>	Chimney swift
	Trochilidae	<i>Archilochus colubris</i>	Ruby-throated hummingbird
Coraciiformes	Alcedinidae	<i>Ceryle alcyon</i>	Belted kingfisher
Piciformes	Picidae	<i>Melanerpes erythrocephalus</i>	Red-headed woodpecker
		<i>Melanerpes carolinensis</i>	Red-bellied woodpecker
		<i>Sphyrapicus varius</i>	Yellow-bellied sapsucker
		<i>Picoides pubescens</i>	Downy woodpecker
		<i>Picoides villosus</i>	Hairy woodpecker
		<i>Colaptes auratus</i>	Northern flicker
		<i>Dryocopus pileatus</i>	Pileated woodpecker
Passeriformes	Tyrannidae	<i>Contopus virens</i>	Eastern wood-pewee
		<i>Empidonax virens</i>	Acadian flycatcher
		<i>Empidonax traillii</i>	Willow flycatcher
		<i>Empidonax minimus</i>	Least flycatcher
		<i>Sayornis phoebe</i>	Eastern phoebe
		<i>Sorornis saya</i>	Say's phoebe
		<i>Myiarchus crinitus</i>	Great crested flycatcher
		<i>Tyrannus verticalis</i>	Western kingbird
		<i>Tyrannus tyrannus</i>	Eastern kingbird
		<i>Tyrannus forficatus</i>	Scissor-tailed flycatcher
	Alaudidae	<i>Eremophila alpesteris</i>	Horned lark
	Hirundinidae	<i>Progne subis</i>	Purple martin
		<i>Tachycineta bicolor</i>	Tree swallow
		<i>Stelgidopteryx serripennis</i>	Northern rough-winged swallow
		<i>Riparia riparia</i>	Bank swallow
		<i>Hirundo pyrrhonota</i>	Cliff swallow
		<i>Hirundo rustica</i>	Barn swallow
	Corvidae	<i>Cyanocitta cristata</i>	Blue jay

Order	Family	Scientific name	Common name
		<i>Pica pica</i>	Black-billed magpie
		<i>Corvus brachyrhynchus</i>	American crow
	Paridae	<i>Parus atricapillus</i>	Black-capped chickadee
		<i>Parus bicolor</i>	Tufted titmouse
	Sittidae	<i>Sitta canadensis</i>	Red-breasted nuthatch
		<i>Sitta carolinensis</i>	White-breasted nuthatch
	Certhiidae	<i>Certhia americana</i>	Brown creeper
	Troglodytidae	<i>Salpinctes obsoletus</i>	Rock wren
		<i>Thryothorus ludovicianus</i>	Carolina wren
		<i>Thryomanes bewickii</i>	Bewick's wren
		<i>Troglodytes aedon</i>	House wren
		<i>Troglodytes troglodytes</i>	Winter wren
		<i>Cistothorus platensis</i>	Sedge wren
		<i>Cistothorus palustris</i>	Marsh wren
	Muscicapidae	<i>Poliophtila caerulea</i>	Blue-gray gnatcatcher
		<i>Sialia sialis</i>	Eastern bluebird
		<i>Catharus fuscescens</i>	Veery
		<i>Hylocichla mustelina</i>	Wood thrush
		<i>Turdus migratorius</i>	American robin
	Mimidae	<i>Dumetella carolinensis</i>	Gray catbird
		<i>Mimus polyglottos</i>	Northern mockingbird
		<i>Toxostoma rufum</i>	Brown thrasher
	Bombycilla	<i>Bombycilla cedrorum</i>	Cedar waxwing
	Laniidae	<i>Lanius ludovicianus</i>	Loggerhead shrike
	Sturnidae	<i>Sturnus vulgaris</i>	European starling
	Vireonidae	<i>Vireo flavifrons</i>	Yellow-throated vireo
		<i>Vireo gilvus</i>	Warbling vireo
		<i>Vireo olivaceus</i>	Red-eyed vireo
		<i>Vireo griseus</i>	White-eyed vireo
		<i>Vireo bellii</i>	Bell's vireo
	Emberizidae	<i>Vermivora pinus</i>	Blue-winged warbler
		<i>Vermivora chrysoptera</i>	Golden-winged warbler
		<i>Parula americana</i>	Northern parula
		<i>Dendroica petechia</i>	Yellow warbler
		<i>Dendroica pensylvanica</i>	Chestnut-sided warbler
		<i>Dendroica dominica</i>	Yellow-throated warbler
		<i>Dendroica discolor</i>	Prairie warbler
		<i>Dendroica cerulea</i>	Cerulean warbler
		<i>Mniotilta varia</i>	Black-and-white warbler
		<i>Setophaga ruticilla</i>	American redstart
		<i>Protonotaria citrea</i>	Prothonotary warbler
		<i>Helmitheros vermivorus</i>	Worm-eating warbler
		<i>Seiurus aurocapillus</i>	Ovenbird
		<i>Seiurus motacilla</i>	Louisiana waterthrush
		<i>Oporornis formosus</i>	Kentucky warbler

Order	Family	Scientific name	Common name
		<i>Geothlypis trichas</i>	Common yellowthroat
		<i>Wilsonia citrina</i>	Hooded warbler
		<i>Icteria virens</i>	Yellow-breasted chat
		<i>Piranga rubra</i>	Summer tanager
		<i>Piranga olivacea</i>	Scarlet tanager
		<i>Cardinalis cardinalis</i>	Northern cardinal
		<i>Pheucticus ludovicianus</i>	Rose-breasted grosbeak
		<i>Guiraca caerulea</i>	Blue grosbeak
		<i>Passerina cyanea</i>	Indigo bunting
		<i>Spiza americana</i>	Dickcissel
		<i>Pipilo erythrophthalmus</i>	Eastern towhee
		<i>Spizella passerina</i>	Chipping sparrow
		<i>Spizella pallida</i>	Clay-colored sparrow
		<i>Spizella pusilla</i>	Field sparrow
		<i>Poocetes gramineus</i>	Vesper sparrow
		<i>Chondestes grammacus</i>	Lark sparrow
		<i>Passerculus sandwichensis</i>	Savannah sparrow
		<i>Ammodramus savannarum</i>	Grasshopper sparrow
		<i>Ammodramus henslowii</i>	B Henslow's sparrow
		<i>Melospiza melodia</i>	Song sparrow
		<i>Melospiza georgiana</i>	Swamp sparrow
		<i>Dolichonyx oryzivorus</i>	Bobolink
		<i>Agelaius phoeniceus</i>	Red-winged blackbird
		<i>Sturnella magna</i>	Eastern meadowlark
		<i>Sturnella neglecta</i>	Western meadowlark
		<i>Xanthocephalus xanthocephalus</i>	Yellow-headed blackbird
		<i>Quiscalus mexicanus</i>	Great-tailed grackle
		<i>Quiscalus quiscula</i>	Common grackle
		<i>Molothrus ater</i>	Brown-headed cowbird
		<i>Icterus spurius</i>	Orchard oriole
		<i>Icterus galbula</i>	Baltimore oriole
	Fringillidae	<i>Carpodacus mexicanus</i>	House finch
		<i>Loxia curvirostra</i>	Red crossbill
		<i>Carduelis pinus</i>	Pine siskin
		<i>Carduelis tristis</i>	American goldfinch
	Passeridae	<i>Passer domesticus</i>	House sparrow
		<i>Passer montanus</i>	Eurasian tree sparrow

*Indicates species in Kent and Dinsmore (1996), but not in GAP (Kane et al. 2003).

^AIowa endangered species.

^BIowa threatened species.

^CIowa species of special concern.

^EFederally endangered species.

^TFederally threatened species.

Iowa's Migratory Birds and Vagrants (Kent and Dinsmore 1996, Kane et al. 2003)

These animals represent 116 genera, 36 families, and 12 orders.

Order	Family	Scientific name	Common name
Gaviformes	Gaviidae	<i>Gavia stellata</i>	Red-throated loon
		<i>Gavia pacifica</i>	Pacific loon
		<i>Gavia adamsii</i>	*Yellow-billed loon
Podicipediformes	Podicipedidae	<i>Podiceps auritus</i>	Horned grebe
		<i>Aechmophorus clarkii</i>	Clark's grebe
Pelicaniformes	Pelicanidae	<i>Pelcanus occidentalis</i>	Brown pelican
	Phalacrocoracidae	<i>Phalacrocorax brasilianus</i>	Neotropic cormorant
	Anhingidae	<i>Ahinga anhinga</i>	Anhinga
	Fregatidae	<i>Fregata magnificens</i>	Magnificent frigatebird
Ciconiiformes	Ardeidae	<i>Egretta thula</i>	Snowy egret
		<i>Egretta tricolor</i>	Tricolored heron
		<i>Egretta rufescens</i>	Reddish egret
	Threskiornithidae	<i>Eudocimus albus</i>	White ibis
		<i>Plegadis falcinellus</i>	Glossy ibis
		<i>Ajaia ajaja</i>	Roseate spoonbill
	Ciconiidae	<i>Mycteria americana</i>	Wood stork
Anseriformes	Anatidae	<i>Dendrocygna autumnalis</i>	Black-bellied whistling duck
		<i>Cygnus columbianus</i>	Tundra swan
		<i>Anser fabalis</i>	Bean goose
		<i>Anser albifrons</i>	Greater white-fronted goose
		<i>Chen caerulescens</i>	Snow goose
		<i>Chen rossii</i>	Ross's goose
		<i>Branta bernicla</i>	Brant
		<i>Anas querquedula</i>	Garganey
		<i>Anas cyanoptera</i>	Cinnamon teal
		<i>Anas penelope</i>	Eurasian wigeon
		<i>Aythya marila</i>	Greater scaup
		<i>Somateria mollissima</i>	Common eider
		<i>Somateria spectabilis</i>	King eider
		<i>Histrionicus histrionicus</i>	Harlequin duck
		<i>Clangula hyemalis</i>	Long-tailed duck
		<i>Melanitta nigra</i>	Black scoter
		<i>Melanitta perspicillata</i>	Surf scoter
		<i>Melanitta fusca</i>	White-winged scoter
		<i>Bucephala clangula</i>	Common goldeneye
		<i>Bucephala islandica</i>	Barrow's goldeneye
		<i>Mergus merganser</i>	Common merganser
		<i>Mergus serrator</i>	Red-breasted merganser
Falconiformes	Cathartidae	<i>Coragyps atratus</i>	Black vulture
	Accipitridae	<i>Accipiter gentilis</i>	Northern goshawk
		<i>Buteo regalis</i>	Ferruginous hawk
		<i>Buteo lagopus</i>	Rough-legged hawk
		<i>Aquila chrysaetos</i>	Golden eagle

Order	Family	Scientific name	Common name
	Falconidae	<i>Falco rusticolus</i>	Gyr Falcon
		<i>Falco mexicanus</i>	Prairie falcon
Gruiformes	Rallidae	<i>Coturnicops noveboracensis</i>	Yellow rail
		<i>Laterallus jamaicensis</i>	Black rail
		<i>Porphyryla martinica</i>	Purple gallinule
Charadriiformes	Charadriidae	<i>Pluvialis squatarola</i>	Black-bellied plover
		<i>Pluvialis dominicus</i>	American golden-plover
		<i>Charadrius alexandrinus</i>	Snowy plover
		<i>Charadrius semipalmatus</i>	Semipalmated plover
	Recurvirostridae	<i>Himantopus mexicanus</i>	Black-necked stilt
		<i>Recurvirostra americana</i>	American avocet
	Scolopacidae	<i>Tringa melanoleuca</i>	Greater yellowlegs
		<i>Tringa flavipes</i>	Lesser yellowlegs
		<i>Tringa solitaria</i>	Solitary sandpiper
		<i>Catoptrophorus semiplamatus</i>	Willet
		<i>Numenius borealis</i>	^E Eskimo curlew
		<i>Numenius phaeopus</i>	Whimbrel
		<i>Limosa haemastica</i>	Hudsonian godwit
		<i>Arenaria interpres</i>	Ruddy turnstone
		<i>Calidris canutus</i>	Red knot
		<i>Calidris alba</i>	Sanderling
		<i>Calidris pusilla</i>	Semipalmated sandpiper
		<i>Calidris mauri</i>	Western sandpiper
		<i>Calidris minutilla</i>	Least sandpiper
		<i>Calidris fuscicollis</i>	White-rumped sandpiper
		<i>Calidris bairdii</i>	Baird's sandpiper
		<i>Calidris melanotos</i>	Pectoral sandpiper
		<i>Calidris acuminata</i>	Sharp-tailed sandpiper
		<i>Calidris alpina</i>	Dunlin
		<i>Calidris ferruginea</i>	Curlew sandpiper
		<i>Calidris himantopus</i>	Stilt sandpiper
		<i>Tryngites subruficollis</i>	Buff-breasted sandpiper
		<i>Philomachus pugnax</i>	Ruff
		<i>Limnodromus griseus</i>	Short-billed dowitcher
		<i>Limnodromus scolopaceus</i>	Long-billed dowitcher
		<i>Phalaropus lobatus</i>	Red-necked phalarope
		<i>Phalaropus fulicaria</i>	Red phalarope
	Laridae	<i>Stercorarius pomarinus</i>	Pomarine jaeger
		<i>Stercorarius parasiticus</i>	Parasitic jaeger
		<i>Stercorarius longicaudus</i>	Long-tailed jaeger
		<i>Larus atricilla</i>	Laughing gull
		<i>Larus minutus</i>	Little gull
		<i>Larus ridibundus</i>	Black-headed gull
		<i>Larus philadelphia</i>	Bonaparte's gull
		<i>Larus canus</i>	Mew gull

