Iowa Multiple Species Inventory & Monitoring Program Technical Manual

Iowa Department of Natural Resources
2016

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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 14 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 (IDNR) 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 IDNR. 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 4. 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 2014. This version contains a few minor changes, including the addition of a protocol for crayfish. 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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Dr. Bill Clark, Iowa State University (ISU)
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Additional changes and updates to datasheets between 2007 and 2011 were made by Josh Otten and Paul Frese. Tyler Harms drafted the crayfish protocol in 2013. Kevin Murphy updated the bat and mussel protocols in 2015.

Chapter 1
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 (SWG), it 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 revision 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 taxa 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 (site) 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 a long term monitoring program. The program consists of surveys instead of censuses for two reasons: (1). Most likely species will be missed in some sites (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 site 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 (IDNR) has the primary responsibility for coordinating the program, but the program is designed so that partners (County Conservation Boards, USFWS, NGO's, 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 US 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 hectares (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 site 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. 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 anticipated to be gained during the inventory stage of the monitoring program include:

1. Estimation of inclusion and exclusion errors in the Iowa GAP models.
2. 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?
3. Are there detectable patterns of co-occurrence between adequately detected species?
    a. This will aid in determining whether the Iowa monitoring program would be better served to switch from a multiple species approach to an indicator species approach. If so, the indicator species selection must be supported with data for both co-occurrence patterns among species and also associations between species occurrence and habitat attributes.
4. 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.

Once the initial inventory phase has been completed and sites are visited repeatedly such that at least 2-3 years of data collection has been completed at each site, the objectives

move from those related to inventory into objectives more specifically related to monitoring. At this stage, 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 sampling sites over time?
    a. If so, what is the change in site 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 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.
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 (such as GAP).

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 taxa 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 2
Sampling Design & Plot Establishment

The strength of the monitoring program design is based upon the randomized site 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 site 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 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 state 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, although limited data will be available for agricultural lands.

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, state-owned lands. Iowa has less than 2% of the total land area in public ownership, with fewer than 1% being owned by the Iowa DNR. The 2% public ownership includes DNR lands, federal lands, and county conservation board lands. Federal entities and county conservation boards are willing to partner with the Iowa DNR to monitor lands in their ownership.

Of the < 2% 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 square miles.

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

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, 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.

Private Lands: As 98% 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 aid NGO's with salary and equipment expenses.

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 km^2 (247 ac) 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 program director (NGO, DNR, LIP, etc.) as to which protocols are or are not of interest or practicality in implementation.

Likewise, it is expected that these 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.

Alternatively, different statistical methodologies have been developed 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 hectares (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 meters in any direction) to the center of the hexagon will 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 km^2 (247 acre) area spanning a distance of 1 km (0.62 miles) centered on the middle point.

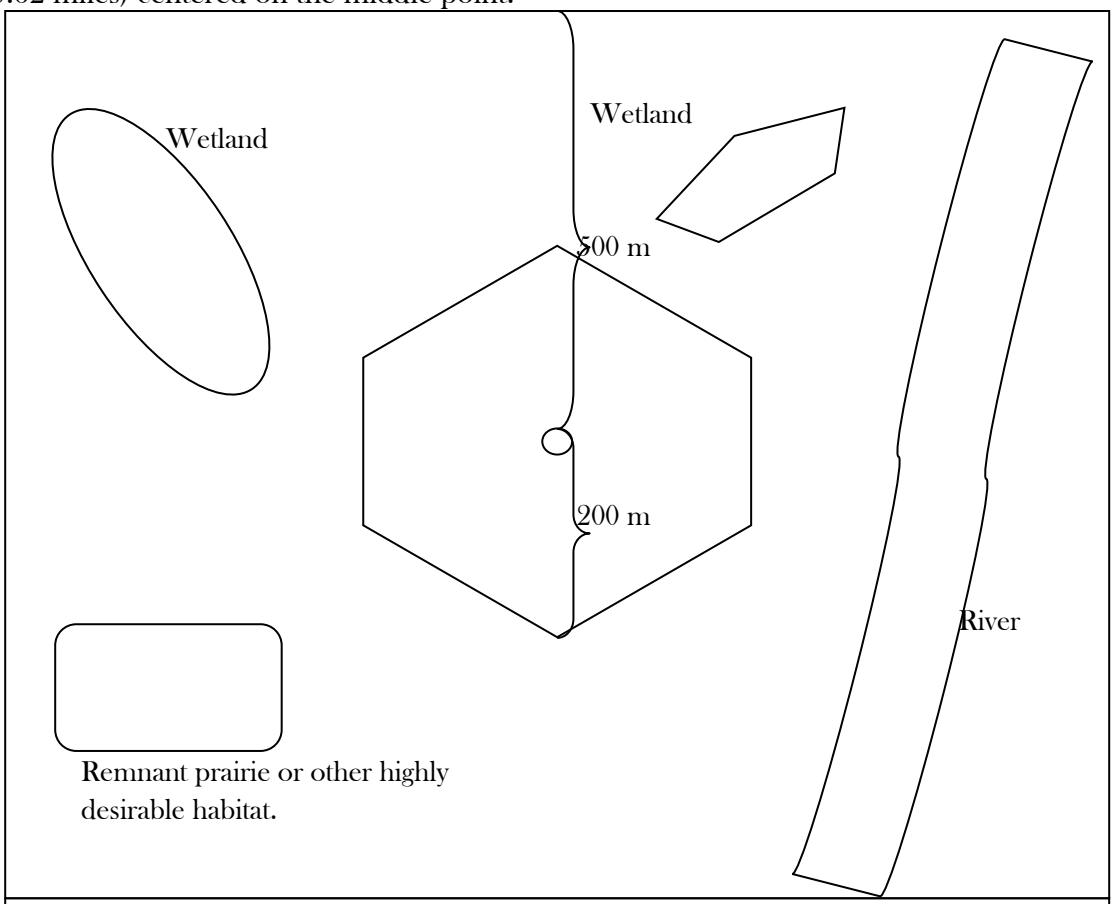


Illustration of a hexagon plot and associated sampling area. The distance from the center point to each outer point of the hexagon is 200 m, the distance from the center point to the outer edge of the additional sampling area is 500 m, or 1000 m centered on the middle point. Additional areas to be searched within this 1-km^2 area are illustrated and labeled as wetlands, remnant prairie, or other highly desirable habitat.

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 habitat being examined have no adequate wetlands to support fish or mussel populations, these protocols would simply be omitted at that site. Similarly, the mammal protocols would not be implemented at a site encompassed by open water.

DATA COLLECTION METHODS:

By utilizing the larger, 1 km^2 (247 ac) 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 km^2 area. Bird point count locations are also tied to the hexagon, yet the nocturnal call back 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 manager. 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 every site. The hexagon shape is the most efficient for spacing locations although other shapes could be used. This shape requires the least amount of walking effort while maintaining a 200 meter distance between point locations.

The GIS is then 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 sites, 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 hectare (250 acres) block. Should the site 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 ground-truthed. 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.

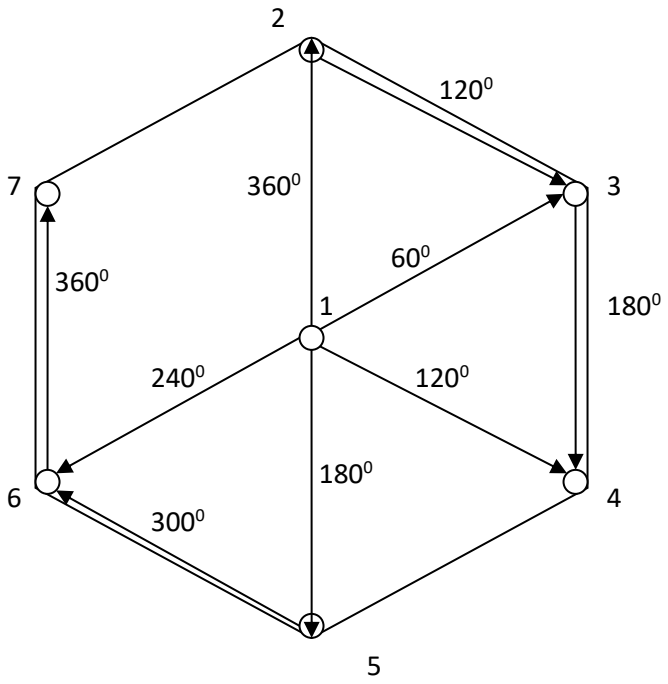


Table of formulas used to choose point locations around the point chosen to center over the representative habitat for the study area.

	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 shore 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). Al information should be ground-truthed.

GROUND-TRUTHING:

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. If it is possible to take photos in each of the 4 seasons, this should be done. Leave at least 3 months between each photo session. The following table lists the angles at which the photos should be taken.

Table of 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
11	6	240	Away from center
12	6	60	Toward center
13	7	300	Away from center
14	7	120	Toward center

It is best to use a tripod to steady the camera while taking the photos. Be sure to set the digital camera to the automatic mode and zoom out until the photo will cover the largest area. Keep a small notebook with the camera – record the number of the photo with the date, site, station number, and angle at which the photo was taken. When the photos are downloaded onto the computer, be sure to label all photos with the pertinent information.

PHOTO/MAP BOOK:

A 3-ring binder of photos and maps should be made for each crew. Included in the binder should be maps to each of the properties for which they are responsible and aerial

photos. 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 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.

EQUIPMENT LIST:

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

STAFF & TRAINING:

Staff will be trained in the basic use of the GPS unit during the training 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

DATA SHEETS:
Data sheets for this protocol are located at the end of each chapter.

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 IDNR 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 IDNR 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:

Property Information

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

For survey sites on a property but NOT on a pre-programmed site, (i.e. Nocturnal Bird Counts, Herpetofauna visual encounter surveys, Camera Stations), there is a button for data enterees to add sites within a property. Choose this and enter UTMs (if known) for the survey site.

For survey sites associated with the property transects but not on a pre-programmed site, (i.e. Butterfly Transects, Small Mammal Traps), choose "BPC-1" (a.k.a. the center point for the hexagon) as the site selection.

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 2008 during 2010, you would not have any choices for property or observer until 2008 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. 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, at least for the first complete inventory survey, 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.

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. 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.