Order	Family	Scientific name	Common name
		<i>Larus californicus</i>	California gull
		<i>Larus argentatus</i>	Herring gull
		<i>Larus thayeri</i>	Thayer's gull
		<i>Larus glaucoides</i>	Iceland gull
		<i>Larus fuscus</i>	Lesser black-backed gull
		<i>Larus schistisagus</i>	Slaty-backed gull
		<i>Larus hyperboreus</i>	Glaucous gull
		<i>Larus marinus</i>	Great black-backed gull
		<i>Rissa tridactyla</i>	Black-legged kittiwake
		<i>Rhodostethia rosea</i>	Ross's gull
		<i>Xema sabini</i>	Sabine's gull
		<i>Pagophila eburnea</i>	Ivory gull
		<i>Sterna caspia</i>	Caspian tern
		<i>Sterna hirundo</i>	Common tern
		<i>Sterna pardisaea</i>	*Arctic tern
	Alcidae	<i>Uria lomvia</i>	Thick-billed murre
		<i>Brachyramphus marmoratus</i> or <i>B.m. perdix</i>	Long-billed murrelet
		<i>Synthliboramphus antiquus</i>	Ancient murrelet
	Columbidae	<i>Zenaida asiatica</i>	*White-winged dove
		<i>Columbina passerina</i>	Common ground-dove
Cuculiformes	Cuculidae	<i>Crotophaga sulcirostris</i>	Groove-billed ani
Strigiformes	Strigidae	<i>Nyctea scandiaca</i>	Snowy owl
		<i>Surnia ulula</i>	Northern hawk owl
		<i>Strix nebulosa</i>	Great gray owl
		<i>Aegolius acadicus</i>	Northern saw-whet owl
	Trochilidae	<i>Selasphorus rufus</i>	Rufous hummingbird
Piciformes	Picidae	<i>Melanerpes lewis</i>	Lewis's woodpecker
		<i>Picoides arcticus</i>	Black-backed woodpecker
Passeriformes	Tyrannidae	<i>Contopus borealis</i>	Olive-sided flycatcher
		<i>Contopus sordidulus</i>	Western wood-pewee
		<i>Empidonax flaviventris</i>	Yellow-bellied flycatcher
		<i>Empidonax alnorum</i>	Alder flycatcher
		<i>Empidonax species</i>	"Western" flycatcher
		<i>Pyrocephalus rubinus</i>	Vermillion flycatcher
	Corvidae	<i>Perisoreus canadensis</i>	Gray jay
		<i>Gymnorhinus cyanocephalus</i>	Pinyon jay
		<i>Nucifraga columbiana</i>	Clark's nutcracker
		<i>Corvus ossifragus</i>	Fish crow
		<i>Corvus corax</i>	Common raven
	Paridae	<i>Parus hudsonicus</i>	Boreal chickadee
	Sittidae	<i>Sitta pygamaea</i>	Pygmy nuthatch
	Muscicapidae	<i>Regulus satrapa</i>	Golden-crowned kinglet
		<i>Regulus calendula</i>	Ruby-crowned kinglet
		<i>Sialia currucoides</i>	Mountain bluebird

Order	Family	Scientific name	Common name
		<i>Myadestes townsendi</i>	Townsend's solitaire
		<i>Catharus ustulatus</i>	Gray-cheeked thrush
		<i>Catharus ustulatus</i>	Swainson's thrush
		<i>Catharus guttatus</i>	Hermit thrush
		<i>Ixoreus naevius</i>	Varied thrush
	Mimidae	<i>Oreoscoptes montanus</i>	Sage thrasher
		<i>Toxostoma curvirostre</i>	Curve-billed thrasher
	Motacillidae	<i>Anthus rubescens</i>	American pipit
		<i>Anthus spragueii</i>	*Sprague's pipit
	Bombycillidae	<i>Bombycilla garrulus</i>	Bohemian waxwing
	Laniidae	<i>Lanius excubitor</i>	Northern shrike
	Sturnidae	<i>Vireo solitarius</i>	Blue-headed vireo
		<i>Vireo philadelphicus</i>	Philadelphia vireo
	Emberizidae	<i>Vermivora peregrina</i>	Tennessee warbler
		<i>Vermivora celata</i>	Orange-crowned warbler
		<i>Vermivora ruficapilla</i>	Nashville warbler
		<i>Dendroica magnolia</i>	Magnolia warbler
		<i>Dendroica tigrina</i>	Cape May warbler
		<i>Dendroica caerulescens</i>	Black-throated blue warbler
		<i>Dendroica coronata</i>	Yellow-rumped warbler
		<i>Dendroica nigrescens</i>	Black-throated gray warbler
		<i>Dendroica townsendii</i>	Townsend's warbler
		<i>Dendroica virens</i>	Black-throated green warbler
		<i>Dendroica fusca</i>	Blackburnian warbler
		<i>Dendroica pinus</i>	Pine warbler
		<i>Dendroica palmarum</i>	Palm warbler
		<i>Dendroica castanea</i>	Bay-breasted warbler
		<i>Dendroica striata</i>	Blackpoll warbler
		<i>Seiurus noveboracensis</i>	Northern waterthrush
		<i>Oporornis agilis</i>	Connecticut warbler
		<i>Oporornis philadelphia</i>	Mourning warbler
		<i>Oporornis tolmiei</i>	MacGillivray's warbler
		<i>Wilsonia pusilla</i>	Wilson's warbler
		<i>Wilsonia canadensis</i>	Canada warbler
		<i>Piranga ludoviciana</i>	Western tanager
		<i>Pheucticus melanocephalus</i>	Black-headed grosbeak
		<i>Passerina amoena</i>	Lazuli bunting
		<i>Passerina ciris</i>	Painted bunting
		<i>Pipilo chlorurus</i>	Green-tailed towhee
		<i>Pipilo maculatus</i>	Spotted towhee
		<i>Spizella arborea</i>	American tree sparrow
		<i>Amphispiza bilineata</i>	Black-throated sparrow
		<i>Calamospiza melanocorys</i>	Lark bunting
		<i>Ammodramus leconteii</i>	Le Conte's sparrow
		<i>Ammodramus nelsoni</i>	Nelson's sharp-tailed sparrow

Order	Family	Scientific name	Common name
		<i>Passerella iliaca</i>	Fox sparrow
		<i>Melospiza lincolni</i>	Lincoln's sparrow
		<i>Zonotrichia albicollis</i>	White-throated sparrow
		<i>Zonotrichia atricapilla</i>	Golden-crowned sparrow
		<i>Zonotrichia leucophrys</i>	White-crowned sparrow
		<i>Zonotrichia querula</i>	Harris's sparrow
		<i>Junco hyemalis</i>	Dark-eyed junco
		<i>Calcarius lapponicus</i>	Lapland longspur
		<i>Calcarius pictus</i>	Smith's longspur
		<i>Calcarius ornatus</i>	Chestnut-collared longspur
		<i>Plectrophenax nivalis</i>	Snow bunting
		<i>Euphagus carolinus</i>	Rusty blackbird
		<i>Euphagus cyanocephalus</i>	Brewer's blackbird
		<i>Icterus bullockii</i>	Bullock's oriole
	Fringillidae	<i>Leucosticte tephrocotis</i>	Gray-crowned rosy finch
		<i>Pinicola enucleator</i>	Pine grosbeak
		<i>Carpodacus purpureus</i>	Purple finch
		<i>Loxia leucoptera</i>	White-winged crossbill
		<i>Carduelis flammea</i>	Common redpoll
		<i>Carduelis hornemanni</i>	Hoary redpoll
		<i>Coccothraustes vespertinus</i>	Evening grosbeak
		<i>Carduelis psaltria</i>	*Lesser goldfinch

*Indicates species in SCWCP Appendix 2 (Zohrer 2005), but not in Kent and Dinsmore (1996).

**Indicates species in Kent and Dinsmore (1996), but not in GAP (Kane et al. 2003).

^AIowa endangered species.

^BIowa threatened species.

^CIowa species of special concern.

^EFederally endangered species.

Iowa Butterflies (Opler and Krezik 1984, Zohrer et al. 2005)

These animals represent 70 genera, 6 families, and 1 order.

Order	Family	Scientific name	Common name
Lepidoptera	Hesperiidae	<i>Achalarus lyciades</i>	Hoary edge
		<i>Amblyscirtes hegon</i>	^C Pepper and salt skipper
		<i>Amblyscirtes vialis</i>	Common roadside skipper
		<i>Ancyloxypha numitor</i>	Least skipper
		<i>Atalopedes campestris</i>	Sachem
		<i>Atrytone arogos</i>	^C Arogos skipper
		<i>Atrytone logan</i>	Delaware skipper
		<i>Atrytonopsis hianna</i>	^C Dusted skipper
		<i>Epargyreus clarus</i>	Silver-spotted skipper
		<i>Erynnis baptisiae</i>	^C Wild indigo duskywing
		<i>Erynnis brizo</i>	^C Sleepy duskywing
		<i>Erynnis horatius</i>	Horace's duskywing
		<i>Erynnis icelus</i>	^C Dreamy duskywing
		<i>Erynnis juvenalis</i>	Juvenal's duskywing

Order	Family	Scientific name	Common name
		<i>Erynnis lucilius</i>	^C Columbine duskywing
		<i>Erynnis martialis</i>	Mottled duskywing
		<i>Erynnis persius</i>	Persius duskywing
		<i>Euphyes bimacula</i>	^C Two-spotted skipper
		<i>Euphyes conspicua</i>	Black dash
		<i>Euphyes dion</i>	^C Sedge (or dion) skipper
		<i>Euphyes vestris</i>	Dun skipper
		<i>Hesperia dacotae</i>	^{A E} Dakota skipper
		<i>Hesperia leonardus</i>	^C Leonard's skipper
		<i>Hesperia ottoe</i>	^C Ottoe skipper
		<i>Hesperia uncas</i>	Uncas skipper
		<i>Hylephila phyleus</i>	Fiery skipper
		<i>Lerodea eufala</i>	Eufala skipper
		<i>Oarisma powesheik</i>	^B Powesheik skipperling
		<i>Pholisora catullus</i>	Common sooty wing
		<i>Poanes hobomok</i>	Hobomok skipper
		<i>Poanes massasoit</i>	^B Mulberry wing
		<i>Poanes viator</i>	^C Broad-winged skipper
		<i>Poanes zabulon</i>	^C Zabulon skipper
		<i>Polites mystic</i>	Long dash
		<i>Polites origenes</i>	Crossline skipper
		<i>Polites peckius</i>	Peck's skipper
		<i>Polites themistocles</i>	Tawny-edged skipper
		<i>Pompeius verna</i>	Little glassywing
		<i>Problema byssus</i>	^B Byssus skipper
		<i>Pyrgus communis</i>	Common checkered skipper
		<i>Staphylus hayhurstii</i>	Southern sooty wing
		<i>Thorybes bathyllus</i>	Southern cloudwing
		<i>Thorybes pylades</i>	Northern cloudwing
		<i>Thymelicus lineola</i>	European skipper
		<i>Wallengrenia egerement</i>	Northern broken-dash
	Libytheidae	<i>Libytheana carinenta</i>	American snout
	Lycaenidae	<i>Calephelis muticum</i>	^C Swamp metalmark
		<i>Callophrys gryneus</i>	Juniper hairstreak
		<i>Callpohrys henrici</i>	Henry's elfin
		<i>Celastrina neglecta</i>	Summer azure
		<i>Everes comyntas</i>	Eastern-tailed blue
		<i>Feniseca tarquinius</i>	Harvester
		<i>Glaucopsyche lygdamus</i>	^B Silvery blue
		<i>Hemiargus isola</i>	Reakirt's blue
		<i>Leptotes marina</i>	Marine blue
		<i>Lycaeides melissa</i>	Melissa blue
		<i>Lycaena dione</i>	Gray copper
		<i>Lycaena helloides</i>	^C Purplish copper
		<i>Lycaena hyllus</i>	Bronze copper

Order	Family	Scientific name	Common name
		<i>Lycaena phlaeas</i>	American copper
		<i>Parrhasius m-album</i>	White M hairstreak
		<i>Plebejus saepiolus</i>	Greenish blue
		<i>Satyrium acadica</i>	^c Acadian hairstreak
		<i>Satyrium calanus</i>	Banded hairstreak
		<i>Satyrium caryaevorum</i>	^c Hickory hairstreak
		<i>Satyrium edwardsii</i>	^c Edward's hairstreak
		<i>Satyrium liparops</i>	^c Striped hairstreak
		<i>Satyrium titus</i>	Coral hairstreak
		<i>Strymon melinus</i>	Gray hairstreak
	Nymphalidae	<i>Agraulis vanillae</i>	Gulf fritillary
		<i>Anaea andria</i>	Goatweed leafwing
		<i>Asterocampa celtis</i>	Hackberry emperor
		<i>Asterocampa clyton</i>	Tawny emperor
		<i>Boloria bellona</i>	Meadow fritillary
		<i>Boloria selene</i>	Silver-bordered fritillary
		<i>Cercyonis pegala</i>	Common wood nymph
		<i>Chlosyne gorgone</i>	Gorgone checkerspot
		<i>Chlosyne nycteis</i>	Silvery checkerspot
		<i>Coenonympha tullia</i>	^A Common ringlet
		<i>Danaus plexippus</i>	Monarch or Queen
		<i>Enodia anthedon</i>	Northern pearly eye
		<i>Euphydryas phaeton</i>	^B Baltimore checkerspot
		<i>Euptoieta claudia</i>	Variegated fritillary
		<i>Junonia coenia</i>	Common buckeye
		<i>Limenitis archippus</i>	Viceroy
		<i>Limenitis arthemis</i>	Red-spotted purple
		<i>Megisto cymela</i>	Little wood satyr
		<i>Nymphalis antiopa</i>	Mourning cloak
		<i>Nymphalis milberti</i>	Milbert's tortoiseshell
		<i>Nymphalis vaualbum</i>	Compton tortoiseshell
		<i>Phyciodes tharos</i>	Pearl crescent
		<i>Phycoides batesii</i>	Tawny crescent
		<i>Polygonia comma</i>	Eastern comma
		<i>Polygonia interrogationis</i>	Question mark
		<i>Polygonia progne</i>	Gray comma
		<i>Satyroides eurydice</i>	Eyed brown
		<i>Speyeria aphrodite</i>	Aphrodite fritillary
		<i>Speyeria cybele</i>	Great spangled fritillary
		<i>Speyeria idalia</i>	^c Regal flitillary
		<i>Vanessa atalanta</i>	Red admiral
		<i>Vanessa cardui</i>	Painted lady
		<i>Vanessa virginiensis</i>	American lady
	Papilionidae	<i>Battus philenor</i>	^c Pipevine swallowtail
		<i>Eurytides marcellus</i>	^c Zebra swallowtail

Order	Family	Scientific name	Common name
		<i>Papilio cresphontes</i>	Giant swallowtail
		<i>Papilio glaucus</i>	Eastern tiger swallowtail
		<i>Papilio polyxenes</i>	Black swallowtail
		<i>Papilio troilus</i>	Spicebush swallowtail
	Pieridae	<i>Colias eurytheme</i>	Orange sulphur
		<i>Colias philodice</i>	Clouded sulphur
		<i>Euchloe olympia</i>	^C Olympia white (or marble)
		<i>Eurema lisa</i>	Little yellow
		<i>Eurema nicippe</i>	Sleepy orange
		<i>Nathalis iole</i>	Dainty orange
		<i>Phoebis sennae</i>	Cloudless sulphur
		<i>Pieris rapae</i>	Cabbage white
		<i>Pontia protodice</i>	Checkered white
		<i>Zerene cesonia</i>	Southern dogface

^AIowa endangered species

^BIowa threatened species

^CIowa species of special concern.