Parameters estimated:

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

Occupancy (□) – 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 it if it is present), it is easy to calculate the occupancy probability, simply divide the number of sites with the species by the total number of sites 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 site 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 site 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 anuran calling surveys, 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 were chosen and followed. It may be necessary to truncate the data 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 sites 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 and a "-" in program PRESENCE. 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.

The instruction manual for Program PRESENCE (available at http://www.mbr-pwrc.usgs.gov/software.html) should be read in order to fully understand this software.

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 researcher 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 for a given site 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 sites are considered 'closed' as they

are in the single season surveys. A site is either occupied or unoccupied during the survey, 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 period, 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 site 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:

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

Data collection procedures:

Multiple visits are made to a given site 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 anuran calling surveys, visual encounter surveys, and bird point count surveys.

Requirements & Assumptions:

More than one year of data collection with several visits to a given site 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 sites) 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. Again, refer to the MARK help files and the Cooch and White manual for information on using covariates in Program MARK. 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 (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 locations 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 employee 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 sites 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 large 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 site has more species than another and the other site with fewer species has endangered or rare species, how do you decide which site 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 study. 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 can be accessed at http://science.nature.nps.gov/im/monitor/docs/VS_Monitoring_Reporting.pdf (last accessed 8/2009). 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. Alternatively, it may be better to stick with the agency's fiscal year (June 30th) 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 SWG 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 the 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 when the data and time allows.

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 IWAP 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 beginning in 2014. 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 http://science.nature.nps.gov/im/monitor/docs/DraftPeerReviewGuidelines.doc (last accessed on October 11, 2006) 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 or, should they choose to remain anonymous, by their representative coordinator. 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 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 and budget 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 was collected and analyzed?
3) Are the conclusions logical?

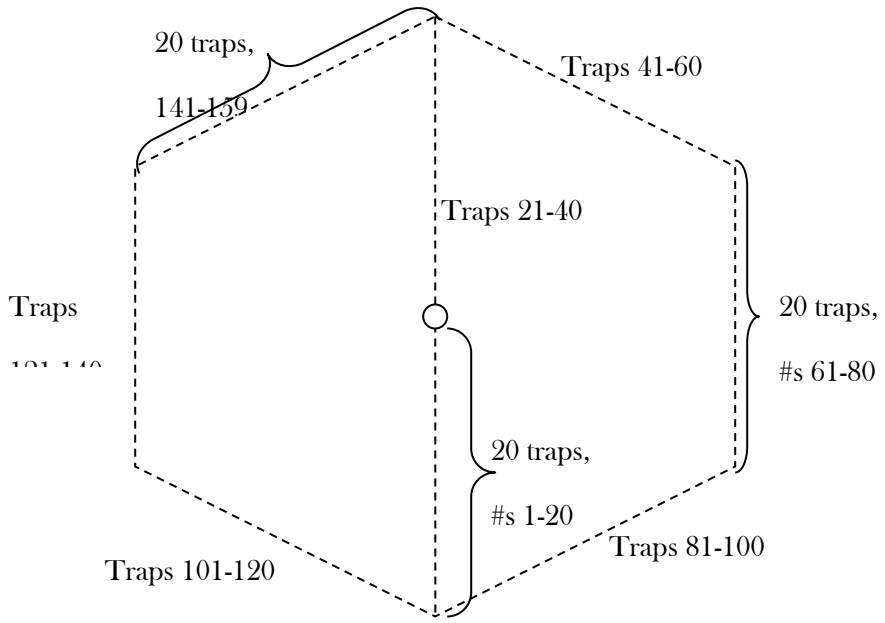
The files maintained as part of the review process should be in the possession of the program scientist (or a copy of these files), with another copy (or the originals) being maintained by the review coordinator. For Iowa, it would be appropriate for the IWAP coordinator to fill the role of MSIM review 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

MAMMAL MONITORING:

Small Mammals:

At each hexagonal sampling plot, the spacing between the traps should be 10 meters 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).

SURVEY METHODS:

Trapping can begin the last weeks of April through the middle or end of July over a 12-14 week period. Iowa MSIM typically devotes the month of August to the habitat protocols, but additional time after September 1 can be used if needed to visit all properties assigned to a particular crew to complete the small mammal trapping. Trap locations should be recorded with a GPS unit so that they can be found in succeeding years. Traps should be numbered before being set and care should be taken to ensure that traps are numbered consecutively. We typically use peanut butter as bait, knowing that the scent of the bait can attract the mammals. Opportunistically, a crew may choose to supplement the peanut butter with carrots, apple slices, rolled oats, or lard, 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 that 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 site 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. Dead animals are collected, frozen, and donated to a museum. At sites 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 sites 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.

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.

Dirty traps can be sprayed with Lysol before being placed into the enclosed truck bed. 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:

For medium and large mammals, trackplates and camera surveys were the primary method of detection through 2008. The same protocols as those outlined in the FS MSIM program were followed, such that 3 track plate stations and 3 camera stations were arranged anywhere within the larger, 101 ha area of the property.

Trail cameras continue to be used in this project. 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 during the summer. Each site is visited every 3 to 4 days for a total of 4 visits to replace bait and check battery life of the cameras.

Camera stations include a digital 35mm camera. We are currently using game cameras with infrared flashes. The camera should be attached to a tree or some other immovable substrate. The bait should be 0.5 m or less from the ground. Bait and camera are placed on either the same tree or on adjacent trees. Cameras are attached to the tree using a tripod, wires, nylon straps, and/or duct tape. A large feather can be hung 1.5 m above the ground to act as a visual cue. Batteries and flash cards are checked and replaced as needed (Manley et al. 2004). Bait can include anything (carp, deer carcass, cat food, etc) but be sure to discuss bait choice with MSIM staff.

An additional attractant *could be* used for the camera stations. This attractant could be a mixture of skunk gland derivative (Gusto, Minnesota Trapline Products, Pennock, MN) and lanolin (M&M Fur, Inc, Bridgewater, SD). A 1 oz jar of Gusto is added to 32 oz of heated lanolin in liquid form. One tablespoon of the mixture is placed approximately 4 meters from each station on something such as a tree branch. The mixture is neither re-applied nor removed for the duration of the 10 days. Alternatively, a commercial scent could be purchased. The Iowa MSIM program has not yet tried any of these alternatives.

All photos should be identified to species and 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.

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 employee the track plate method.

Most tracks are found along the edges of water bodies or after rains. Under correct environmental conditions (i.e. the day after rain events), technicians will search for tracks in appropriate habitats, recording the species and location of the tracks and the time spent searching the property. Photos should be taken of each new track on each property. Photos should be downloaded to the computer and labeled with date, property, site, and track ID. Using the same naming template as above, each photo should be saved, with the Survey type in this case being MVES for Mammal Visual Encounter Survey.

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.

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 NEEDED:

Per site:

Small mammal protocol:
- 159 Sherman traps, plus replacements, per site
- 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:	3 game cameras and cards 3 wires or cables 100 feet of 22 gauge bailing wire OR airplane cables Turkey feathers or other bait 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; ad 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 occupied using either program PRESENCE or MARK. (See the Data Analysis Protocol is in the Office Protocols for additional information).

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, with all records being checked by at least 2 individuals. Detection probabilities and the number of sites 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 were 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.

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 "*" indicates species considered to be Data Deficient in the IWAP.

Target species:

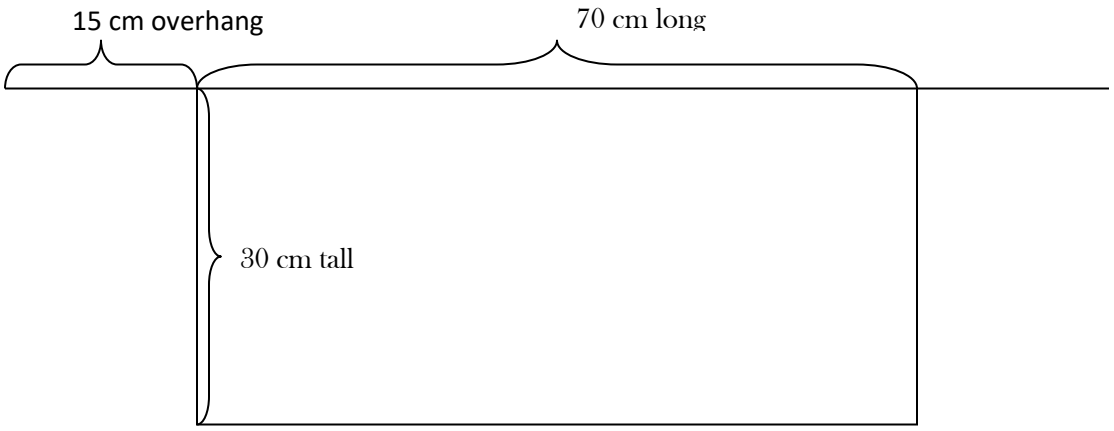
Common Name	Scientific Name	Habitat
Hayden's shrew*	*Sorex haydeni*	Grassland, woodland, riparian
Elliot's short-tailed shrew*	*Blarina hylophaga*	Forest, woodland, savanna, grassland
Southern short-tailed shrew*	*Blarina carolinensis*	Forests, shrublands, grasslands, wetalnds
Least shrew	*Cryptotis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

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.

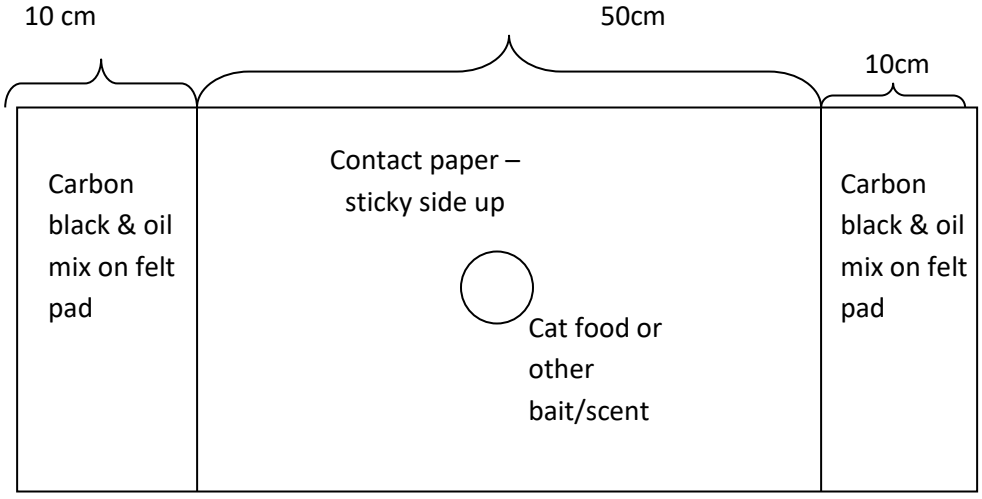
Trackplates

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 meters 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.