^EFederally endangered

Iowa Damselflies and Dragonflies

These animals represent 35 genera, 8 families, and 1 order. Odonata (suborder Anisoptera) are dragonflies, Odonata (suborder Zygoptera) are damselflies.

Order	Family	Scientific name	Common name
Odonata	Aeshnidae	<i>Aeshna canadensis</i>	Canada darner
Suborder		<i>Aeshna constricta</i>	Lance-tipped darner
Anisoptera		<i>Aeshna interrupta</i>	Variable darner
		<i>Aeshna multicolor</i>	Blue-eyed darner
		<i>Aeshna mutata</i>	Spatterdock darner
		<i>Aeshna tuberculifera</i>	Black-tipped darner
		<i>Aeshna unbrosa</i>	Shadow darner
		<i>Aeshna verticalis</i>	Green striped darner
		<i>Aeshna verticalis</i>	Green-striped darner
		<i>Anax junius</i>	Common green darner
		<i>Boyeria vinosa</i>	Fawn darner
		<i>Epiaeschna heros</i>	Swamp darner
		<i>Nasiaeschna pentacantha</i>	Cyrano darner
	Corduliidae	<i>Epithea cynosura</i>	Common baskettail
		<i>Epithea princeps</i>	Prince baskettail
		<i>Neurocordulia molesta</i>	Smoky shadowdragon
		<i>Neurocordulia yamaskanensis</i>	Stygian shadowdragon
		<i>Somatochlora ensigera</i>	Plains emerald
		<i>Somatochlora linearis</i>	Mocha emerald
	Gomphidae	<i>Arigomphus cornutus</i>	Horned clubtail
		<i>Arigomphus submedianus</i>	Jade clubtail
		<i>Dromogomphus spoliatus</i>	Flag-tailed spinyleg
		<i>Gomphus externus</i>	Plains clubtail

Order	Family	Scientific name	Common name
		<i>Gomphus fraternus</i>	Midland clubtail
		<i>Gomphus graslinellus</i>	Pronghorn clubtail
		<i>Gomphus militaris</i>	Sulpher-tipped clubtail
		<i>Gomphus quadricolor</i>	Rapids clubtail
		<i>Gomphus vastus</i>	Cobra clubtail
		<i>Ophiogomphus rupinsulensis</i>	Rusty snaketail
		<i>Ophiogomphus smithi</i>	Sioux snaketail
		<i>Progomphus obsurus</i>	Common sanddragon
		<i>Stylurus amnicola</i>	Riverine clubtail
		<i>Stylurus intricatus</i>	Brimstone clubtail
		<i>Stylurus notatus</i>	Elusive clubtail
		<i>Stylurus plagiatu</i> s	Russet-tipped clubtail
		<i>Stylurus spiniceps</i>	Arrow clubtail
	Libellulidae	<i>Celithemis elisa</i>	Calico pennant
		<i>Celithemis eponina</i>	Halloween pennant
		<i>Erythemis simplicicollis</i>	Eastern pondhawk
		<i>Leucorrhinia intacta</i>	Dot-tailed whiteface
		<i>Libellula cyanea</i>	Spangled skimmer
		<i>Libellula incesta</i>	Slaty skimmer
		<i>Libellula luctuosa</i>	Widow skimmer
		<i>Plathemis lydia</i>	Common whitetail
		<i>Libellula pulchella</i>	Twelve-spotted skimmer
		<i>Libellula quadrimaculata</i>	Four-spotted skimmer
		<i>Pachydiplax longipennis</i>	Blue dasher
		<i>Pantela flavescens</i>	Wandering glider
		<i>Pantela hymenaea</i>	Spot-winged glider
		<i>Perithemis tenera</i>	Eastern amberwing
		<i>Sympetrum ambiguum</i>	Blue-faced meadowhawk
		<i>Sympetrum corruptum</i>	Variegated meadowhawk
		<i>Sympetrum costiferum</i>	Saffron-winged meadowhawk
		<i>Sympetrum danae</i>	Black meadowhawk
		<i>Sympetrum internum</i>	Cherry-faced meadowhawk
		<i>Sympetrum obtrusum</i>	White-faced meadowhawk
		<i>Sympetrum occidentale</i>	Western meadowhawk
		<i>Sympetrum rubicundulum</i>	Ruby meadowhawk
		<i>Sympetrum vicinum</i>	Yellow-legged meadowhawk
		<i>Tamea carolina</i>	Carolina saddlebags
		<i>Tamea lacerate</i>	Black saddlebags
		<i>Tamea onusta</i>	Red saddlebags
	Macromiidae	<i>Macromia illinoiensis</i>	Illinois river cruiser
		<i>Macromia taeniolata</i>	Royal river cruiser
Odonata	Calopterygidae	<i>Calopteryx aequabilis</i>	River jewelwing
Suborder		<i>Calopteryx maculata</i>	Ebony jewelwing
Zygoptera		<i>Hetaerina titia</i>	Smoky rubyspot
		<i>Hetaeruba americana</i>	American rubyspot

Order	Family	Scientific name	Common name
	Coenagrionidae	<i>Amphiogrion saucium</i>	Eastern red damsel
		<i>Argia alberta</i>	Paiute dancer
		<i>Argia apicalis</i>	Blue-fronted dancer
		<i>Argia emma</i>	Emma's dancer
		<i>Argia fumipennis</i>	Variable dancer
		<i>Argia moesta</i>	Powdered dancer
		<i>Argia plana</i>	Springwater dancer
		<i>Argia tibialis</i>	Blue-tipped dancer
		<i>Coenagrion angulatum</i>	Prairie bluet
		<i>Coenagrion resolutum</i>	Taiga bluet
		<i>Enallagma anna</i>	River bluet
		<i>Enallagma antennatum</i>	Rainbow bluet
		<i>Enallagma aspersum</i>	Azure bluet
		<i>Enallagma basidens</i>	Double-striped bluet
		<i>Enallagma boreale</i>	Boreal bluet
		<i>Enallagma caruncularum</i>	Tulle bluet
		<i>Enallagma civile</i>	Familiar bluet
		<i>Enallagma clausum</i>	Alkali bluet
		<i>Enallagma cyathigerium</i>	Northern bluet
		<i>Enallagma erbiium</i>	Marsh bluet
		<i>Enallagma exulans</i>	Stream bluet
		<i>Enallagma geminatum</i>	Skimming bluet
		<i>Enallagma hageni</i>	Hagen's bluet
		<i>Enallagma signatum</i>	Orange bluet
		<i>Enallagma traviatum</i>	Slender bluet
		<i>Enallagma vesperum</i>	Vesper bluet
		<i>Ischnura hastata</i>	Citrine forktail
		<i>Ischnura posita</i>	Fragile forktail
		<i>Ischnura verticalis</i>	Eastern forktail
		<i>Nehalennia irene</i>	Sedge sprite
	Lestidae	<i>Archilestes grandis</i>	Great spreadwing
		<i>Lestes congener</i>	Spotted spreadwing
		<i>Lestes disjunctus</i>	Common spreadwing
		<i>Lestes dryas</i>	Emerald spreadwing
		<i>Lestes eurinus</i>	Amber-winged spreadwing
		<i>Lestes inaequalis</i>	Elegant spreadwing
		<i>Lestes forcipatus</i>	Sweetflag spreadwing
		<i>Lestes rectangularis</i>	Slender spreadwing
		<i>Lestes unguiculatus</i>	Lyre-tipped spreadwing

At this time, no odonates are recognized as T&E species in Iowa.

Iowa Bumble Bees

These animals represent 1 genera, 1 family, and 1 order. Hymenoptera.

Order	Family	Scientific Name	Common Name
Hymenoptera	Apidea	<i>Bombus affinis</i>	^E Rusty Patched Bumble Bee
		<i>Bombus auricomus</i>	Black and Gold Bumble Bee
		<i>Bombus bimaculatus</i>	Two-spotted Bumble Bee
		<i>Bombus bohemicus</i>	Ashton's Cuckoo Bumble Bee
		<i>Bombus borealis</i>	Northern Amber Bumble Bee
		<i>Bombus citrinus</i>	Lemon Cuckoo Bumble Bee
		<i>Bombus fervidus</i>	Yellow Bumble Bee
		<i>Bombus fraternus</i>	Southern Plains Bumble Bee
		<i>Bombus griseocollis</i>	Brown-belted Bumble Bee
		<i>Bombus impatiens</i>	Common Eastern Bumble Bee
		<i>Bombus pensylvanicus</i>	American Bumble Bee
		<i>Bombus ternarius</i>	Tri-colored Bumble Bee
		<i>Bombus vagans</i>	Half-black Bumble Bee
		<i>Bombus variabilis</i>	Variable Cuckoo Bumble Bee
		<i>Bombus rufocinctus</i>	Red-belted Bumble Bee
		<i>Bombus frigidus</i>	Frigid Bumble Bee
		<i>Bombus morrisoni</i>	Morrison Bumble Bee
		<i>Bombus perplexus</i>	Confusing Bumble Bee

^EFederally endangered species

Iowa Fish (From Zohrer et al. 2005)

These animals represent 71 genera, 26 families, and 16 orders.

Order	Family	Scientific name	Common name
Petromyzontiformes	Petromyzontidae	<i>Ichthyomyzon castaneus</i>	^B Chestnut lamprey
		<i>Ichthyomyzon fossor</i>	Northern brook lamprey
		<i>Ichthyomyzon unicuspis</i>	Silver lamprey
		<i>Lampetra appendix</i>	^B American brook lamprey
Acipenseriformes	Acipenseridae	<i>Acipenser fulvescens</i>	^A Lake sturgeon
		<i>Scaphirhynchus albus</i>	^A ^E Pallid sturgeon
		<i>Scaphirhynchus platyrhynchus</i>	Shovelnose sturgeon
	Polyodontidae	<i>Polyodon spathula</i>	Paddlefish
Amiiformes	Amiidae	<i>Amia calva</i>	Bowfin
Lepisosteiformes	Lepisosteidae	<i>Lepirosteus platostomus</i>	Shortnose gar
		<i>Lepisosteus osseus</i>	Longnose gar
		<i>Lepisosteus oculatus</i>	Spotted gar
Anguilliformes	Anguillidae	<i>Anguilla rostrata</i>	American eel
Clupeiformes	Clupeidae	<i>Dorosoma cepedianum</i>	Gizzard shad
		<i>Alosa alabamae</i>	Alabama shad
		<i>Alosa chrysochloris</i>	Skipjack herring
Osteoglossiformes	Hiodontidae	<i>Hiodon tergisus</i>	Mooneye
		<i>Hiodon alosoides</i>	Goldeye
Salmoniformes	Salmonidae	<i>Oncorhynchus mykiss</i>	Rainbow trout
		<i>Salmo trutta</i>	Brown trout