Track Plate & 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) Polyethylene enclosures- In areas with heavy precipitation, the track plates boxes should be covered with polyethylene.
4) 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. However, this is still a developing field and much more work need to be done before this technique is practical and monetarily feasible. 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.

MAMMAL TRACK VES: pg____ of____

Property:________________________ Observer(s):_____________ Date:_________ Visit #:______

Start time:_____ End time:_____ Start Temp(F):_____ End Temp(F):____ %clouds:____ Rain: (Y/N)

Foot length in mm.

Species				Common Name	Foot length	Habitat area name (Location)	Photo voucher	Comments

Corresponding #:__________ by:___ Date data entered:______ Date checked:_______by:___

SHERMAN TRAP SMALL MAMMAL LIVE TRAPPING:

Property:_____ Date:_____ Observer(s)._____ pg ___of___

Day (1-5):___ (am/pm) Visit #:____ Start time:_____ End time:_____ Start temp (F):____ End temp:____ %clouds:____ Rain: (Yes/No)

Fate: A=alive; D=dead; E=escape. Age: A=adult; SA=sub-adult; J=juvenile; U=unknown. Sex: M=male; F=female; U=unknown.

Br. (Breeding status): 1=non-breeding; 2=pregnant; 3=lactating; 4=testes enlarged (breeding condition); 5=unknown. Recapture: Yes/No if so, what's the mark?

Weight in grams. Length in mm. Foot lgth=Hind foot length. Please remember to list incidental animal sightings on back or in the margins of this data sheet. Please remember to record "tripped but empty" and "open but no bait" trap numbers.

Trap number	Species code	Fate	Age	Sex	Br.	Recap	Mark	Tot. wgt	Bag wgt	Ind wgt	Tot. lgth	Tail lgth	Foot lgth	Ear lgth	Comments

Corresponding #:_____ Date entered:_____ Entered by:_____ Date checked:_____ Checked by:_____

CAMERA DATA SHEET:

This should be filled out in the office or lab. Make sure date on camera is set correctly. Date start is the date the camera was set out on the property. Date end is the date the camera was picked up. Date=Date on photo. Picture number(s)=Appropriate numbers/labels that correlate with data for each individual.

Property:_____ Observer(s)._____ Date Start:_____ Date End:_____ pg ___ of___

Camera locations (GPS Coordinates): #1:_____ #2:_____ 3#:_____

Camera number	Species code				# of individuals	Date	Picture number(s)	Photo name on computer	Comments

Corresponding #:_____Date entered:_____ Entered by:_____ Date checked:_____ Checked by:_____

Chapter Nine
Bat Monitoring Protocol

BAT MONITORING:

Bat detections will be done primarily using ANABAT detectors. In secure locations, 2 detectors could be deployed at suitable habitat locations for 3 occasions during the season, for a total of 6-9 nights of recording bat calls per year per detector. Most likely, secure locations will be lacking, in which case, technicians will walk through the habitat recording their routes with a GPS while recording bat activity with the ANABAT.

These waling 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).

ANABAT CALL LIBRARY:

Should additional calls need to be collected in establishing regional keys for the ANABAT detectors, bats will be captured at different locations and the calls of known individuals recorded using the detectors. Calls should be recorded both directly into the detector and also through the weather protection covers for the detectors. Mist nets will be the primary method used to capture bats. Suitable habitats will be chosen from aerial photos or GIS database. The USFWS (1999) recovery plan for the Indiana bat suggests that no more than 1 net site per 1 km of stream be used, and no more than 2 net sites per 1 km^2 be used. In either case, it suggests that nets should be spaced at least 30 m apart.

The most promising habitat should be chosen for net placement. Ideally, the nets would be placed over water but under a closed canopy to increase capture probability. These sites can be either 'high quality' (streams, ponds, & lakes), 'moderate quality' (meadows), or 'lower quality' (roads & canopy openings in forests). Streams and ponds are the best habitats to sample as many bats forage over water, resulting in potential clusters of individuals. The road or canopy openings within a forest should help funnel bats that do not forage over water. Once the sampling sites have been randomly selected, a field visit should be done to ensure that the habitat is the correct one (as chosen from aerial photos or GIS database during the selection process). This visit can coincide with other work on the site. For example, when doing point counts for birds or trapping for small mammals, make note of potentially good habitat to trap bats at that location.

SURVEY METHODS:

ANABAT SURVEYS:

Each area is surveyed at least three times during the summer season (May 15 through August 15), with at least a week separating the visits. Indiana bats, a federally endangered species, detected outside of this timeframe may be transient or migratory (USFWS 1999). On secure properties (i.e. private lands), 2 to 3 ANABAT detectors could be deployed for 2 nights at each site. Detectors should be moved between suitable habitats each night, such that as many habitats as possible can be sampled during the 2 night timeframe. This process should be repeated 3 times during the summer season resulting in 6 nights of ANABAT deployment.

On most properties, the threat of equipment theft will be substantial and will preclude leaving the detectors unattended. At these locations, technicians will walk through suitable habitat beginning at dusk and continuing until no later than 2 AM. Routes should be recorded using a

GPS. At least 3 evenings should be spent on each property, with at least 1 week separating each 1-night visit. Each ANABAT walking survey should last for at least 2 hours.

ANABAT data and GPS coordinates or routes should be downloaded into organized folders on the computer. Be sure to label each folder with the date and property of the downloaded data.

Anabat setup and tips

There are many options for how to set up and utilize the ANABAT units used for acoustic monitoring. It is important make sure that all settings are consistent throughout a survey season so that survey methods are as well-standardized as possible. Before starting an ANABAT survey, make sure that the Data Division Ration is set to 8, the Audio Division Ratio is set to 16, and the Sensitivity is set to 7. It is imperative that the sensitivity setting on the recorder is not changed during the survey and that the sensitivity setting stays constant between surveys.

It is possible that at some point an error light may turn on (or multiple LED lights may stay lit that should turn off). These indicate that the recorder has malfunctioned in some way and should not be used again until the problem is resolved as it may not be recording data during use.

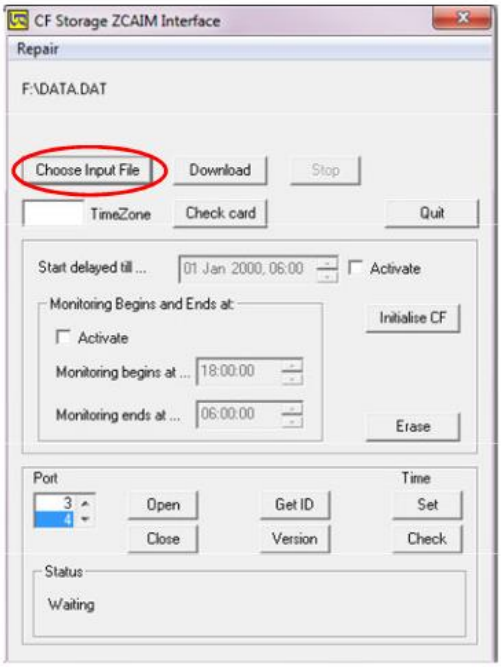
Before compact flash (CF) cards can be used in the ANABAT for the first time they must be formatted in a Windows PC and initialized with CFCread. In the event that a new card is used reformat the card into FAT (FAT16 or FAT32) file system format. Open CFCread, make sure that no check boxes are selected in the window, select "Choose Input File", navigate and select the location of the CF card in the navigation window, and then select "Initialise CF". If it has been formatted it is ready for initialization and you can confirm this. Wait patiently while it completes the process, it may take several minutes with larger cards. The card is then ready to use in the ANABAT detector.

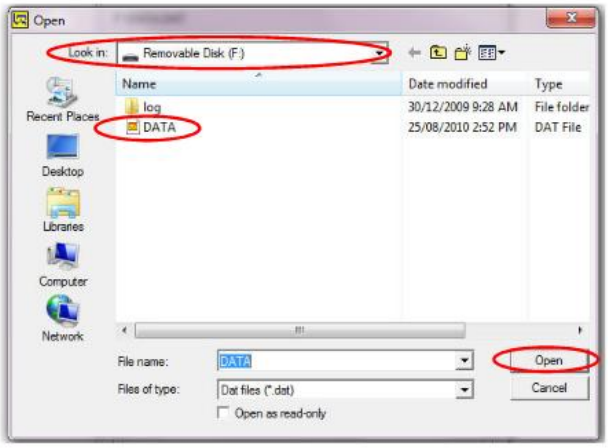
After a survey has been completed make sure that you keep a record of start and end times so that the digital files downloaded from the recorders can be matched up with the time and place they were recorded.

Steps for downloading ANABAT Data:

1) Make sure the ANABAT unit is turned off, remove the compact flash (CF) card and place it into a CF Card reader.
2) Open CFCread (a program used to initialize and download data from these cards)

3) Select “Choose Input File” and navigate to the data file (DATA.DAT) on the CF card, click “Open”

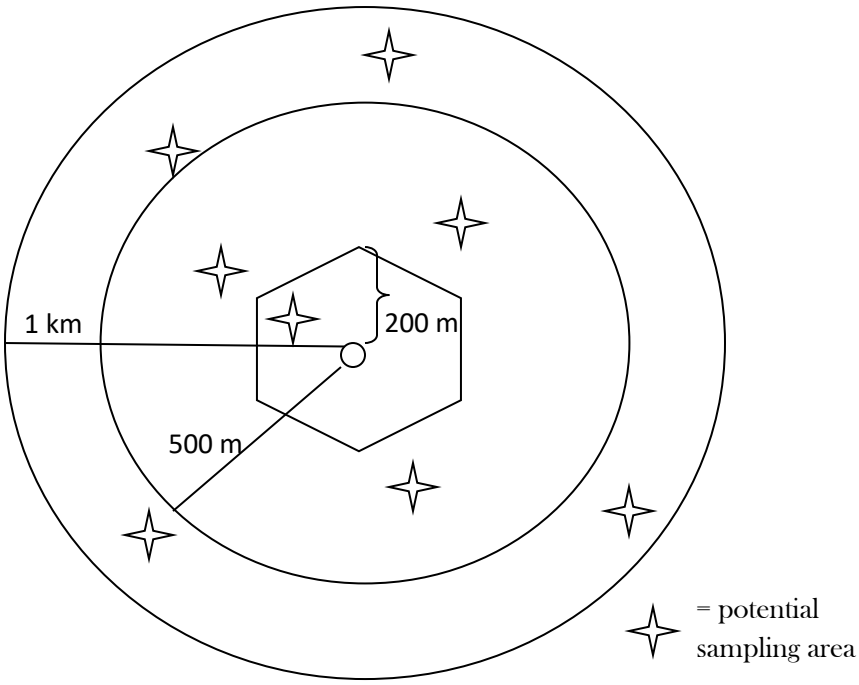




4) Click “Download” on the CFCread interface and a “Download Options” window will appear. Check the boxes for “Generate Anabat files”, “Use AutoSave Parameters”, and “Generate” for ZCA files. If an SD1 detector was used the option to use for ZCA files is “5m synch”, if an SD2 detector was used select “Raw”. After this is complete click “OK”.
5) Next choose where to save your ANABAT data to. On nights where you complete more than one survey it may be best to save it to one “downloads” folder and then separate the files out by survey when you are able to examine them to determine when and where they were recorded. Click “Download” and wait patiently for the download to complete, do not remove the CF card from the reader during this process.
6) Once the download has completed (without errors) and you have verified that the files downloaded in the correct location you can click “Erase” in the CFCread main window to clear the data from the card and prepare the card for the next surveys. The file names of the recorded files indicate what time and date they were recorded, use this information and your record of start and end times for surveys to place the files into appropriate folders for location and survey date. <u>Immediately</u> back up the data in a separate location to prevent data loss.

POSSIBLE 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.

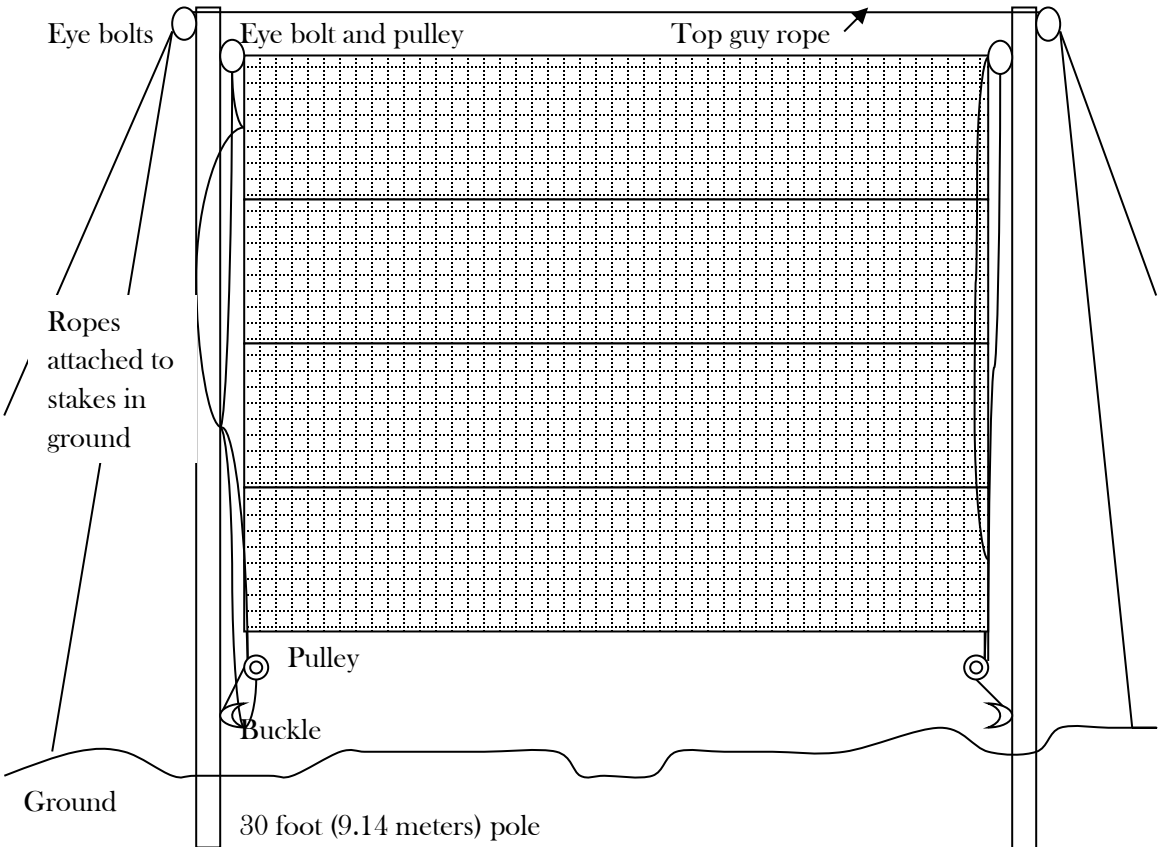


Net mesh size should be the smallest available, approximately 38 mm (1 ½" x 1 ½") openings, although 50 mm (2" x 2") can also be used. Nets should be placed such that they stretch perpendicularly across the corridor opening. Nets should cross the corridor/stream completely and reach from the stream/ground 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) available at: http://www.fws.gov/WhiteNoseSyndrome/pdf/WNSDecontaminationProtocol_v012511.pdf 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 on a pulley and rope system to the other pole.

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 capture or ANABAT field 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:

ANABAT detection: ANABAT Equipment
Weather proofing equipment
Extra batteries
Extra data memory cards
GIS
Headlamps

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

Trapping:
- 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 & TRAINNING:

ANABATs can be utilized by any field crew. A computer person will be needed to analyze calls recorded with the ANABAT detectors.

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 & MANAGEMENT:

ANABAT

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 ANABAT, it will be easy to store the data for future scrutiny. All calls should be kept on suitable electronic storage (CDs, memory stick, etc). For species identification purposes, these calls will be compared to calls of known individuals to determine species.

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.

At the end of each survey, 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 areas occupied using Program MARK or PRESENCE. See chapter 5 (Data Analysis) for more information.

SAFETY CONSIDERATIONS:

All personnel that will be handling bats need to have the pre-exposure rabies vaccination series.

The ANABAT 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 and/or radios, 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 '*' indicates species designated as Data Deficient.

Target bat species:

Common Name	Scientific Name	Habitat
[A]Northern long-eared bat	*Myotis septentrionalis*	Forest, woodlands
Little Brown Bat	*Myotis lucifugus*	Woodland near waterways, readily rears young in buildings and bat houses
[A]Indiana bat	*Myotis sodalis*	Forest, upland, & riparian
Silver haired bat	*Lasionycteris noctivagans*	Woodland edge, riparian
Eastern pipistrelle	*Perimyotid subflavus*	Woodland edge, riparian
Evening bat*	*Nycticeius humeralis*	Forest, riparian

[A]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.

ANABAT SURVEY DATA SHEET.

Property:_____ DATE:_____ OBS._____ pg ___of____
Start time:_____End Time:_____ Start temp (F):_____ End temp (F):_____ %clouds:_____
Rain:_____ Wind speed:_____ Visit #:_____

Segment Start Time				Segment End Time				Segment location, description of habitat, other comments

Corresponding #:_____ Data entered by:_____ date:_____ Checked by:_____ date:_____

BAT MIST NET TRAPPING. Property:____________________ DATE:___________ OBS._________________ pg ___of____
Start time:_____End Time:_______ Start temp (F):______ End temp (F):_______%clouds:________Rain:______Wind speed:________
NOTES ON NET SIZE & CONFIGURATION:____

Sex: F- Female, M- Male, U- unknown; Re.st. = Reproductive status: P=Pregnant (female), L=lactating (female), NR=Non-repro, U=unknown, DT=Descended testicles (male), ND=not descended (male); Age: A=adult, J=juvenile; The calcar is checked to determine keel presence. Trg=Tragus length

Time				Net #	Species				Sex	Re. st.	Age	Forearm (mm)			Ear (mm)		Thumb (mm)		Foot (mm)		Keel Y/N	Trg. (mm)	Comments, other measurements

Corresponding #:_________ Data entered by:______ date:____________ Checked by:________ date:_______

Back of Bat Trapping Data Sheet. If habitat data has been collected as part of another sampling protocol, the following information should still be collected during mist netting:

MAP: Diagram trapping location within the 26 acre plot, include water source/net site placement, net configuration and numbers, net length and height, and a North arrow. Also indicate potential roosting locations such as caves, hollow snags, bridges, etc….

Habitat:
Water type:_____Diameter or distance across:_____
Other trapping habitat (road, trail, etc):_____
Percent of emergent vegetative cover in water body being trapped:_____
Turbidity (clear, semi-clear, murky):_____Water depth:_____
Distance to forest edge:_____
Comments:_____
_____
_____
_____

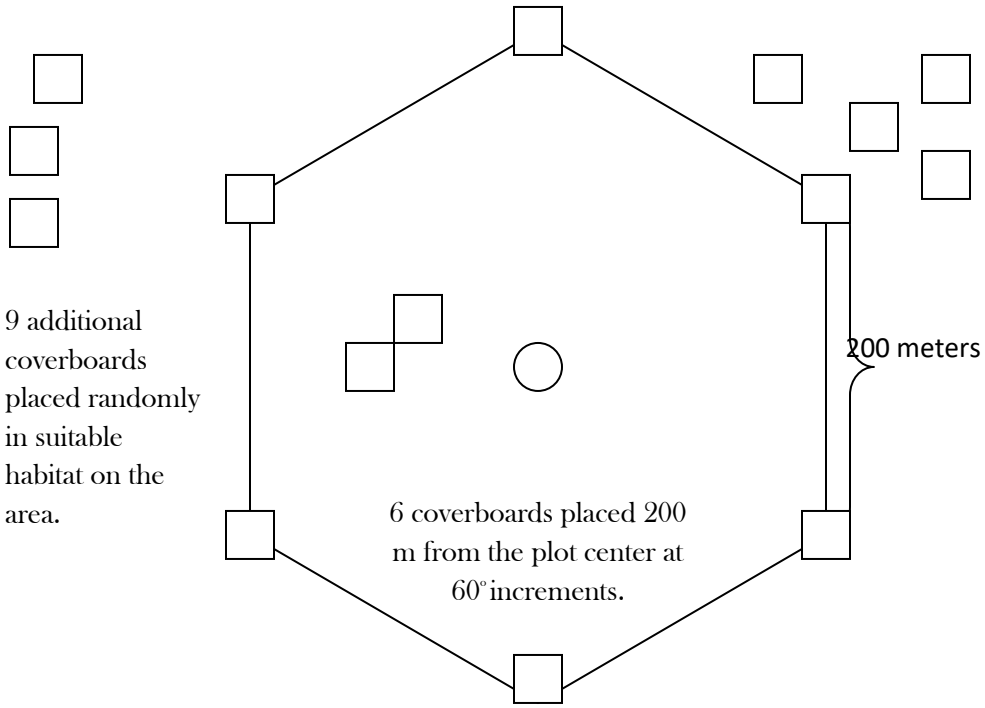
Notes on other species encountered:

Data entered by:_____Checked by:_____

Chapter Ten
Amphibian & Reptile Monitoring Protocol

AMPHIBIAN AND REPTILE MONITORING:

Visual encounter surveys (VES) will be one of the 3 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 hectare) 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 is much larger than that usually incorporated into a VES, but will allow for a large variety of habitat types to be searched.