Order	Family	Scientific name	Common name
		<i>Salvelinus fontinalis</i>	Brook trout
	Esocidae	<i>Esox lucius</i>	Northern pike
		<i>Esox masquinongy</i>	Muskellunge
		<i>Esox americanus</i>	^B Grass pickerel
	Umbridae	<i>Umbra limi</i>	Central mudminnow
Cypriniformes	Cyprinidae	<i>Campostoma anomalum</i>	Central stoneroller
		<i>Campostoma oligolepis</i>	Largescale stoneroller
		<i>Carassius auratus</i>	Goldfish
		<i>Ctenopharyngodon idella</i>	Grass carp
		<i>Cyprinus carpio</i>	Common carp
		<i>Hypophthalmichthys nobilis</i>	Bighead carp
		<i>Hypophthalmichthys molitrix</i>	Silver carp
		<i>Phenacobius mirabelis</i>	Suckermouth minnow
		<i>Pimephales notatus</i>	Bluntnose minnow
		<i>Pimephales promelas</i>	Fathead minnow
		<i>Pimephales vigilax</i>	Bullhead minor
		<i>Hybognathus agryritis</i>	Western silvery minnow
		<i>Hybognathus nuchalis</i>	Mississippi silvery minnow
		<i>Hybognathus hankinsoni</i>	Brassy minnow
		<i>Hybognathus placitus</i>	Plains minnow
		<i>Semotilus atromaculatus</i>	Creek chub
		<i>Couesius plumbeus</i>	Lake chub
		<i>Macrhybopsis aestivalis</i>	Speckled chub
		<i>Macrhybopsis gelida</i>	Sturgeon chub
		<i>Platygobio gracillis</i>	Flathead chub
		<i>Macrybopsis meeki</i>	Sicklefin chub
		<i>Macrybopsis storeriana</i>	Silver chub
		<i>Erimytax x-punctatus</i>	Gravel chub
		<i>Nocomis biguttatus</i>	Horneyhead chub
		<i>Notemigonus crysoleucas</i>	Golden shiner
		<i>*Hybopsis amnis</i>	<i>*Pallid shiner</i>
		<i>Opsopoeodus emiliae</i>	^C Pugnose minnow
		<i>Notropis anogenus</i>	^A Pugnose shiner
		<i>Notropis atherinoides</i>	Emerald shiner
		<i>Notropis blennioides</i>	River shiner
		<i>Notropis bethunei</i>	Ghost shiner
		<i>Notropis chalybaeus</i>	Ironcolor shiner
		<i>Luxilus cornutus</i>	Common shiner
		<i>Notropis dorsalis</i>	Bigmouth shiner
		<i>Notropis heterodon</i>	Blackchin shiner
		<i>Notropis heterolepis</i>	^B Blacknose shiner
		<i>Notropis hudsonius</i>	Spottail shiner
		<i>Cyprinella lutrensis</i>	Red shiner
		<i>Notropis nubilus</i>	Ozark minnow
		<i>Notropis rubellus</i>	Rosyface shiner

Order	Family	Scientific name	Common name
		<i>Notropis shumardi</i>	Silverband shiner
		<i>Cyprinella spiloptera</i>	Spotfin shiner
		<i>Notropis texanus</i>	^A Weed shiner
		<i>Notropis Topeka</i>	^{B E} Topeka shiner
		<i>Lythrurus umbratilis</i>	Redfin shiner
		<i>Notropis volucellus</i>	Mimic shiner
		<i>Notropis wickliffi</i>	Channel shiner
		<i>Rhinichthys atratulus</i>	Blacknose dace
		<i>Rhinichthys cataractae</i>	Longnose dace
		<i>Margariscus margarita</i>	^A Pearl dace
		<i>Phoxinus erythrogaster</i>	Southern redbelly dace
		<i>Clinostomus elongatus</i>	Redside dace
	Catostomidae	<i>Cycleptus elongates</i>	Blue sucker
		<i>Erimyzon sucetta</i>	Lake chubsucker
		<i>Ictiobus cyprinellus</i>	Bigmouth buffalo
		<i>Ictiobus bubalus</i>	Smallmouth buffalo
		<i>Ictiobus niger</i>	Black buffalo
		<i>Carpionodes cyprinus</i>	Quillback
		<i>Carpionodes carpio</i>	River carpsucker
		<i>Carpionodes velifer</i>	Highfin carpsucker
		<i>Moxostoma duquesnei</i>	^B Black redbhorse
		<i>Moxostoma erythrurum</i>	Golden redbhorse
		<i>Moxostoma anisurum</i>	Silver redbhorse
		<i>Moxostoma macrolepidotum</i>	Shorthead redbhorse
		<i>Moxostoma carinatum</i>	River redbhorse
		<i>Moxostoma valenciennesi</i>	Greater redbhorse
		<i>Minytrema melanops</i>	Spotted sucker
		<i>Hypentelium nigricans</i>	Northern hog sucker
		<i>Catostomus commersoni</i>	White sucker
Siluriformes	Ictaluridae	<i>Ictalurus punctatus</i>	Channel catfish
		<i>Pylodictus olivaris</i>	Flathead catfish
		<i>Ictalurus furcatus</i>	Blue catfish
		<i>Ameiurus melas</i>	Black bullhead
		<i>Ameiurus natalis</i>	Yellow bullhead
		<i>Ameiurus nebulosus</i>	Brown bullhead
		<i>Noturus exilis</i>	Slender madtom
		<i>Noturus gyrinus</i>	Tadpole madtom
		<i>Noturus flavus</i>	Stonecat
		<i>Noturus gyrinus</i>	^A Freckled madtom
Percopsiformes	Aphredoderidae	<i>Aphredoderus sayanus</i>	^C Pirate perch
	Percopsidae	<i>Percopsis omiscomycus</i>	Trout perch
Gadiformes	Gadidae	<i>Lota lota</i>	^B Burbot
Atheriniformes	Cyprinodontidae	<i>Fundulus diaphanous</i>	Banded killifish
		<i>Fundulus notatus</i>	Blackstripe topminnow
		<i>Fundulus dispar</i>	Starhead topminnow

Order	Family	Scientific name	Common name
		<i>Fundulus sciadicus</i>	Plains topminnow
	Poeciliidae	<i>Gambusia affinis</i>	Western mosquito fish
	Atherinidae	<i>Labidesthes sicculus</i>	Brook silverside
Gasterosteiformes	Gasterosteidae	<i>Culaea inconstans</i>	Brook stickleback
Scorpaeniformes	Cottidae	<i>Cottus bairdi</i>	Mottled sulpin
		<i>Cottus cognatus</i>	Slimy sculpin
Perciformes	Percichthyidae	<i>Morone chrysops</i>	White bass
		<i>Morone mississippiensis</i>	Yellow bass
		<i>Morone saxatilis</i>	Striped bass
	Centrarchidae	<i>Micropterus salmoides</i>	Largemouth bass
		<i>Micropterus dolomieu</i>	Smallmouth bass
		<i>Micropterus punctulatus</i>	Spotted bass
		<i>Pomoxis annularis</i>	White crappie
		<i>Pomoxis nigromaculatus</i>	Black crappie
		<i>Ambloplites rupestris</i>	Rock bass
		<i>Lepomis macrochirus</i>	Bluegill
		<i>Lepomis microlophus</i>	Redear sunfish
		<i>Lepomis gulosus</i>	Warmouth
		<i>Lepomis cyanellus</i>	Green sunfish
		<i>Lepomis gibbosus</i>	Pumpkinseed
		<i>Lepomis humilis</i>	Orangespotted sunfish
		<i>Lepomis megalotis</i>	Longear sunfish
	Percidae	<i>Perca flavescens</i>	Yellow perch
		**Sander vitreus	**Walleye
		**Sander canadensis	**Sauger
		<i>Percina phoxocephala</i>	Slenderhead darter
		<i>Percina maculate</i>	Blackside darter
		<i>Percina evides</i>	Gilt darter
		<i>Percina shumardi</i>	River darter
		<i>Percina caprodes</i>	Northern logperch
		***Crystallaria asprella	***Crystal darter
		<i>Ammocrypta clara</i>	^B Western sand darter
		<i>Etheostoma zonale</i>	Banded darter
		<i>Etheostoma nigrum</i>	Johnny darter
		<i>Etheostoma chlorosomum</i>	^A Bluntnose darter
		<i>Etheostoma asprigene</i>	Mud darter
		<i>Etheostoma caeruleum</i>	Rainbow darter
		<i>Etheostoma exile</i>	Iowa darter
		<i>Etheostoma spectabile</i>	^B Orangethroat darter
		<i>Etheostoma flabellare</i>	Fantail darter
		<i>Etheostoma microperca</i>	^A Least darter
	Sciaenidae	<i>Aplodinotus grunniens</i>	Freshwater drum

*Genus listed as *Notropis* in Robbins et al. (1991) and *Hybopsis* in Zohrer (2005).

**Genus listed as *Stizostedion* in Robbins et al. (1991) and *Sander* in Zohrer (2005).

***Genus listed as *Ammocrypta* in Robbins et al. (1991) and *Crystallaria* in Zohrer (2005).

^AIowa endangered species.

^BIowa threatened species.

^CIowa species of special concern.

^EFederally endangered species.

Iowa Mussels (Zohrer et al. 2005)

These animals represent 35 genera, 5 families, and 2 orders.

Order	Family	Scientific name	Common name
Unionoida	Unionidae	<i>Actinonaias ligmentina</i>	Mucket
		<i>Alasmidonta marginata</i>	Elktoe
		<i>Alasmidonta viridis</i>	^A Slippershell
		<i>Amblema plicata</i>	Three ridge
		<i>Anodonta suborbiculata</i>	Flat floater
		<i>Anodontoides ferussacianus</i>	^B Cylinder
		<i>Arcidens confragosus</i>	Rock pocketbook
		<i>Cyclonaias tuberculata</i>	^B Purple pimpleback
		<i>Ellipsaria lineolata</i>	^B Butterfly
		<i>Elliptio crassidens</i>	Elephant ear
		<i>Elliptio dilatata</i>	Spike
		<i>Epioblasma triquetra</i>	Snuffbox
		<i>Fusconaia ebena</i>	Ebony shell
		<i>Fusconaia flava</i>	Wabash pigtoe
		<i>Fusconaia ozarkensis</i>	^A Ozark pigtoe
		<i>Lampsilis cardium</i>	Plain pocketbook
		<i>Lampsilis higginsii</i>	^{A E} Higgin's eye pearlymussel
		<i>Lampsilis siliquoidea</i>	Fatmucket
		<i>Lampsilis teres anodontoides</i>	^A Yellow sandshell
		<i>Lampsilis teres teres</i>	^A Slough sandshell
		<i>Lasmigona camplanata</i>	White heelsplitter
		<i>Lasmigona compressa</i>	^B Creek heelsplitter
		<i>Lasmigona costata</i>	Fluted shell
		<i>Leptodea fragilis</i>	Fragile papershell
		<i>Leptodea leptodon</i>	^E Scaleshell
		<i>Ligumia recta</i>	Black sandshell
		<i>Ligumia subrostrata</i>	Pondmussel
		<i>Megalonaias nervosa</i>	Washboard
		<i>Obliquaria reflexa</i>	Threehorn wartback
		<i>Obovaria olivaria</i>	Hickorynut
		<i>Plethobasus cyphus</i>	^A Bullhead (Sheepnose)
		<i>Pleurobema rubrum</i>	Pyramid pigtoe
		<i>Pleurobema sintoxia</i>	^A Round (Ohio River) pigtoe
		<i>Potamilus alatus</i>	Pink heelsplitter
		<i>Potamilus capax</i>	^E Fat pockebook
		<i>Potamilus ohioensis</i>	Pink papershell
		<i>Pyganodon grandis</i>	Giant floater
		<i>Quadrula fragosa</i>	Winged mapleleaf

Checklist of Iowa mussels (Zohrer et al. 2005). These animals represent 35 genera, 5 families, and 2 orders.

Order	Family	Scientific name	Common name
		<i>Quadrula metanerva</i>	Monkeyface
		<i>Quadrula nodulata</i>	Wartyback
		<i>Quadrula pustulosa</i>	Pimpleback
		<i>Quadrula quadrula</i>	^E Mapleleaf
		<i>Simpsonaias ambiua</i>	Salamander mussel
		<i>Strophitus undulates</i>	^B Strange floater (Squawfoot)
		<i>Toxolasma parvus</i>	Lilliput
		<i>Tritogonia verrucosa</i>	^A Pistolgrip or Buckhorn
		<i>Truncilla donaciformis</i>	Fawnsfoot
		<i>Truncilla truncata</i>	Deertoe
		<i>Unio merus tetralasmus</i>	Pondhorn
		<i>Utterbackia imbecillis</i>	Paper pondshell
		<i>Venusta concha ellipsiformis</i>	^B Ellipse
	Margaritiferidae	<i>Cumberlandia monodonta</i>	^A Spectacle case
Veneroida	Sphaeriidae	<i>Musculium sp. Pisidium sp. Sphaerium sp.</i>	Fingernail clams
	Corbinulidae	<i>Corbicula fluminea</i>	Asian clam
	Dreissenidae	<i>Dreissena polymorpha</i>	Zebra mussel

^AIowa endangered species.

^BIowa threatened species.

^EFederally endangered species.