Two other methods used in this protocol include cover boards and aquatic traps. The diagram above shows the placement of cover boards. Aquatic traps such as minnow traps should be deployed in wetlands for 3 days (and 2 nights) (Kolozsvary and Swihart 1999) and checked daily.

SURVEY METHODS:

Visual Encounter Surveys:

Each of the sampling sites 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 a 2 week lag between all site visits. 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. In addition, wetlands should be walked to a reasonable depth (the shoreline to about a 0.5 meter 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-m^2 sheet that is at least 1 cm thick. Ours tend to be 4 feet by 2 feet. Six coverboards should be placed approximately 200 meters from the center of the sample plot in the 6 directions of the point count stations. This means the first coverboard should be placed due north, 200 m from the center of the plot. Every 60 degrees (so, 0^o, 60^o, 120^o, 180^o, 240^o, and 300^o), 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 meters from the pole to prevent people from accidental stepping on the coverboard. 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. Nine additional coverboards should be placed in suitable habitat to attract snakes. 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 to allow them to be found in the future. If possible, hang a piece of flagging nearby to assist in relocating the boards.

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. Minnow traps do not need to be baited. For these traps, try to find areas of vegetation. Traps should be checked daily 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 can be baited with sardines, frozen fish, or fresh carp (which may work best). 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, 2 crab nets, 3 minnow traps, and if appropriate habitat, 1 fyke net. See diagram below for more description of these traps.

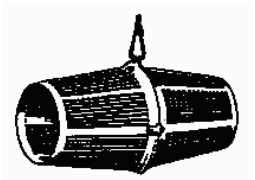


Diagram of a minnow trap. Taken from the British Columbia Integrated Land Management Bureau website (10/22/2011).

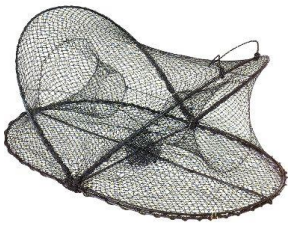


Diagram of a crab trap. Taken from Cabela's website (10/22/2011).

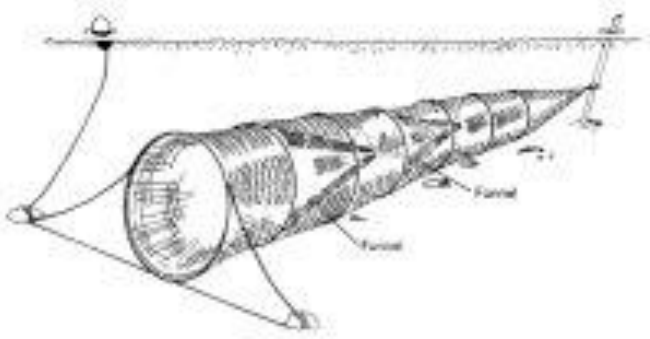


Diagram of a hoop net. Taken from the EPA's website (10/22/2011).

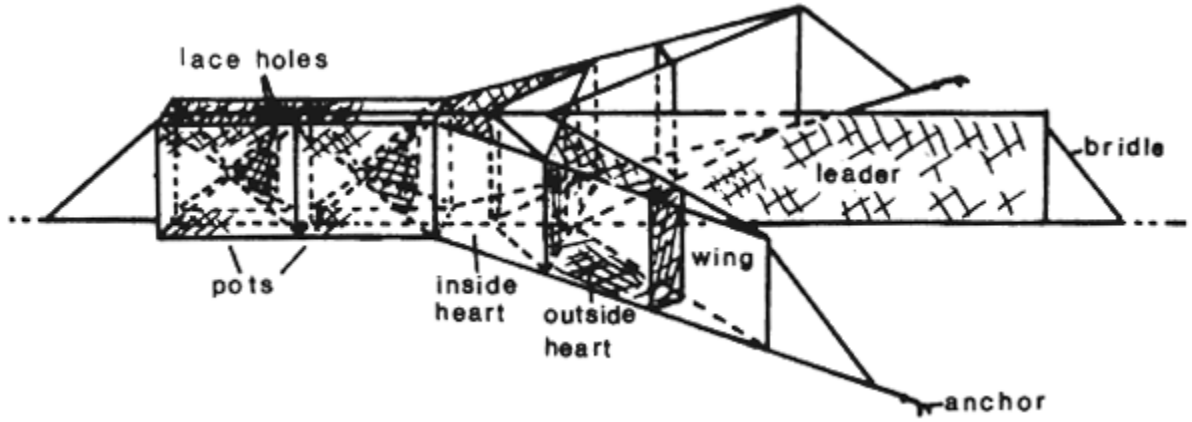


Diagram of a fyke net. Taken from the Food and Agriculture Organization of the United Nations website (10/22/2011).

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.

Recording Captures:

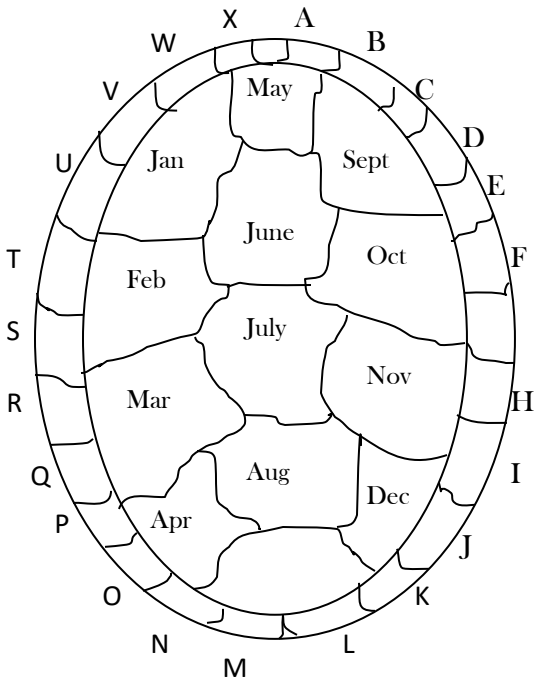
To protect the amphibian, it is critical that the technician's hands be free of any chemicals or lotions. Insect repellent can be absorbed through the skin of the amphibian, resulting in the animal's death.

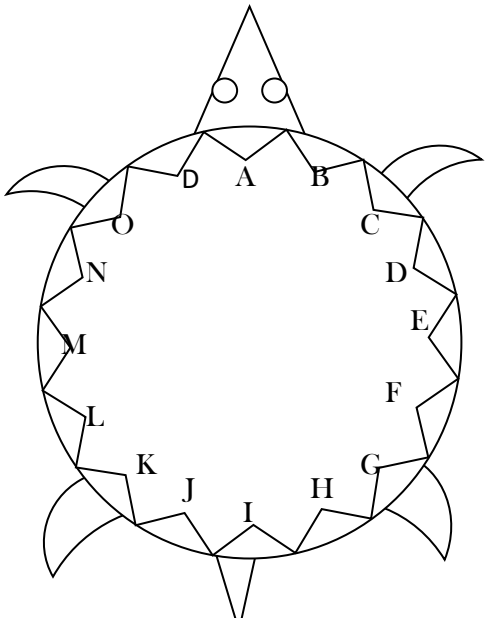
For each observation, record the species, detection type (visual, auditory, sign), age class (adult, sub-adult, juvenile), and substrate type (rock, log, bare ground, etc.). In addition, animals that are captured should be measured (snout to vent and total length), assigned to sex and status, and marked. 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 IDNR 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.

Snakes and lizards can be marked by placing a dot of nail polish on the back. Turtles should be marked using a shell notch system (see Appendix 4). Notches can be created using a 3-sided file. By marking 1 to 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). With all turtles, 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.
Amphibians could be marked with either toe-clipping (cheaper) or VIE 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, should be marked if the site is to be visited each year.

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" & "X". The posterior 2 are always "L" & "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 the 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 ground-truthed in the field.

EQUIPMENT LIST:

Kestrel temperature and wind gauge
Water thermometer and pH meter
Pair leather gloves (for large snake captures)
Hand spade or rake
Field guides & Anuran call tape for reference (leave in truck)
Hand lens
Stop watches
Digital camera
Snake sticks
Pair hip waders
15 Coverboards
Minnow traps
Frye nets
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 & TRAINNING:

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 4) proficiency testing. 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 using programs MARK or PRESENCE (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.

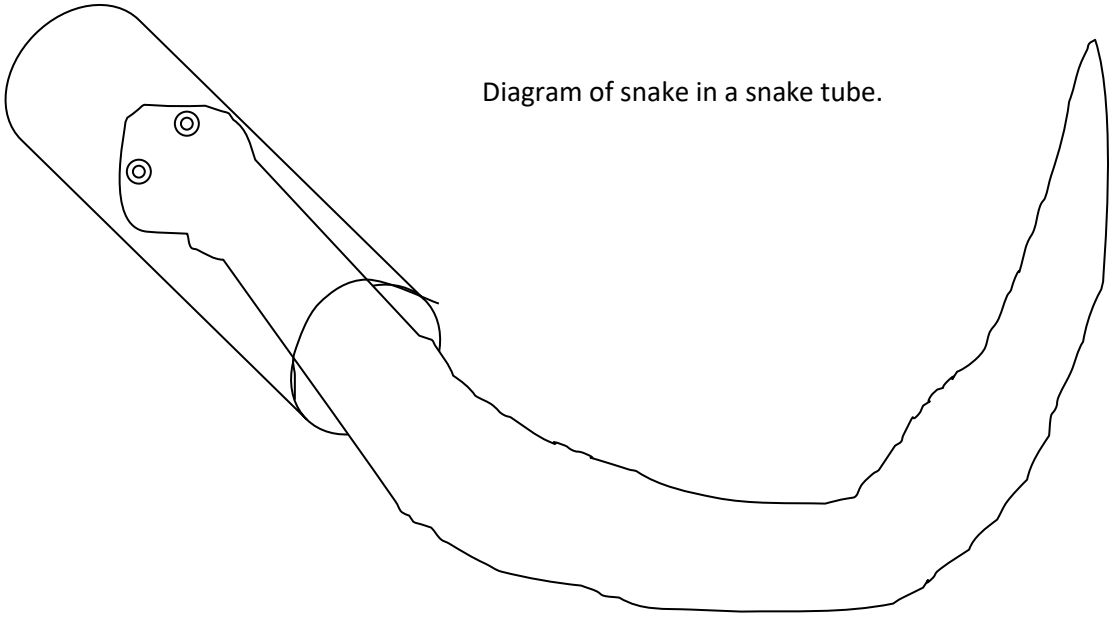


Diagram of snake in a snake tube.

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 '*' indicates species designated as Data Deficient.

Target amphibian species:

Common Name	Scientific Name	Habitat
Mudpuppy*	*Necturus maculosus*	Clean rivers, streams, lakes, reservoirs
Eastern newt	*Notophthalmus viridescens*	Vegetated woodland ponds, roadside flooded ditches, & adjacent habitat
Tiger salamander	*Ambystoma tigrinum*	Generalist
Smallmouth salamander	*Ambystoma texanum*	Woodland pools, open woods
Blue-spotted salamander	*Ambystoma laterale*	Woodland pools, open woods
Great plains toad	*Anaxyrus cognatus*	Prairie, nonnative grassland
Fowler's toad	*Anaxyrus fowleri*	Grasslands & woodlands
Woodhouse's toad	*Anaxyrus woodhousii*	Woodlands & savanna
Blanchard's Cricket frog	*Acris crepitans*	Shallow wetlands & streams
Cope's gray treefrog	*Hyla chrysocelis*	Wetlands, savanna grasslands, woodland edges
Eastern gray treefrog	*Hyla versicolor*	Forests, woodlands, riparian areas
Crawfish frog	*Lithobates areolata*	Prairie marshes, ponds, river floodplains
Pickerel frog	*Lithobates palustris*	Cold water streams, rivers, and impoundments
Northern leopard frog	*Lithobates pipiens*	All aquatic habitats
Southern leopard frog	*Lithobates sphenocephalus*	On-stream impoundments, wetlands, ponds, and backwaters
Plains spadefoot toad	*Spea bombifrons*	Grasslands in the Loess Hills

Target reptile species:

Common Name	Scientific Name	Habitat
Snapping turtle	*Chelydra serpentina*	All aquatic habitats
Blanding's turtle	*Emydoidea blandingii*	Shallow, well vegetated wetlands
Wood turtle	*Clemmys insculpta*	Floodplain forest, rivers
Northern map turtle	*Graptemys geographica*	Rivers & streams
Southern map turtle	*Graptemys ouachitensis*	Mississippi River and Associated oxbows
False map turtle	*Graptemys pseudogeographica*	Slow portions of large rivers
Ornate box turtle	*Terrepene ornata*	Sand prairies, savanna
Yellow mud turtle	*Kinosternon flavescens*	Shallow, ephemeral pools, adjacent areas with nearly pure sand soils
Eastern musk turtle	*Sternotherus odoratus*	Backwaters and spring fed ponds adjacent to sandy uplands
Smooth softshell turtle	*Apalone mutica*	Large rivers
Spiny softshell turtle	*Apolone spinifera*	Rivers, streams, large lakes
Slender glass lizard	*Ophisaurus attenuatus*	Prairie, pasture, forest edge, savanna
Common five-lined skink	*Plestiodon fasciatus*	Deciduous forests along the blufflands of the Mississippi River and large eastern Iowa rivers
Great plains skink	*Plestiodon obsoletus*	Rocky prairie, forest edge
Northern prairie skink	*Plestiodon septentrionalis*	Sandy prairie-forest edge, wetland edge
Six-lined racerunner	*Aspidocelissexlineatus*	Sand prairies, savanna
Western worm snake	*Carphophis amoenus*	Rocky woodland
Prairie ringneck Snake	*Diadophis punctatus*	Woodlands, savanna & adjacent grasslands
Western (plains) hognose snake	*Heterodon nasicus*	Sand prairie
Eastern hognose snake	*Heterodon platirhinos*	Woodland, savanna, & grassland
Prairie kingsnake	*Lampropeltis calligaster*	Woodland edge, open woodland, grassland, savanna
Speckled kingsnake*	*Lampropeltis getulus*	Prairie, woodland edge, savanna
Copperbelly water snake	*Nerodia erythrogaster neglecta*	Backwater sloughs and forested wetlands
Diamondback water snake	*Nerodia rhombifera*	Quiet pools and backwater sloughs
Common water snake	*Nerodia sipedon*	Lakes, ponds, marshes, streams, backwaters
Smooth green snake	*Opheodrys vernalis*	Old field, savanna, wet prairie, marsh

Target Reptile Species continued:

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

[A]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.

<u>VES Augmentation</u>

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.

AMPHIBIAN AND REPTILE VES:

Property:_____ Date:_____ Observer(s):_____ Visit #:_____ pg ____ of____

Start time:_____ End time:_____ Start temp:_____ End temp:_____ %clouds:_____ Rain: (Yes/No) Wind speed:_____

Area corresponds to the GIS map of the location being surveyed. Each map should have wetlands numbered. If the area being searched or where an animal is encountered is not marked on the map, record approximate location. Det. Type=Detection type: V=visual; C=capture; A=auditory; S=sign. Sub.=Subsrate type: R=rock; L=log; W=water; V=vegetation; X=litter; S=soil. SVL=Snout to Vent Length or plastron length for turtles. Lengths in cm. Age: A=adult; SA=sub-adult; J=juvenile; M=metamorph; L=tadpole; E=egg mass. Sex: M/F/U. Photo #=corresponding photo number/s for voucher. Include breeding status if known in comments.

Det.type	Time	Species	Number	Sub.	Mark	Total Length	SVL/ Plastron	Age	Sex	Photo #	Comments

Please remember to list any incidental animal sightings on the back or in the margins of this data sheet.

Corresponding #:_____Date entered:_____ Entered by:_____ Date checked:_____ Checked by:____

AQUATIC TRAP DATA SHEET:

Property:_____ Date:_____ Observer(s):_____ Visit #:_____ pg____ of____
Start time:_____ End time:_____ Start temp:_____ End temp:_____ %clouds:_____ Rain: (Yes/No) Wind speed:_____

Area corresponds to the map of the location being surveyed. Each map should have wetlands numbered. If the area being trapped or where an animal is caught is not marked on the map, record approximate location. Trap type= TT=turtle hoop trap; LN=large net trap (fish or crab); MM=metal minnow trap. Mark ID=for turtles.

Fate: Alive/Dead/Recapture/Escape. Sex: M/F/U. Age: A=adult; SA=sub-adult; J=juvenile; M=metamorph; L=tadpole. Photo #=corresponding photo number/s for voucher. Be sure to list ALL traps (if empty record as 'none' or 'null'.

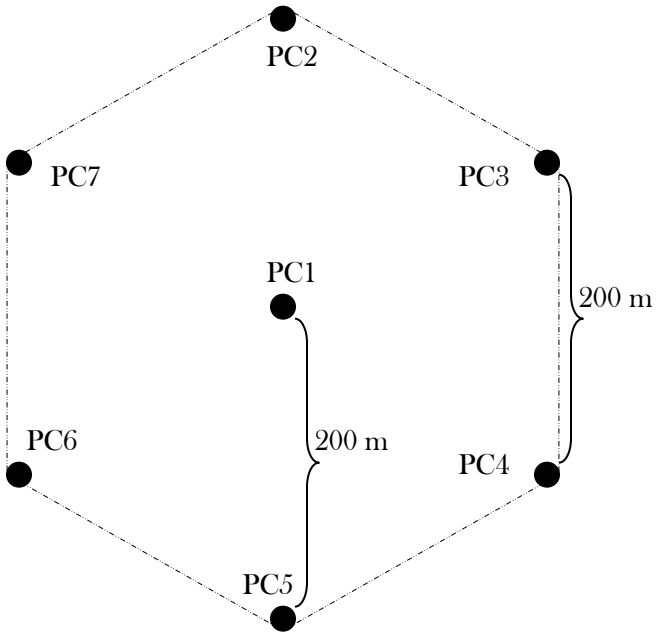
Area or GPS coordinates	Trap type		Trap #	Species				Common name	# caught	Mark ID	Fate/ Recap	Plastron or length (cm)	Sex	Age	Photo #	Comments

Herp Corresponding #:_____Date entered:_____ Entered by:_____ Date checked:_____ Checked by:____
Fish Corresponding #:_____Date entered:_____ Entered by:_____ Date checked:_____ Checked by:____

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 0^0, 60^0, 120^0, 180^0, 240^0, and 300^0, respectively. The point count stations are 200 meters 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 timing of observations at the point counts will include 3 seasons, basically. 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 Chair and 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 permanent sampling locations 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 (http://www.mbr-pwrc.usgs.gov/bbs/instruct.html), and including fog, steady drizzle, prolonged rain, and wind > 20 km/h (12 mph), will 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 birds of the same species spread out on territories within the sampling area. These distinctions are critically important in analyzing the data.

Other data to be collected at every sampling hexagonal plot include the date, cloud cover, wind speed, and start & end times and temperature. 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 site, 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). 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.