Appendix 2: Turtle Marking Codes & Herpetofauna Photo Voucher Guidelines

Marking Codes for Turtles

2 scutes marked = 120 individual marks:

AB	BC	CH	HI	IJ	JK	KL	LM	MN	NO	OP	PQ	QV	VW	WX
AC	BH	CI	HJ	IK	JL	KM	LN	MO	NP	OQ	PV	QW	VX	
AH	BI	CJ	HK	IL	JM	KN	LO	MP	NQ	OV	PW	QX		
AI	BJ	CK	HL	IM	JN	KO	LP	MQ	NV	OW	PX			
AJ	BK	CL	HM	IN	JO	KP	LQ	MV	NW	OX				
AK	BL	CM	HN	IO	JP	KQ	LV	MW	NX					
AL	BM	CN	HO	IP	JQ	KV	LW	MX						
AM	BN	CO	HP	IQ	JV	KW	LX							
AN	BO	CP	HQ	IV	JW	KX								
AO	BP	CQ	HV	IW	JX									
AP	BQ	CV	HW	IX										
AQ	BV	CW	HX											
AV	BW	CX												
AW	BX													
AX														

Marking codes for turtles:

3 scutes marked = 560 individual marks:

ABC	ACW	AJK	ALW	AQW	BHP	BJX	BNO	CHM	CJQ	CMV	HIK	HKQ	HOP
ABH	ACX	AJL	ALX	AQX	BHQ	BKL	BNP	CHN	CJV	CMW	HIL	HKV	HOQ
ABI	AHI	AJM	AMN	AVW	BHV	BKM	BNQ	CHO	CJW	CMX	HIM	HKW	HOV
ABJ	AHJ	AJN	AMO	AVX	BHW	BKN	BNV	CHP	CJX	CNO	HIN	HKX	HOW
ABK	AHK	AJO	AMP	AWX	BHX	BKO	BNW	CHQ	CKL	CNP	HIO	HLM	HOX
ABL	AHL	AJP	AMQ	BCH	BIJ	BKP	BNX	CHV	CKM	CNQ	HIP	HLN	HPQ
ABM	AHM	AJQ	AMV	BCI	BIK	BKQ	BOP	CHW	CKN	CNV	HIQ	HLO	HPV
ABN	AHN	AJV	AMW	BCJ	BIL	BKV	BOQ	CHX	CKO	CNW	HIV	HLP	HPW
ABO	AHO	AJW	AMX	BCK	BIM	BKW	BOV	CIJ	CKP	CNX	HIW	HLQ	HPX
ABP	AHP	AJX	ANO	BCL	BIN	BKX	BOW	CIK	CKQ	COP	HIX	HLV	HQV
ABQ	AHQ	AKL	ANP	BCM	BIO	BLM	BOX	CIL	CKV	COQ	HJK	HLW	HQW
ABV	AHV	AKM	ANQ	BCN	BIP	BLN	BPQ	CIM	CKW	COV	HJL	HLX	HQX
ABW	AHW	AKN	ANV	BCO	BIQ	BLO	BPV	CIN	CKX	COW	HJM	HMN	HVW
ABX	AHX	AKO	ANW	BCP	BIV	BLP	BPW	CIO	CLM	COX	HJN	HMO	HVX
ACH	AIJ	AKP	ANX	BCQ	BIW	BLQ	BPX	CIP	CLN	CPQ	HJO	HMP	HWX
ACI	AIK	AKQ	AOP	BCV	BIX	BLV	BQV	CIQ	CLO	CPV	HJP	HMQ	IJK
ACJ	AIL	AKV	AOQ	BCW	BJK	BLW	BQW	CIV	CLP	CPW	HJQ	HMV	IJL
ACK	AIM	AKW	AOV	BCX	BJL	BLX	BQX	CIW	CLQ	CPX	HJV	HMW	IJM
ACL	AIN	AKX	AOW	BHI	BJM	BMN	BVW	CIX	CLV	CQV	HJW	HMX	IJN
ACM	AIO	ALM	AOX	BHJ	BJN	BMO	BVX	CJK	CLW	CQW	HJX	HNO	IJO
CAN	AIP	ALN	APQ	BHK	BJO	BMP	BWX	CJL	CLX	CQX	HKL	HNP	IJP
ACO	AIQ	ALO	APV	BHL	BJP	BMQ	CHI	CJM	CMN	CVW	HKM	HNQ	IJQ
ACP	AIV	ALP	APW	BHM	BJQ	BMV	CHJ	CJN	CMO	CVX	HKN	HNW	IJV
ACQ	AIW	ALQ	APX	BHN	BJV	BMW	CHK	CJO	CMP	CWX	HKO	HNW	IJW
ACV	AIX	ALV	AQV	BHO	BJW	BMX	CHL	CJP	CMQ	HIJ	HKP	HNX	IJX

Marking codes for turtles:

3 scutes marked = 560 individual marks (continued from previous page):

IKL	INP	JKQ	JOP	KMP	KWX	LQX	NOP	PQV
IKM	INQ	JKV	JOQ	KMQ	LMN	LVW	NOQ	PQW
IKN	INV	JKW	JOV	KMV	LMO	LVX	NOV	PQX
IKO	INW	JKX	JOW	KMW	LMP	LWX	NOW	PVW
IKP	INX	JLM	JOX	KMX	LMQ	MNO	NOX	PVX
IKQ	IOP	JLN	JPQ	KNO	LMV	MNP	NPQ	PWX
IKV	IOQ	JLO	JPV	KNP	LMW	MNQ	NPV	QVW
IKW	IOV	JLP	JPW	KNQ	LMX	MNV	NPW	QVX
IKX	IOW	JLQ	JPX	KNV	LNO	MNW	NPX	QWX
ILM	IOX	JLV	JQV	KNW	LNP	MNX	NQV	VWX
ILN	IPQ	JLW	JQW	KNX	LNQ	MOP	NQW	
ILO	IPV	JLX	JQX	KOP	LNW	MOQ	NQX	
ILP	IPW	JMN	JVW	KOQ	LNW	MOV	NVW	
ILQ	IPX	JMO	JVX	KOV	LNX	MOW	NVX	
ILV	IQV	JMP	JWX	KOQ	LOP	MOX	NWX	
ILW	IQW	JMQ	KLM	KOX	LOQ	MPQ	OPQ	
ILX	IQX	JMV	KLN	KPQ	LOV	MPV	OPV	
IMN	IVW	JMW	KLO	KPV	LOW	MPW	OPW	
IMO	IVX	JMX	KLP	KPW	LOX	MPX	OPX	
IMP	IWX	JNO	KLQ	KPX	LPQ	MQV	OQV	
IMQ	JKL	JNP	KLW	KQV	LPV	MQW	OQW	
IMV	JKM	JNQ	KLW	KQW	LPW	MQX	OQX	
IMW	JKN	JNV	KLX	KQX	LPX	MVW	OVW	
IMX	JKO	JNW	KMN	KVW	LQV	MVX	OVX	
INO	JKP	JNX	KMO	KVX	LQW	MWX	OWX	

Marking codes for turtles:

4 scutes marked = 1,799 individual marks:

ABCH	ABIJ	ABKP	ABNX	ACHV	ACKM	ACNQ	AHIP	AHLN	AHPQ	AIKQ	AIOP	AJLN	AJPQ
ABCI	ABIK	ABKQ	ABOP	ACHW	ACKN	ACNV	AHIQ	AHLO	AHPV	AIKV	AIOQ	AJLO	AJPV
ABCJ	ABIL	ABKV	ABOQ	ACHX	ACKO	ACNW	AHIV	AHLP	AHPW	AIKW	AIOV	AJLP	AJPW
ABCK	ABIM	ABKW	ABOV	ACIJ	ACKP	ACNX	AHIW	AHLQ	AHPX	AIKX	AIOW	AJLQ	AJPX
ABCL	ABIN	ABKX	ABOW	ACIK	ACKQ	ACOP	AHIX	AHLV	AHQV	AILM	AIOX	AJLV	AJQV
ABCM	ABIO	ABLM	ABOX	ACIL	ACKV	ACOQ	AHJK	AHLW	AHQW	AILN	AIPQ	AJLW	AJQW
ABCN	ABIP	ABLN	ABPQ	ACIM	ACKW	ACOV	AHJL	AHLX	AHQX	AILO	AIPV	AJLX	AJQX
ABCO	ABIQ	ABLO	ABPV	ACIN	ACKX	ACOW	AHJM	AHMN	AHVV	AILP	AIPW	AJMN	AJVV
ABCP	ABIV	ABLP	ABPW	ACIO	ACLM	ACOX	AHJN	AHMO	AHVX	AILQ	AIPX	AJMO	AJVV
ABCQ	ABIW	ABLQ	ABPX	ACIP	ACLN	ACPQ	AHJO	AHMP	AHWX	AILV	AIQV	AJMP	AJWX
ABCV	ABIX	ABLV	ABQV	ACIQ	ACLO	ACPV	AHJP	AHMQ	AIJK	AILW	AIQW	AJMQ	AKLM
ABCW	ABJK	ABLW	ABQW	ACIV	ACLP	ACPW	AHJQ	AHMV	AIJL	AILX	AIQX	AJMV	AKLN
ABCX	ABJL	ABLX	ABQX	ACIW	ACLQ	ACPX	AHJV	AHMW	AIJM	AIMN	AIVW	AJMW	AKLO
ABHI	ABJM	ABMN	ABVW	ACIX	ACLV	ACQV	AHJW	AHMX	AIJN	AIMO	AIVX	AJMX	AKLP
ABHJ	ABJN	ABMO	ABVX	ACJK	ACLW	ACQW	AHJX	AHNO	AIJO	AIMP	AIWX	AJNO	AKLQ
ABHK	ABJO	ABMP	ABWX	ACJL	ACLX	ACQX	AHKL	AHNP	AIJP	AIMQ	AJKL	AJNP	AKLV
ABHL	ABJP	ABMQ	ACHI	ACJM	ACMN	ACVW	AHKM	AHNQ	AIJQ	AIMV	AJKM	AJNQ	AKLW
ABHM	ABJQ	ABMV	ACHJ	ACJN	ACMO	ACVX	AHKN	AHNV	AIJV	AIMW	AJKN	AJNV	AKLX
ABHN	ABJV	ABMW	ACHK	ACJO	ACMP	ACWX	AHKO	AHNV	AIJW	AIMX	AJKO	AJNV	AKMN
ABHO	ABJW	ABMX	ACHL	ACJP	ACMQ	AHIJ	AHKP	AHNV	AIJX	AINO	AJKP	AJNV	AKMO
ABHP	ABJX	ABNO	ACHM	ACJQ	ACMV	AHIK	AHKQ	AHOP	AIKL	AINP	AJKQ	AJOP	AKMP
ABHQ	ABKL	ABNP	ACHN	ACJV	ACMW	AHIL	AHKV	AHOQ	AIKM	AINQ	AJKV	AJOQ	AKMQ
ABHV	ABKM	ABNQ	ACHO	ACJW	ACMX	AHIM	AHKW	AHOV	AIKN	AINV	AJKW	AJOV	AKMV
ABHW	ABKN	ABNV	ACHP	ACJX	ACNO	AHIN	AHKX	AHOW	AIKO	AINW	AJKX	AJOW	AKMW
ABHX	ABKO	ABNW	ACHQ	ACKL	ACNP	AHIO	AHLM	AHOX	AIKP	AINX	AJLM	AJOX	AKMX

Marking codes for turtles:

4 scutes marked = 1,799 individual marks (continued from previous page):

AKNO	ALNO	AMQO	ANWX	BCHP	BCKN	BCOP	BHJM	BHMQ	BIJN	BIMV	BJKP	VJOV	BKNO
AKNP	ALNP	AMOV	AOPQ	BCHQ	BCKO	BCOQ	BHJN	BHMQ	BIJO	BIMW	BJKQ	BJOW	BKNP
AKNQ	ALNQ	AMOW	AOPV	BCHV	BCKP	BCOV	BHJO	BHMQ	BIJP	BIMX	BJKV	BJOX	BKNQ
AKNV	ALNV	AMOX	AOPW	BCHW	BCKQ	BCOW	BHJP	BHMX	BIJQ	BINO	BJKW	BJPQ	BKNV
AKNW	ALNW	AMPQ	AOPX	BCHX	BCKV	BCOX	BHJQ	BHNO	BIJV	BINP	BJKX	BJPV	BKNW
AKNX	ALNX	AMPV	AOQV	BCIJ	BCKW	BCPQ	BHJV	BHNP	BIJW	BINQ	BJLM	BJPW	BKNX
AKOP	ALOP	AMPW	AOQW	BCIK	BCKX	BCPV	BHJW	BHNP	BIJX	BINV	BJLN	BJPX	BKOP
AKOQ	ALOQ	AMPX	AOQX	BCIL	BCLM	BCPW	BHJX	BHNP	BIKL	BINW	BJLO	BJQV	BKOQ
AKOV	ALOV	AMQV	AOVW	BCIM	BCLN	BCPX	BHKL	BHNP	BIKM	BINX	BJLP	BJQW	BKOV
AKOW	ALOW	AMQW	AOVX	BCIN	BCLO	BCQV	BHKM	BHNP	BIKN	BIOP	BJLQ	BJQX	BKOW
AKOX	ALOX	AMQX	AOWX	BCIO	BCLP	BCQW	BHKN	BHNP	BIKO	BIOQ	BJLV	BJVW	BKOX
AKPQ	ALPQ	AMVW	APQV	BCIP	BCLQ	BCQX	BHKO	BHOQ	BIKP	BIOV	BJLW	BJVX	BKPQ
AKPV	ALPV	AMVX	APQW	BCIQ	BCLV	BCVW	BHKP	BHOV	BIKQ	BIOW	BJLX	BJWX	BKPV
AKPW	ALPW	AMWX	APQX	BCIV	BCLW	BCVX	BHKQ	BHOW	BIKV	BIOX	BJMN	BKLM	BKPW
AKPX	ALPX	ANOP	APVW	BCIW	BCLX	BCWX	BHKV	BHOX	BIKW	BIPQ	BJMO	BKLN	BKPX
AKQV	ALQV	ANOQ	APVX	BCIX	BCMN	BHIJ	BHKW	BHPQ	BIKX	BIPV	BJMP	BKLO	BKQV
AKQW	ALQW	ANOV	APWX	BCJK	BCMO	BHIK	BHKX	BHPV	BILM	BIPW	BJMQ	BKLP	BKQW
AKQX	ALQX	ANOW	AQVW	BCJL	BCMP	BHIL	BHLM	BHPW	BILN	BIPX	BJMV	BKLQ	BKQX
AKVW	ALVW	ANOX	AQVX	BCJM	BCMQ	BHIM	BHLN	BHPX	BILO	BIQV	BJMW	BKLV	BKVW
AKVX	ALWX	ANPQ	AQWX	BCJN	BCMV	BHIN	BHLO	BHQV	BILP	BIQW	BJMX	BKLW	BKVX
AKWX	ALWX	ANPV	AVWX	BCJO	BCMW	BHIO	BHLP	BHQW	BILQ	BIQX	BJNO	BKLX	BKWX
ALMN	AMNO	ANPW	BCHI	BCJP	BCMX	BHIP	BHLQ	BHQX	BILV	BIVW	BJNP	BKMN	BLMN
ALMO	AMNP	ANPX	BCHJ	BCJQ	BCNO	BHIQ	BHLV	BHVV	BILW	BIVX	BJNQ	BKMO	BLMO
ALMP	AMNQ	ANQV	BCHK	BCJV	BCNP	BHIV	BHLW	BHVX	BILX	BIWX	BJNV	BKMP	BLMP
ALMQ	AMNV	ANQW	BCHL	BCJW	BCNQ	BHIW	BHLX	BHWX	BIMN	BJKL	BJNW	BKMQ	BLMQ
ALMV	AMNW	ANQX	BCHM	BCJX	BCNV	BHIX	BHMN	BIJK	BIMO	BJKM	BJNX	BKMV	BLMV
ALMW	AMNX	ANVW	BCHN	BCKL	BCNW	BHJK	BHMO	BIJL	BIMP	BJKN	BJOP	BKMW	BLMW
ALMX	AMOP	ANVX	BCHO	BCKM	BCNX	BHJL	BHMP	BIJM	BIMQ	BJKO	BJOQ	BKMX	BLMX