ENIVERONMENTAL 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) & MP3 player
- Stopwatch
- 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:

Program PRESENCE (MacKenzie et al. 2002) will calculate probability of detection estimates and proportion of points occupied for all of the data collected during these surveys. Program MARK has the same analysis capabilities using either the "Occupancy Estimation" or the "Robust Design Occupancy" data type selection buttons 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. 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 '*' 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	*Cygnus buccinator*	Wetland
American wigeon	*Anas americana*	Wetland
Blue-winged teal	*Anas discors*	Wetland
Northern pintail	*Anas acuta*	Wetland, grassland
Canvasback	*Aythya valisineria*	Wetland
Redhead	*Aythya americana*	Wetland
Ring-necked duck	*Authya collaris*	Wetland
Lesser scaup	*Aythya affinis*	Wetland
Northern bobwhite	*Colinus virginianus*	Grassland, shrubland
Ruffed grouse	*Bonasa umbellus*	Dense forest, open woodland
Sharp-tailed grouse	*Tympanuchus phasianellus*	Grassland, shrubland
Greater prairie chicken	*Tympanuchus cupido*	Grassland
Red-necked grebe	*Podiceps grisegena*	Wetland, hemi-marsh
Eared grebe	*Podiceps nigricollis*	Wetland, hemi-marsh
American white pelican	*Pelecanus erythrorhynchos*	Wetland
American bittern	*Botaurus lentiginosus*	Wetland
Black-crowned night heron	*Nycticorax nycticorax*	Wetland, wet shrubland
White-faced ibis	*Plegadis chihi*	Marsh
Bald eagle	*Haliaeetus leucocephalus*	Riparian forest, deciduous forest
Northern harrier	*Circus cyaneus*	Grassland, marsh
Red-shouldered hawk	*Buteo lineatus*	Riparian forest
Broad-winged hawk	*Buteo platypterus*	Deciduous forest
Swainson's hawk	*Buteo swainsoni*	Savanna, open woodland
King rail	*Rallus elegans*	Wetland
Common gallinule (moorhen)	*Gallinula chloropus*	Wetland
[A]Piping plover	*Charadrius melodus*	Wetland
Upland sandpiper	*Bartramia longicauda*	Grassland
Wilson's snipe	*Gallinago delicata*	Hemi-marsh
American woodcock	*Scolopax minor*	Deciduous forest, open woodland, riparian forest
Wilson's phalarope	*Phalaropus tricolor*	Wetland, grassland
Franklin's gull	*Larus pipixcan*	Wetland
[A]Least tern	*Sterna antillarum*	Wetland
Black tern	*Chlidonias niger*	Wetland
Forster's tern	*Sterna forsteri*	Wetland
Yellow-billed cuckoo	*Coccyzus americanus*	Deciduous forest, shrubland, open woodland

Target breeding bird species continued:

Common Name	Scientific Name	Habitat
Black-billed cuckoo	*Coccyzus erythropthalmus*	Riparian and deciduous forests, open woodland, shrubland
Barn owl	*Tyto alba*	Savanna
Eastern screech owl*	*Otus asio*	Riparian forest/woodland, savanna
Burrowing owl	*Speotyto cunicularia*	Grassland
Long-eared owl*	*Asio otus*	Open woodland, savanna, deciduous forest
Short-eared owl	*Asio flammeus*	Grassland
Common nighthawk	*Chordeiles minor*	Grassland, savanna
Chuck-will's-widow	*Caprimulgus carolinensis*	Open woodland, savanna
Eastern whip-poor-will	*Caprimulgus vociferus*	Deciduous forest, open woodland
Chimney swift	*Chaetura pelagica*	Older growth forests, woodlands, savanna, towns
Belted kingfisher	*Ceryle alcyon*	Wetlands, riparian, with cutbanks
Red-headed woodpecker	*Melanerpes erythrocephalus*	Savanna, open woodland, deciduous forest
Northern flicker	*Colaptes auratus*	Savanna, open woodland
American kestrel	*Falco sparverius*	Savanna, open woodland
Peregrine falcon	*Falco peregrinus*	Riparian forest, deciduous forest
Eastern wood-pewee	*Contopus virens*	Forest/woodland, savanna
Acadian flycatcher	*Empidonax virescens*	Deciduous forest, riparian forest
Say's phoebe	*Sayornis saya*	Grassland
Eastern kingbird	*Tyrannus tyrannus*	Grassland-shrub, edges
Loggerhead shrike	*Lanius ludovicianus*	Savanna, shrubland
Bell's vireo	*Vireo bellii*	Shrubland, savanna
Horned lark	*Eremophila alpestris*	Sparse grassland, agricultural land
Purple martin	*Progne subis*	Riparian forests, towns, and wetlands
Bank swallow	*Riparia riparia*	Rivers and streams with cutbanks
Sedge wren	*Cistothorus platensis*	Grassland, wetland
Bewick's wren	*Thryomanes bewickii*	Open woodland, shrubland
Veery	*Catharus fuscescens*	Riparian and deciduous forest
Wood thrush	*Hylocichla mustelina*	Deciduous and riparian forest
Brown thrasher	*Toxostoma rufum*	Shrubby grassland
Worm-eating warbler	*Helmitheros vermivorus*	Deciduous forest
Golden winged warbler	*Vermivora chrysoptera*	Shrubby open woodland
Prothonotary warbler	*Prothonotaria citrea*	Riparian forest
Kentucky warbler	*Oporornis formosus*	Deciduous and riparian forest
Common yellowthroat	*Geothlypis trichas*	Shrubland and grassland

Target breeding bird species continued:

Common Name	Scientific Name	Habitat
Cerulean warbler	*Dendroica cerulea*	Deciduous forest
Field sparrow	*Spizella pusilla*	Shrubland, grassland
Grasshopper sparrow	*Ammodramus savannarum*	Grassland
Henslow's sparrow	*Ammodramus henslowii*	Grassland
Dickcissel	*Spiza americana*	Grassland
Bobolink	*Dolichonyx oryzivorus*	Grassland
Eastern meadowlark	*Sturnella magna*	Grassland, savanna
Western meadowlark	*Sturnella neglecta*	Grassland
Baltimore oriole	*Icterus glabula*	Open woodland, savanna

Target migratory bird species:

Common Name	Scientific Name	Habitat
Greater scaup	*Aythya marila*	Lakes, rivers, wetlands
Common loon	*Gavia immer*	Clear, large, open water lakes
Little blue heron	*Egretta caerulea*	Marsh
Yellow rail	*Coturnicops noveboracensis*	Wetland, grassland
*Black rail	*Laterallus jamaicensis*	Marsh
Whooping crane	*Grus americana*	Wetland, grassland
Black-bellied plover	*Pluvialis squatarola*	Wetland
American golden-plover	*Pluvialis dominica*	Wetland
Lesser yellowlegs	*Tringa flavipes*	Wetland
Whimbrel	*Numenius phaeopus*	Wetland
Long-billed curlew	*Numenius americanus*	Grassland-wetland
Hudsonian godwit	*Limosa haemastica*	Wetland
Marbled godwit	*Limosa fedoa*	Wetland
Ruddy turnstone	*Arenaria interpres*	Grassland-wetland
^Red knot	*Calidris canutus*	Wetland
Sanderling	*Calidris alba*	Wetland
Semi-palmated sandpiper	*Calidris pusilla*	Wetland
White-rumped sandpiper	*Calidris fuscicollis*	Wetland
Pectoral sandpiper	*Calidris melanotos*	Wetland and grassland
Stilt sandpiper	*Micropalama himantopus*	Wetland
Buff-breasted sandpiper	*Tryngites subruficollis*	Wetland, short grassland
Short-billed dowitcher	*Limnodromus griseus*	Wetland
Long-billed dowitcher	*Limnodromus scolopaceus*	Marsh
Caspian tern	*Sterna caspia*	Wetland
Olive-sided flycatcher	*Contopus cooperi*	Coniferous forests, openings and edges
Sprague's pipit	*Anthus spragueii*	Grassland

Target migratory bird species continued:

Common Name	Scientific Name	Habitat
Bohemian waxwing	*Bombycilla garrulus*	Coniferous or mixed forest
Smith's longspur	*Calcarius pictus*	Grassland
Bay-breasted warbler	*Dendroica castanea*	Forest/woodland
Canada warbler	*Wilsonia canadensis*	Deciduous forest
American tree sparrow*	*Spizella arborea*	Open woodland
Le Conte's sparrow	*Ammodramus leconteii*	Grassland
Harris's sparrow	*Zonotrichia querula*	Pastures, hedgerows
White-winged crossbill	*Loxia leucoptera*	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 hectare 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 areas, 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 calling tape (or CD) 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 MP3 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 frog loggers instead of technicians to record bird calls.

Nest Searching

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

BIRD VES: pg_____ of______

Property:_____________________ Observer(s):________________ Date:____________ Visit #:___

Start time:_______ End time:_______ Start Temp (F):______ End Temp (F):______ %clouds:___

Rain: (Y/N) Wind spd:____ Disturbance:_____________________ Comments: ________________

Species				Common Name	Tally	Total Count	Habitat area name (Location)	Comments

Corresponding #:______________ by:___ Date data entered:_______ Date checked:_______by:___

INCIDENTAL SPECIES.

Location:_____ Observer(s):_____ Date:_____ Visit #:___Start time: End time:_____ Start Temp (F):_____ End Temp (F):_____ %clouds:_____ Rain: (Yes/No) Wind spd:____ Disturbance:_____ Comments: _____

List additional wildlife seen today.

Species	Location	Type of detection	# of animals	Comments
E.g.: Red winged blackbird	*Between PS 5 & 6*	*Visual*	*5*	

Corresponding #:_____ Date entered:_____ by:_____ Date checked:_____by:___

In addition to % cloud cover on other side of data sheet, indicate sky condition:_____

0=clear sky, few clouds (<25%); 1=Partly cloudy (25-70% clouds); 2=Cloudy (70-100%); 3=Rain; 4=Fog or smoke; and 6=Snow

Beaufort Wind Scale:

Beaufort Number	Wind speed (mph)	Indicators
0	<1	Smoke rises vertically
1	1-3	Wind direction shown by smoke drift
2	4-7	Wind felt on face, leaves rustle
3	8-12	Leaves in constant motion, light flag extended
4	13-18	Raises dust and loose paper, small branches move
5	19-24	Small, leafy trees sway

BIRD POINT COUNT DATA:

pg____ of_____

Location:_____ Observer(s):_____ Date:_____ Visit #:_____Start time:_____ End time:_____ Start Temp(F):____ End Temp(F):____ %clouds:___ Rain: (Y/N) Wind spd:_____ Disturbance:_____

Comments:_____

Time frame=0-3 min, 3-5 min, 5-10 min. Be sure to draw line to distinguish time frame change. Time=military time. 0-25 (etc.)=distance codes in meters, FO=flyover. If the detected individual is a singing male, indicate this under 'comment'. A=Auditory V=Visual. Remember to record species other than birds which you observe on incidental sheet.