Marking codes for turtles:

4 scutes marked = 1,799 individual marks (continued from previous pages):

BLNO	BNOQ	BPVX	CHKM	CHNX	CIKN	CIOP	CJLQ	CJQX	CKOW	CLOW	CMQW	CPVX	HIKV
BLNP	BNOV	BPWX	CHKN	CHOP	CIKO	CIOQ	CJLV	CJVW	CKOX	CLOX	CMQX	CPWX	HIKW
BLNQ	BNOW	BQVW	CHKO	CHOQ	CIKP	CIOV	CJLW	CJVX	CKPQ	CLPQ	CMVW	CQVW	HIKX
BLNV	BNOX	BQVX	CHKP	CHOV	CIKQ	CIOW	CJLX	CJWX	CKPV	CLPV	CMVX	CQVX	HILM
BLNW	BNPQ	BQWX	CHKQ	CHOW	CIKV	CIOX	CJMN	CKLM	CKPW	CLPW	CMWX	CQWX	HILN
BLNX	BNPV	BVWX	CHKV	CHOX	CIKW	CIPQ	CJMO	CKLN	CKPX	CLPX	CNOP	CVWX	HILO
BLOP	BNPW	CHIJ	CHKW	CHPQ	CIKX	CIPV	CJMP	CKLO	CKQV	CLQV	CNOQ	CPVX	HILP
BLOQ	BNPX	CHIK	CHKX	CHPV	CILM	CIPW	CJMQ	CKLP	CKQW	CLQW	CNOV	CPWX	HILQ
BLOV	BNQV	CHIL	CHLM	CHPW	CILN	CIPX	CJMV	CKLQ	CKQX	CLQX	CNOW	CQVW	HILV
BLOW	BNQW	CHIM	CHLN	CHPX	CILO	CIQV	CJMW	CKLV	CKVW	CLVW	CNOX	CQVX	HILW
BLOX	BNQX	CHIN	CHLO	CHQV	CILP	CIQW	CJMX	CKLW	CKVX	CLVX	CNPQ	CQWX	HILX
BLPQ	BNVW	CHIO	CHLP	CHQW	CILQ	CIQX	CJNO	CKLX	CKWX	CLWX	CNPV	CVWX	HIMN
BLPV	BNVX	CHIP	CHLQ	CHQX	CILV	CIVW	CJNP	CKMN	CLMN	CMNO	CNPW	HIJK	HIMO
BLPW	BNWX	CHIQ	CHLV	CHVW	CILW	CIVX	CJNQ	CKMO	CLMO	CMNP	CNPX	HIJL	HIMP
BLPX	BOPQ	CHIV	CJLW	CHVX	CILX	CIWX	CJNV	CKMP	CLMP	CMNQ	CNQV	HIJM	HIMQ
BLQV	BOPV	CHIW	CHLX	CHWX	CIMN	CJKL	CJNW	CKMQ	CLMQ	CMNV	CNQW	HIJN	HIMV
BLQW	BOPW	CHIX	CHMN	CIJK	CIMO	CJKM	CJNX	CKMV	CLMV	CMNW	CNQX	HIJO	HIMW
BLQX	BOPX	CHJK	CHMO	CIJL	CIMP	CJKN	CJOP	CKMW	CLMW	CMNX	CNVW	HIJP	HIMX
BLVW	BOQV	CHJL	CHMP	CIJM	CIMQ	CJKO	CJOQ	CKMX	CLMX	CMOP	CNVX	HIJQ	HINO
BLVX	BOQW	CHJM	CHMQ	CIJN	CIMV	CJKP	CJOV	CKNO	CLNO	CMOQ	CNWX	HIJV	HINP
BLWX	BOQX	CHJN	CHMV	CIJO	CIMW	CJKQ	CJOW	CKNP	CLNP	CMOV	COPQ	HIJW	HINQ
BMNO	BOVW	CHJO	CHMW	CIJP	CIMX	CJKV	CJOX	CKNQ	CLNQ	CMOW	COPV	HIJX	HINV
BMNP	BOVX	CHJP	CHMX	CIJQ	CINO	CJKW	CJPQ	CKNV	CLNV	CMOX	COPW	HIKL	HINW
BMNQ	BOWX	CHJQ	CHNO	CIJV	CINP	CJKX	CJPV	CKNW	CLNW	CMPQ	COPX	HIKM	HINX
BMNV	BPQV	CHJV	CHNP	CIJW	CINQ	CJLM	CJPW	CKNX	CLNX	CMPV	CPQV	HIKN	HIOP
BMNW	BPQW	CHJW	CHNQ	CIJX	CINV	CJLN	CJPX	CKOP	CLOP	CMPW	CPQW	HIKO	HIOQ
BMNX	BPQX	CHJX	CHNV	CIKL	CINW	CJLO	CJQV	CKOQ	CLOQ	CMPX	CPQX	HIKP	HIOV
BNOP	BPVW	CHKL	CHNW	CIKM	CINX	CJLP	CJQW	CKOV	CLOV	CMQV	CPVW	HIKQ	HIOW

Marking codes for turtles:

4 scutes marked = 1,799 individual marks (continued from previous pages):

HIOX	HJMN	HKLM	HKPW	HLPW	HMWX	HPQX	IJMQ	IKLP	IKQW	ILQW	INOV	IPWX	JKOV
HIPQ	HJMO	HKLN	HKPX	HLPX	HNOP	HPVW	IJMV	IKLQ	IKQX	ILQX	INOW	IQVW	JKOW
HIPV	HJMP	HKLO	HKQV	HLQV	HNOQ	HPVX	IJMW	IKLV	IKVW	ILVW	INOX	IQVX	JKOX
HIPW	HJMQ	HKLP	HKQW	HLQW	HNOV	HPWX	IJMX	IKLW	IKVX	ILVX	INPQ	IQWX	JKPQ
HIPX	HJMV	HKLQ	HKQX	HLQX	HNOW	HQVW	IJNO	IKLX	IKWX	ILWX	INPV	IVWX	JKPV
HIQV	HJMW	HKLV	HKVW	HLVW	HNOX	HQVX	IJNP	IKMN	ILMN	IMNO	INPW	JKLM	JKPW
HIQW	HJMX	HKLW	HKVX	HLVX	HNPQ	HQWX	IJNQ	IKMO	ILMO	IMNP	INPX	JKLN	JKPX
HIQX	HJNO	HKLX	HKWX	HLWX	HNPV	HVWX	IJNV	IKMP	ILMP	IMNQ	INQV	JKLO	JKQV
HIVW	HJNP	HKMN	HLMN	HMNO	HNPW	IJKL	IJNW	IKMQ	ILMQ	IMNV	INQW	JKLP	JKQW
HIVX	HJNQ	HKMO	HLMO	HMNP	HNPX	IJKM	IJNX	IKMV	ILMV	IMNW	INQX	JKLQ	JKQX
HIWX	HJNV	HKMP	HLMP	HMNQ	HNQV	IJKN	IJOP	IKMW	ILMW	IMNX	INVW	JKLV	JKVW
HJKL	HJNW	HKMQ	HLMQ	HMNV	HNQW	IJKO	IJOQ	IKMX	ILMX	IMOP	INVX	JKLW	JKVX
HJKM	HJNX	HKMV	HLMV	HMNW	HNQX	IJKP	IJOV	IKNO	ILNO	IMOQ	INWX	JKLX	JKWX
HJKN	HJOP	HKMW	HLMW	HMNX	HNQV	IJKQ	IJOW	IKNP	ILNP	IMOV	IOPQ	JKMN	JLMN
HJKO	HJOQ	HKMX	HLMX	HMOP	HNQX	IJKV	IJOX	IKNQ	ILNQ	IMOW	IOPV	JKMO	JLMO
HJKP	HJOV	HKNO	HLNO	HMOQ	HNWX	IJKW	IJPQ	IKNV	ILNV	IMOX	IOPW	JKMP	JLMP
HJKQ	HJOW	HKNP	HLNP	HMOV	HOPQ	IJKX	IJPV	IKNW	ILNW	IMPQ	IOPX	JKMQ	JLMQ
HJKV	HJOX	HKNQ	HLNQ	HMOW	HOPV	IJLM	IJPW	IKNX	ILNX	IMPV	IOQV	JKMV	JLMV
HJKW	HJPQ	HKNV	HLNV	HMOX	HOPW	IJLN	IJPX	IKOP	ILOP	IMPW	IOQW	JKMW	JLMW
HJKX	HJPV	HKNW	HLNW	HMPQ	HOPX	IJLO	IJQV	IKOQ	ILOQ	IMPX	IOQX	JKMX	JLMX
HJLM	HJPW	HKNX	HLNX	HMPV	HOQV	IJLP	IJQW	IKOV	ILOV	IMQV	IOVW	JKNO	JLNO
HJLN	HJPX	HKOP	HLOP	HMPW	HOQW	IJLQ	IJQX	IKOW	ILOW	IMQW	IOVX	JKNP	JLNP
HJLO	HJQV	HKOQ	HLOQ	HMPX	HOQX	IJLV	IJVW	IKOX	ILOX	IMQX	IOWX	JKNQ	JLNQ
HJLP	HJQW	HKOV	HLOV	HMQV	HOVW	IJLW	IJVX	IKPQ	ILPQ	IMVW	IPQV	JKNV	JLNV
HJLQ	HJQX	HKOW	HLOW	HMQW	HOVX	IJLX	IJWX	IKPV	ILPV	IMVX	IPQW	JKNW	JLNW
HJLV	HJVW	HKOX	HLOX	HMQX	HOWX	IJMN	IKLM	IKPW	ILPW	IMWX	IPQX	JKNX	JLNX
HJLW	HJVX	HKPQ	HLPQ	HMVW	HPQV	IJMO	IKLN	IKPX	ILPX	INOP	IPVW	JKOP	JLOP
HJLX	HJWX	HKPV	HLPV	HMVX	HPQW	IJMP	IKLO	IKQV	ILQV	INOQ	IPVX	JKOQ	JLOQ

Marking codes for turtles:

4 scutes marked = 1,799 individual marks (continued from previous pages):

JLOV	JMQV	JOVW	KLOV	KMQV	KOVW	LMQV	LOVW	MOPQ	NOVX
JLOW	JMQW	JOVX	KLOW	KMQW	KOVX	LMQW	LOVX	MOPV	NOWX
JLOX	JMQX	JOWX	KLOX	KMQX	KOWX	LMQX	LOWX	MOPW	NPQV
JLPQ	JMVW	JPQV	KLPQ	KMVW	KPQV	LMVW	LPQV	MOPX	NPQW
JLPV	JMVX	JPQW	KLPV	KMVX	KPQW	LMVX	LPQW	MOQV	NPQX
JLPW	JMWX	JPQX	KLPW	KMWX	KPQX	LMWX	LPQX	MOQW	NPVW
JLPX	JNOP	JPVW	KLPX	KNOP	KPVW	LNOP	LPVW	MOQX	NPVX
JLQV	JNOQ	JPVX	KLQV	KNOQ	KPVX	LNOQ	LPVX	MOVW	NPWX
JLQW	JNOV	JPWX	KLQW	KNOV	KPWX	LNOV	LPWX	MOVX	NQVW
JLQX	JNOW	JQVW	KLQX	KNOW	KQVW	LNOW	LQVW	MOWX	NQVX
JLVW	JNOX	JQVX	KLVW	KNOX	KQVX	LNOX	LQVX	MPQV	NQWX
JLVX	JNPQ	JQWX	KLVX	KNPQ	KQWX	LNPQ	LQWX	MPQW	NVWX
JLWX	JNPV	JVWX	KLWX	KNPV	KVWX	LNPV	LVWX	MPQX	OPQV
JMNO	JNPW	KLMN	KMNO	KNPW	LMNO	LNPW	MNOP	MPVW	OPQW
JMNP	JNPX	KLMO	KMNP	KNPX	LMNP	LNPX	MNOQ	MPVX	OPQX
JMNQ	JNQV	KLMP	KMNQ	KNQV	LMNQ	LNQV	MNOV	MPWX	OPVW
JMNV	JNQW	KLMQ	KMNV	KNQW	LMNV	LNQW	MNOW	MQVW	OPVX
JMNW	JNQX	KLMV	KMNW	KNQX	LMNW	LNQX	MNOX	MQVX	OPWX
JMNX	JNVW	KLMW	KMNX	KNVW	LMNX	LVNW	MNPQ	MQWX	OQVW
JMOP	JNVX	KLMX	KMOP	KNVX	LMOP	LVNX	MNPV	MVWX	OQVX
JMOQ	JNWX	KLNO	KMOQ	KNWX	LMOQ	LVNX	MNPW	NOPQ	OQWX
JMOV	JOPQ	KLNP	KMOV	KOPQ	LMOV	LOPQ	MNPX	NOPV	OVWX
JMOW	JOPV	KLNQ	KMOW	KOPV	LMOW	LOPV	MNQV	NOPW	PQVW
JMOX	JOPW	KLNV	KMOX	KOPW	LMOX	LOPW	MNQW	NOPX	PQVX
JMPQ	JOPX	KLNW	KMPQ	KOPX	LMPQ	LOPX	MNQX	NOQV	PQWX
JMPV	JOQV	KLNX	KMPV	KOQV	LMPV	LOQV	MNVW	NOQW	PVWX
JMPW	JOQW	KLOP	KMPW	KOQW	LMPW	LOQW	MNVX	NOQX	QVWX
JMPX	JOQX	KLOQ	KMPX	KOQX	LMPX	LOQX	MNWX	NOVW	

Herptofauna Photo Vouchering

The following is adapted from a chapter in the PARC Herpetofauna Inventory and Monitoring Techniques book, edited by Gabrielle Grater and Kurt Buhlmann of the Savanna River Ecology Lab. The photo vouchering chapter was compiled by John Jensen with the Georgia Department of Natural Resources.

All photo vouchers (regardless of taxa) should be labeled as:

Speciesname_property_date_photographerinitials(optional).jpg

For example: Tigersalamdaner_Mccoywma_July122011_KEK.jpg OR Tigersalamander_Mccoywma_July112011.jpg

If more than one photo would have the same label, simply add a number after the name of the species:

Tigersalamander2_Mccoywma_July112011.jpg

The purpose of photo vouchering is to provide evidence that a species occurs in a given area. This is necessary to ensure confidence in reported records and to ensure that the sightings are accurate. For some species, photo records may not be adequate for vouchers. In these situations, a voucher specimen may be needed. Iowa's amphibians are distinct enough from each other that they can be voucher with high quality photography, with one exception. The gray tree frogs (*Hyla versicolor* and *H. chrysoscelis*) can only be distinguished by DNA chromosomal analysis or by the sound of their calls with *H. chrysoscelis* having a call with a higher pitch. It is recommended that if vouchering of these species is needed, a recording be made of their calls. It should be possible to document all of Iowa's reptiles with photographs if the objective is only to document the occurrence of a species at a given site.

Definitions

Anal plate - The wide scale anterior to the anus.

Dorsal - Of or involving the back.

Dorsolateral - Of or involving both the back and the side.

Keeled - A ridge down the center of the scale, resulting in a rough or dull appearance.

Lateral - Of or involving the side.

Ventral - Of or involving the abdomen.

Basic Guidelines

1. Include a scale in the photo (or at least some of the photos of the same individual) such as a ruler, coin, pencil, or human hand.
2. Photograph animal as soon after capture as possible - some amphibians can change color when placed into containers.
3. It may be easier or necessary to place a frog or salamander into a Ziploc baggie. This will help immobilize the animal. The photo can be made through the clear plastic of the baggie. Be sure to include a small amount of water in the baggie so the amphibian does not desiccate and do NOT leave the animal in the baggie for more than a few minutes at most.

A whole body shot should be taken of each individual to be photo vouchered.

This first, standard photo should be at a dorsal angle (looking at the back of the individual). In addition to this photo, several additional photos will be needed depending on the species. Most field guides describe the characteristics that will be seen (or absent) from each of the photo angles. If the field guide lists a defining characteristic that is not accounted for in the lists for each group of animals below, please also photograph the presence or absence of that characteristic.

Salamanders

For the 5 salamander species which occur in Iowa, all should have an additional photo taken which clearly shows the whole body from a dorsolateral angle. For 4 of the 5 species, clear, high definition photos from those 2 angles (dorsal and dorsolateral) should be sufficient for identification. The mudpuppy (*Necturus maculosus*), however, should also have the belly photographed.

Anurans

Frogs

The following list of 6 additional photos should be taken to voucher the *Lithobates* species listed by the photo angle. *Lithobates* of unknown species or those found outside of their known range should have photos taken from all of the following angles:

1. Dorsal showing the full extent of the dorsolateral ridges (*R. blairi*, *R. pipiens*, *R. utricularia*)
2. Dorsolateral (All *Rana* species)
3. Head - dorsal (*R. catesbeiana*)
4. Head - lateral (*R. pipiens*, *R. sphenoccephala*)
5. Toe webbing (*R. catesbeiana*, *R. clamitans*)
6. Ventral or belly (*R. palustris*)

*Suspected wood frogs (*R. sylvatica*) should have all of the above photos taken.

Treefrogs

The following list of additional photos should be taken to voucher treefrog species listed by each angle. Treefrog species have toe-pads. If you are unsure of which treefrog species is in hand, please take photos of all the following angles:

1. Back of thigh (*Acris crepitans*, *Hyla chrysoscelis*, *H. versicolor*)
2. Dorsolateral (*H. chrysoscelis*, *H. versicolor*, *Pseudacris crucifer*, *P. triseriata*)
3. Head - dorsal (*A. crepitans*)
4. Toe webbing (*A. crepitans*)

*Remember that the only way to distinguish between *H. chrysoscelis* and *H. versicolor* in the field is by the sound of their calls. If it is necessary to voucher these species from a particular site, a recording of their calls must be made. If you document a *Hyla* from one of the following counties, please make an audio recording of the call of the individual. The counties listed have had no known records of the species in the last 20 years.

Hyla chrysoscelis:

Audubon	Des Moines	Keokuk	Pottawattamie
Buena Vista	Fayette	Kossuth	Poweshiek
Calhoun	Franklin	Lee	Shelby
Carroll	Grundy	Marrion	Sioux
Cedar	Hancock	Mitchell	Taylor
Chickasaw	Hardin	Montgomery	Union
Clayton	Henry	Osceola	Washington
Dallas	Ida	Plymouth	Worth
Delaware	Jefferson	Pocahontas	Wright

Hyla versicolor:

Audubon	Crawford	Ida	Plymouth
Buena Vista	Dickinson	Iowa	Pocahontas
Carroll	Fayette	Kossuth	Poweshiek
Cass	Floyd	Lyon	Shelby
Cedar	Grundy	Mitchell	Taylor
Cherokee	Hamilton	Montgomery	Union
Chickasaw	Hardin	Osceloa	Worth
Clay	Humbolt	Page	

Toads

The following list of additional photos should be taken for each of Iowa's 4 toad species in order to voucher the species presence for a given site.

1. Head - dorsal
2. Dorsolateral
3. Number of warts occurring per spot (the photo should be a close-up of several spots where the number of warts can be clearly counted)

4. Ventral

Spadefoot toad

There is only one species of spadefoot toad known to occur in Iowa. In addition to the lateral photo for this species, additional photos showing the eyes and the spade on the back foot would clearly voucher this species.

Snakes

Nonvenomous Snakes

For all nonvenomous snakes, a dorsolateral photo should be taken in addition to that of the lateral angle. It is also suggested to photograph both the ventral side and the anal plate of many of the species in Iowa. These 4 photos should be sufficient to document the majority of the nonvenomous species with a few exceptions:

1. Additional dorsolateral photo, close enough to be able to count the rows of scales with striping above the ventral scales (all *Thamnophis* sp.)
2. Head - dorsal (*Virginia valeriae*)
3. Head - lateral (*Heterodon* sp.)
4. Photograph showing the keeled or lack of keeled scales (*Opheodrys aestivus* & *O. vernalis*)

Venomous Snakes

It is not recommended to handle a venomous snake without the proper training on the appropriate handling techniques. Take the best photo possible without endangering yourself. The dorsal pattern on Iowa's 4 venomous snakes is distinct enough to allow identification. This picture can be taken without touching the animal. Remember to keep an appropriate distance from the snake at all times.

Lizards

Iowa has 5 lizards. Photographs of the dorsolateral angle should be taken of each of them in addition to the dorsal photo. In addition, the slender glass lizard (*Ophisaurus attenuatus*) should be photographed from the lateral angle, and the 3 skink species (*Eumeces fasciatus*, *E. obsoletus*, and *E. septentrionalis*) should be documented from the lateral side of the head, specifically focusing on the ear area, and the lateral side of the tail.

Turtles

All of Iowa's turtles should have photographs of the dorsolateral side and the ventral side (also known as the plastron in turtles) taken in addition to the dorsal photograph.

Other suggested photos include:

1. Head - dorsal (*Graptemys geographica*, & *G. pseudogeographica*)
2. Head - lateral (*Clemmys insculpta*, *Emydoidea blandingi*, *Kinosternon flavescens*, *Sternotherus odoratus*, & *Trachemys scripta*)
3. Leading (neck) edge of carapace (*Apalone mutica* & *A. spinifera*)
4. Nostrils (*A. mutica* & *A. spinifera*)

Appendix 3:

Macroinvertebrate Sampling

The following is copied verbatim from the INDR (2001) sampling protocol, pages 6-14: The upstream side is a mesh window that allows water to flow through the sampler while keeping all drifting macroinvertebrates out of the sampler. The downstream side of the cylinder has a funnel-shaped mesh collection bag and collection container for capturing macroinvertebrates dislodged as substrates inside the sampler are agitated. The modified-Hess sampler is most effective in shallow riffles and runs (<1.5 feet or 45.7 cm) with abundant rock substrates. This sampling device performs well in streams where there is a mixture of substrate particle sizes and the sampler can be penetrated 2-4 inches (5-10 cm) into the stream bottom.

Whenever possible, collect the triplicate samples from the same riffle or run. If the riffle or run is too small to obtain 3 samples, collect the remaining samples from another suitable riffle or run in the sampling reach. Record observations on the amount and type of periphyton growing on the substrates, the amount of embeddedness of coarse substrates, and the amount of macroinvertebrate colonization on the field data sheet (see appendix).

Apply the following protocol when collecting the modified-Hess samples:

1. Approach the riffle or sampling area from downstream to minimize disturbance.
2. Select the area to place the sampler and push the sampler 2-4 inches in to the substrate, with the funnel collection bag downstream.
3. Carefully wash all cobbles and large gravel particles within the cylinder and remove all clinging organisms before discarding.
4. Vigorously agitate the remaining substrate to approximately the same depth as the base of the sampler.
5. Try to rinse as many macroinvertebrates as possible off of the sampler and funnel net, down into the collection container.
6. Transfer the contents of the collection container and all remaining organisms on the sampler into the sample container.

Process the triplicate modified-Hess samples individually and do not composite them in the field. Add a 10% formalin solution to the sample containers to field preserve them for later analysis. Buffer the sample by adding 3 grams of borax to one liter of solution to neutralize the pH of formalin and prevent shrinkage and damage to the tissue of preserved organisms (USGS 1993).

Label the sample containers with indelible ink. The information on the label must include stream name, site identification number, sampling date, collector, and a unique sample identification number. Complete a sampling documentation form for each sample according to University of Iowa Hygienic Laboratory (UHL) Limnology field sampling protocols. Record the sample identification numbers on the field observation data sheet.

Artificial Substrates

In streams that lack productive riffle or run habitat, use the modified Hester-Dendy artificial substrates to obtain the semi-quantitative samples. Deployment of 4 multi-plate artificial substrates occurs at each sampling site. The colonization period lasts a minimum of 4 weeks and must not exceed 6 weeks. The advantages of artificial substrates, which include habitat standardization and macroinvertebrate productivity, seem to outweigh their disadvantages that include habitat artificiality and taxa selectivity.

Each artificial substrate consists of 8 1/8" x 4" x 4" (or 20.6 cm x 10.2 cm x 10.2 cm) wood plates and 12 1/8" thick and 1" diameter (or 30.8 cm thick and 2.5 cm diameter) cylindrical PVC spacers. The total surface area of the multi-plate unit is 145.6 in² (0.094 m²) (OEPA 1989).

Placement of the spacers between the wood plates on a 1/4" (0.64 cm) threaded steel rod is as follows: 3 single spacers on top, 3 double spacers in the middle, and 1 triple spacer on the bottom.

Artificial substrate placement - Try to deploy the artificial substrates in moderately swift run habitat with firm substrate (sand or sand/gravel, not silt or muck). Apply the following deployment criteria to ensure consistent artificial substrate placement across sampling sites and ecoregions:

1. Deploy the artificial substrates in flowing water having a current velocity of 0.5-1.5 feet per second (15.2-45.7 cm per second).
2. Deploy the artificial substrates in runs with depths of 1-3 feet (30.8-91.4 cm). Consider the anticipated flow stability when determining the appropriate distance from the top plate to the surface of the water. Ideally, deploy the sampling unit in the photic zone of the water column and sufficiently deep to ensure that the top plates remain submersed throughout the 4-6 week colonization period if flow levels decline. The distance from the top plate to the surface of the water is normally between 4-8 inches (10.2-20.3 cm). The bottom plate should be at least 3 inches (7.62 cm) above the bottom to prevent sedimentation of the sampling unit.
3. Deploy the artificial substrate units in the main axis of flow and at least 3 feet (0.91 m) from shore. Place the 4 sampling units in a diamond configuration approximately 3-5 feet (0.91-1.5 m) apart.

Whenever possible, locate the sampling units near the downstream boundary of the sampling reach to enable the benthic macroinvertebrates residing on natural substrates in the sampling reach an opportunity to colonize the artificial substrates via drift. Careful consideration of the susceptibility to vandalism and damage from high flows is critical in the placement of artificial substrates.

Illustrate on a hand-sketched map, the location of the substrates with distances to at least 2 landmarks on the shore indicated. Attachment of brightly colored nylon flagging tape to the artificial substrate units may make them easier to find after colonization. Using wooden survey stakes or flagging tape to mark the approximate locations of artificial substrates is also accepted.

Field sample processing - Retrieve the artificial substrates in a downstream to upstream manner. Remove all artificial substrate units present after the colonization period from the stream. Evaluate the status of the substrates and choose the 3 'best' substrates to process. 'Best' is those substrates that are still completely submersed at time of retrieval and free from an extraordinary amount of silt or debris. Samples obtained from heavily damaged or silted units are discarded only after determining that 3 acceptable samples, containing at least 100 organisms per sample, are available.

Examine each artificial substrate during removal and record the following observations on the field data sheet (see appendix):

1. Amount and type of periphyton growth on the plates.
2. Amount of sedimentation and/or other damages to the plates.
3. Amount of benthic macroinvertebrate colonization.

Remove the artificial substrates from the streambed with care to minimize the loss of macroinvertebrates. Carefully remove any extraneous debris, such as leaves or sticks, residing against the sampling unit before removing the unit from the stream bottom. Place a 500 µm mesh collection bag over the sampling unit and draw tightly at the base to insure that any dislodged organisms are not lost while the artificial substrate is pulled from the stream bed.

Empty the artificial substrate unit and other contents of the collection bag into a white enamel pan containing a small amount of clean water. Remove all clinging organisms from the collection bag with forceps and place in the pan. Disassemble the artificial substrate unit and remove the macroinvertebrates from the plates by gentle scraping each plate surface with a single-edge razor blade or pocketknife. Rinse and examine all extraneous debris (e.g., leaves and sticks) for macroinvertebrates and then discard. Transfer the pan contents to a labeled sample jar containing 10% formalin solution. Use separate labeled containers for the artificial substrate samples and do not composite the samples in the field or laboratory.

Label the sample jars with the following information: stream name, site identification number, sampling date, collector, and the unique sample identification number. Fill out a sample documentation form for each sample according to UHL Limnology field sampling protocols.

Record the sample identification numbers on the field observation data sheet.

Multi-Habitat Sampling Procedures

The purpose of sampling multi-habitat is to increase the number of macroinvertebrate taxa represented on the qualitative list of taxa for the sampling site. Habitat-specific sampling (e.g., riffle-only sampling) is known to result in an underestimate of taxa richness for an entire reach of stream compared to multi-habitat sampling methods (Lenat 1988, Mackey 1984).

Multi-habitat sampling is preferably conducted on the same day but after, or simultaneous to, the retrieval of artificial substrates of natural substrate sampling. The multi-habitat sampling requires 2 or 3 crew members. Before initiating the sampling, crewmembers must review sampling procedures and divide-up tasks. Time allocation for natural substrate multi-habitat sampling and processing is approximately 1.5 person hours. In stream reaches that have complex benthic habitat and/or high biological diversity, extend the sampling time to ensure adequate sampling of the reach. Indicate the amount of extra sampling time on the field data sheet.

Sampling approach - Subdivide the sampling reach into 3 areas: upper, middle, and lower reach. One crewmember is responsible for each of the areas. Typically, crewmembers use standard No. 30 brass sieves to collect and concentrate organisms; however, wash buckets, kick-nets, or other sampling gear are also accepted. The mesh size of all nets, sieves, wash buckets, or other sampling gear used in multi-habitat sampling ranges from 500-600 μ m. Collect macroinvertebrates from all accessible types of benthic substrates by handpicking or sieving.

Common techniques used to collect insects include

- Sieving the gravel, fine substrate, clay hardpan, and overhanging vegetation
- Disturbing the rocky riffle and run areas by foot and using the sieve as a drift capture tool
- Handpicking macroinvertebrates from large cobbles and boulders, woody debris, and any other large substrates found in the stream.

It is important to sample as many different substrates as possible by not lingering in 1 area too long. When 3 crewmembers conduct the sampling, each crewmember should try to collect approximately 40-50 organisms. When 2 crewmembers are sampling, each should try to collect 60-75 organisms. It is important to collect as many different types of organisms as possible.

However, if during sampling, it appears the taxa richness is minimal, the number of organisms per crewmember mentioned above still applies.

Each crewmember carries a plastic sampling container that serves as a temporary receptacle during sampling. At the end of the allotted sampling time (1.5 combined person-hours), combine the sample containers into one labeled sample jar containing a 10% formalin solution. Label the sample container with the following information: stream name, site identification number, sampling date, collector(s), and a unique sample identification number. Complete the sample documentation for the multi-habitat sample according to UHL Limnology field sampling protocol. Record the unique sample identification number on the field observation data sheet.

Laboratory Macroinvertebrate Sample Processing: Field preserved benthic macroinvertebrate samples are transported to UHL, transferred into 85% ethanol solution, and stored until identification. Obtain (pick) a random subsample of 100 organisms from each triplicate semi-quantitative sample (Modified-Hess or artificial substrate). Sort and identify every organism in the composited qualitative multi-habitat samples (all picks). Initially sort all organisms by order in preparation of the more detailed taxonomic analysis.

Identify the macroinvertebrates in the samples to the “lowest practical taxonomic level”. The lowest practical taxonomic level varies between and within invertebrate orders depending on the availability of appropriate taxonomic keys and the amount of time and expertise needed to attain precise determinations. The lowest practical taxonomic level is usually genus or species, however, in certain problematic taxa (e.g., Chironomidae and Oligochaetes) it is family level. If desired, retain several representative individuals of each problematic taxon for a more precise taxonomic analysis later. Follow

UHL protocol for taxonomic verification and laboratory QA/QC procedures.

Record the totals of each taxon in the subsample on laboratory bench sheets. The data will eventually reside in the STORET/EDAS database. Following data storage, compare data printouts against laboratory bench sheets similarly to the verification process of the DNR/UHL ambient stream monitoring data in STORET.

Appendix 4:

Fish Abnormalities

Methods for Examinations of Fish External Abnormalities - Adopted from the Ohio EPA, and copied verbatim from DNR 2001.

External Abnormalities - All fish that are captured are examined for the presence of gross external anomalies and their occurrence is recorded in the fish data sheet and subsequently entered into the FINV database. In order to standardize the procedure for counting and identifying anomalies the following criteria should be followed.

All fish are examined for gross external anomalies. These are anomalies that are visible to the naked eye when the fish are captured, identified, and counted. Table 1 lists the types of anomalies which are recorded on the fish data sheet and subsequently entered into FINV. Exact counts of anomalies present (i.e. the number of tumors, lesions, etc. per fish) are not made; however, light and heavy infestations are noted for certain types of anomalies (Table 1). An external anomaly is defined as the presence of an externally visible skin or subcutaneous disorder. Ultimately, the number and percentage of DELTs and non-DELTs are computed and recorded in the FINV database. Then the total percent anomalies for a specific type of anomaly or group of anomalies can be calculated for 1 or more sites.

The following is a review of some anomalies commonly encountered in freshwater fishes. These characteristics should be used in determining the types of external anomalies present and in coding the fish data sheets.

1. Deformities - These can affect the head, spinal vertebrae, fins, stomach shape, and have a variety of causes including toxic chemicals, viruses, bacteria, (e.g. *Mycobacterium* spp.), infections, and protozoan parasites (e.g. *Myxosoma carebais*, Post 1983). Fish with extruded eyes (see Popeye disease) or obvious injuries should not be included.
2. Eroded fins - These are the result of a chronic disease principally caused by flexibacteria invading the fins and causing a necrosis of the tissue (Post 1983). Necrosis of the fins may also be caused by gryodactylids, a small trematode parasite. When necrosis occurs in the tissue at the base of the caudal fin, it is referred to as peduncle disease. Erosions also occur on the preopercle and operculum and these should be included. In Ohio streams and rivers this anomaly is generally absent in least impacted fish communities, but can have a high incidence in polluted areas. It occurs most frequently in areas with multiple stresses, particularly low or marginal dissolved oxygen (D.O.) or high temperatures in combination with chronic toxicity (Pippy and Hare 1969, Sniezko 1962).
3. Lesions and ulcers - These appear as open sores or exposed tissue and can be caused by viral (e.g. *Lymphocystis* sp.) and bacterial (e.g. *Flexibacter columnaris*, *Aeromonas* spp., *Vibrio* sp.) infections. Prominent bloody areas on fish should also be included. Small, uncharacteristic sores left by anchor worms and leeches should not be included unless they too, are likewise infected. As with eroded fins, lesions often times appear in areas impacted by multiple stresses, particularly marginal D.O. in combination with sublethal levels of toxics.
4. Tumors - These result from the loss of carefully regulated cellular proliferative growth in tissue and are generally referred to as neoplasia (Post 1983). In wild fish populations, tumors can be the result of exposure to toxic chemicals. Baumann et al. (1987) identified polynuclear aromatic hydrocarbons (PAHs) as the cause of hepatic tumors in brown bullheads in the Black River (Ohio). Viral infections (e.g. *Lymphocystis*) can also cause tumors. Parasites (e.g. *Glugea anomala*, and *Ceratomyxa hasta*; Post 1983) may cause tumor like masses, but these should not be considered as tumors. Parasite masses can be squeezed and broken between thumb and forefinger; whereas true tumors are firm and not easily broken (P. Baumann, personal communication).
5. Anchor worm (*Lernaea cyprinacea*) - This is a common parasitic copepod and can be identified by the presence of an adult female which appears as a slender worm-like body with the head attached (buried) in the flesh of the fish. A small, characteristic sore is left after the anchor worm detaches. Attachment sites are included in the determination of light and heavy infestations. If the formed attachment site becomes infected and enlarged as the result of an infection, it should be recorded as a lesion.
6. Black spot - This disease is common to fish and is caused by the larval stage of a trematode parasite (e.g. *Uvulifer ambloplitis* and *Crassiphiala bulboglossa*). They are easily identified as small black cysts (approximately the size of a pin head) on the skin and fins. Black spot has been reported as being most prevalent on fish inhabiting relatively shallow stream and lake habitats which have an abundance of aquatic vegetation with snails and fish

eating birds, 2 of its intermediate animal hosts. It may also increase in frequency in mildly polluted streams or where fish are crowded due to intermittent pooling.

7. Leeches - These parasites belong to the family Piscicolidae and are usually greenish brown in color and 5-25 mm long (Allison et al. 1977). Leeches can be identified by the presence of 2 suckers (one on each end) and the ability to contract or elongate their body. They may occur almost anywhere on the external surface of the fish, but are most frequently seen on the anteroventral surface of bullheads (*Ictalurus* spp.). Field investigators should become familiar with the small sores or scars left by leeches as these are included in the determination of light and heavy infestations. If these sores become enlarged and infected, they are also regarded as lesions. Leeches are seldom harmful to fish unless the infestation is very heavy.
8. Fungus - There is a growth that can appear on a fish's body as a white cottony growth and is most frequently caused by *Saprolegnia parasitica*. This fungus usually attacks an injured or open area of the fish and can eventually cause further disease or death.
9. Ich or *Icthyophthirus multifilis* - This is a protozoan that manifests itself on a fish's skin and fins as a white spotting. This disease rarely occurs in wild fish populations.
10. Popeye - This disease is generally identified by bulging eyes and can be caused by gas accumulation in areas where the water is gas supersaturated. It occurs most frequently in Ohio as the result of fluid accumulation from viral infection, nematodes (*Philometra* sp.), or certain trematode larvae (Rogers and Plumb 1977).

Information on external anomalies is recorded because many are either caused or exacerbated by environmental factors and often times indicate the presence of multiple, sublethal stresses. Komanda (1980) found that morphological abnormalities are uncommon in unimpacted, natural fish populations. The effects of temperature, salinity, dissolved oxygen, diet, chemicals, organic wastes, etc, especially during the ontogeny and larval stages of fish can be the cause of many types of anomalies (Berra and Au 1981). The presence of anomalies on fish may act as an index of pollution stress. A high frequency of DELT anomalies (deformities, eroded fins, lesions, and tumors) is a good indication of stress caused by sublethal stresses, intermittent stresses, and chemically contaminated substrates. The percent DELT anomalies is a metric of the IBI (Ohio EPA 1987). Field investigators are urged to refer to texts on fish health for further information and pictures of specific anomalies. If necessary, affected fish should be preserved for laboratory examination.

Table 1. Anomaly codes utilized to record external anomalies on fish.

Anomaly code	Description of the anomaly
D	Deformities of the head, skeleton, fins, and any body parts.
E	Eroded fins.
L	Lesions, ulcers.
T	Tumors.
M	Multiple DELT anomalies (e.g. lesions, tumors, etc.) on the same individual fish.
AL	Anchor worm - light infestation: fish with 5 or fewer attached worms and/or previous attachment sites.
AH	Anchor worm - heavy infestation: fish with 6 or more attached worms and/or previous attachment sites.
BL	Black spot - light infestation: spots do not cover most of the body with the average distance between spots greater than the diameter of the eye.
BH	Black spot - heavy infestation: Spots cover most of the body and fins with the average distance between spots less than or equal to the eye diameter.
CL	Leeches - light infestation: Fish with 5 or fewer attached leeches and/or previous attachment sites.
CH	Leeches - heavy infestation: Fish with 6 or more attached leeches and/or previous attachment sites.
F	Fungus.
I	Ich (<i>Icthyophthirus multifilis</i>).
N	Blind - one or both eyes; includes missing and grown over eyes (does not include eyes missing due to Popeye disease).
S	Emaciated (poor condition, thin, lacking form).

Anomaly code	Description of the anomaly
P	External parasites (other than those already specified).
W	Swirled scales.
Y	Popeye disease.
Z	Wound, other, not included above.