BPC location	Time frame	Species Code				0-25	25-50	50-75	75-100	100-200	200-300	>300	FO	A/V	Male/ Female	Flock? Brood?	Comments

Corresponding #:_____ by:_____ Date entered:_____ Date checked:_____by:____

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. Each habitat section should be labeled differently so that presence data can be linked to habitat data. 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. 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.

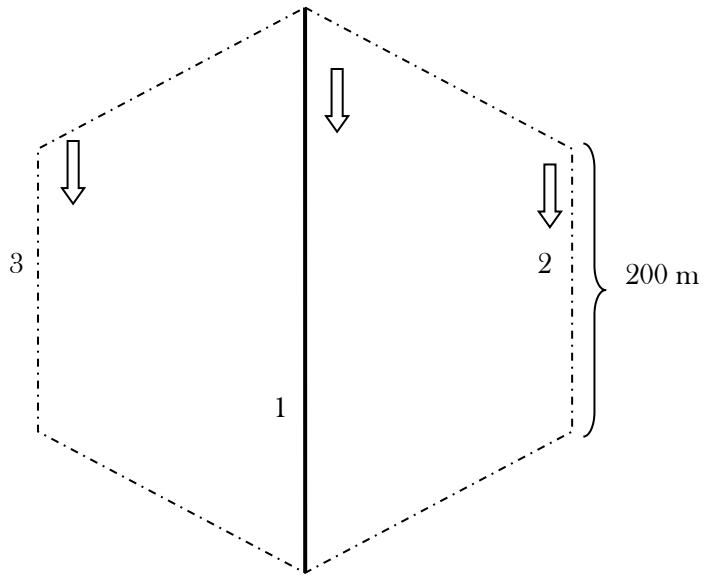
The primary transect can be divided into habitat sections and is the dividing line of the permanent hexagonal plot. Should a transect cross a paved or major road, this should be treated as a break between sections (Pollard and Yates 1993). The primary transect is 400 m in length. Transects should be 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.

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 not 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.

The butterfly season will begin the last week of May and continue through August 31, depending on weather conditions. 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). 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 during 2 hours 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 envelops 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).

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 NEEDED:

Compass
Flagging, flags & tall bamboo stakes
Stopwatch
Butterfly forceps
Glassine envelops
Pinning kit
Butterfly net
Hand lens
Field guides
Zip-lock baggies
Digital camera with macro lens
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 digital cameras, or by the field supervisor working periodically with each technician.
    - Photos should be sent to the IWAP Butterfly Subcommittee Chair 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 PRESENCE or MARK. 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 "*" 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	*Battus philenor*	Forest, open fields, & roadsides
Zebra swallowtail	*Eurytides marcellus*	Woodland along rivers
Spicebush swallowtail	*Papilio troilus*	Woodlands
Olympia marble	*Euchlow olympia*	Open woods, river bluffs, poor soils, & grasslands
Harvester	*Feniseca tarquinius*	Woodland and stream
Purplish copper	*Lycaena helloides*	Moist or disturbed areas
Acadian hairstreak	*Satyrium acadica*	Riparian & oldfield
Edward's hairstreak	*Satyrium edwardsii*	Woodlands, clearings, & areas of poor soil
Hickory hairstreak	*Satyrium caryaevorum*	Forest
Striped hairstreak	*Satyrium liparops*	Forest openings and edges, prairie streamsides
White M hairstreak*	*Parrhasius m-album*	Woodland, savanna
Henry's elfin	*Callophrys henrici*	Woodland
Reakirt's blue	*Echinargus (Hemiargus) isola*	Native prairie
Silvery blue	*Glaucopsyche lygdamus*	Open fields & woodland openings
Melissa blue	*Plebejus (Lycaeides) melissa*	Xeric prairie and gravel ridges
Aphrodite fritillary	*Speyeria aphrodite*	High quality prairie, wetlands, and fens

Target butterfly species continued:

Common Name	Scientific Name	Habitat
Regal fritillary	*Speyeria idalia*	Prairie & open grassland
Silver-bordered fritillary	*Boloria selene*	Fens, wet prairie, and meadows
Gorgone checkerspot	*Chlosyne gorgone*	Oldfield roadsides, pastures, vacant lots, and native prairie
Baltimore checkerspot	*Euphydryas phaeton*	Wetlands
Ozark Baltimore checkerspot*	*Euphydrayas phaeton ozarkae*	Wetlands, fens, bogs, woodlands
Compton Tortoiseshell	*Nymphalis vaualbum (l-album)*	Large tracts of forest
Common ringlet	*Coenoympha tullia*	Prairie & marsh edge
Eyed brown	*Satyrodes eurydice*	Fens, wet prairies, and marshes
Monarch	*Danaus plexippus*	Open habitat and disturbed areas
Southern cloudywing	*Thorybes bathyllus*	Xeric prairie
Hayhurst's scallopwing*	*Staphylus hayhurstii*	Floodplain forest, Loess Hills forests
Dreamy duskywing*	*Erynnis icelus*	Woodland or edge
Sleepy duskywing	*Errynis brizo*	Oak barrens, sand or shale soils
Juvenal's duskywing	*Erynnis juvenalis*	Oak forests
Mottled duskywing	*Erynnis martialia*	Xeric prairie
Columbine duskywing	*Erynnis lucilius*	Rocky wooded ravines
Powesheik skipperling[L]	*Oarisma powesheik*	High-quality tallgrass prairie
Ottoe skipper	*Hesperia ottoe*	Mid- and tall grass, high-quality prairie
Leonard's skipper	*Hesperia leonardus*	Open grassy areas
Dakota skipper[L]	*Hesperia dacotae*	Prairie
Crossline skipper	*Polites origines*	Xeric prairie
Long dash	*Polites mystic*	Xeric prairie in northwest, fens and wet prairies in northeast
Northern broken-dash	*Wallengrenia egeremet*	Fens, xeric prairie, forest/woodlands
Little glassywing	*Pompeius verna*	Woodland edge
Arogos skipper	*Atrytone arogos*	Prairies & grasslands
Byssus skipper	*Problema byssus*	Tallgrass prairie
Mulberry wing	*Poanes massasoit*	Wetland fens
Broad-winged skipper	*Poanes viator*	Wetalnd fens
Dion skipper	*Euphyes dion*	Sedge wetlands
Black dash	*Euphyes conspicua*	Fens, wet prairies, marshes
Two-spotted skipper	*Euphyes bimacula*	Sedge meadows & marshes
Dusted skipper	*Atrytonopsis hianna*	Bluestem grasslands & oldfields
Pepper and salt skipper	*Amblyscirtes hegon*	Edge of woods & grass waterways
Common roadside skipper*	*Amblyscirtes vialis*	High quality, xeric prairie
Swarthy skipper	*Nastra lherminier*	

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.

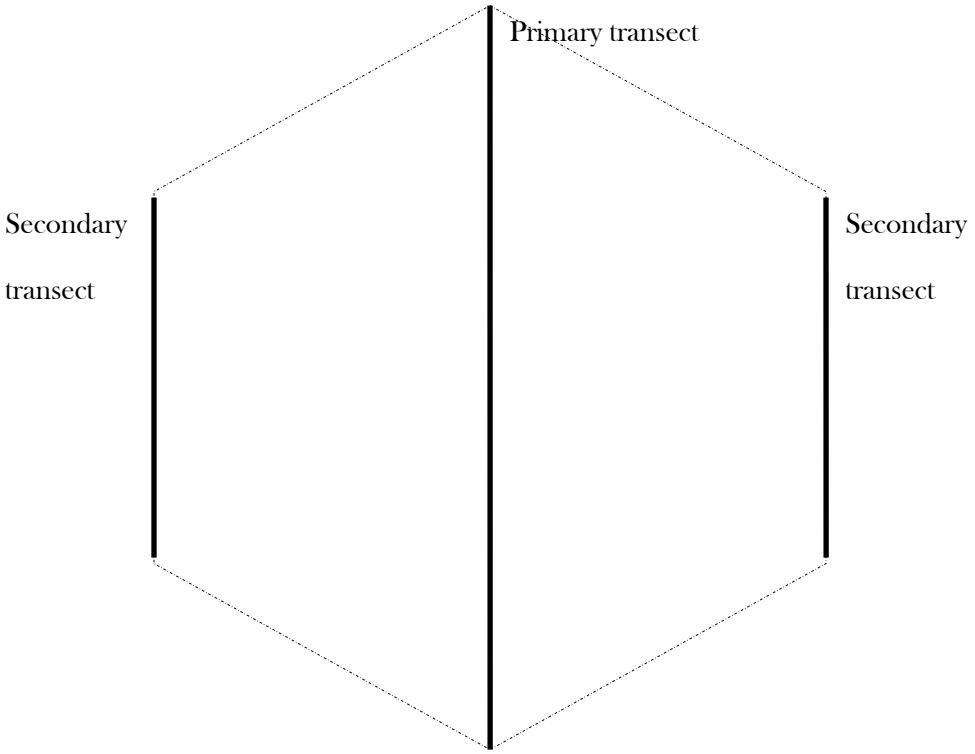
Heitzman, JR, and JE Heitzman. 1987. Butterflies and Moths of Missouri. Missouri Department of Conservation. Jefferson City, MO.

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).

Butterfly transect map. Observer:_____ Date:_____Location:_____
Sketch habitats/section breaks/roads, also record whether the canopy is open or closed for each section of the transect:

Remember, each hexagonal side is 200 m in length and the dividing transect is 400 m long.



It may be possible to do this in the lab using the GIS database, however, this data should be ground-truthed on the first butterfly transect data collection. Subsequent data collection will not need to re-confirm this information unless conditions have changed (i.e. the site was burned or logged or plowed, etc.)

BUTTERFLY TRANSECT DATA: pg___of___

Property:______________________ Observer(s):____________ Date:___________ Visit #:____

Start time:_______ End time:________ Start Temp (F):_____ End Temp (F):___ %clouds:_______ Rain: (Yes/No) Wind spd:____ Disturbance:__________________ Comments: _______________

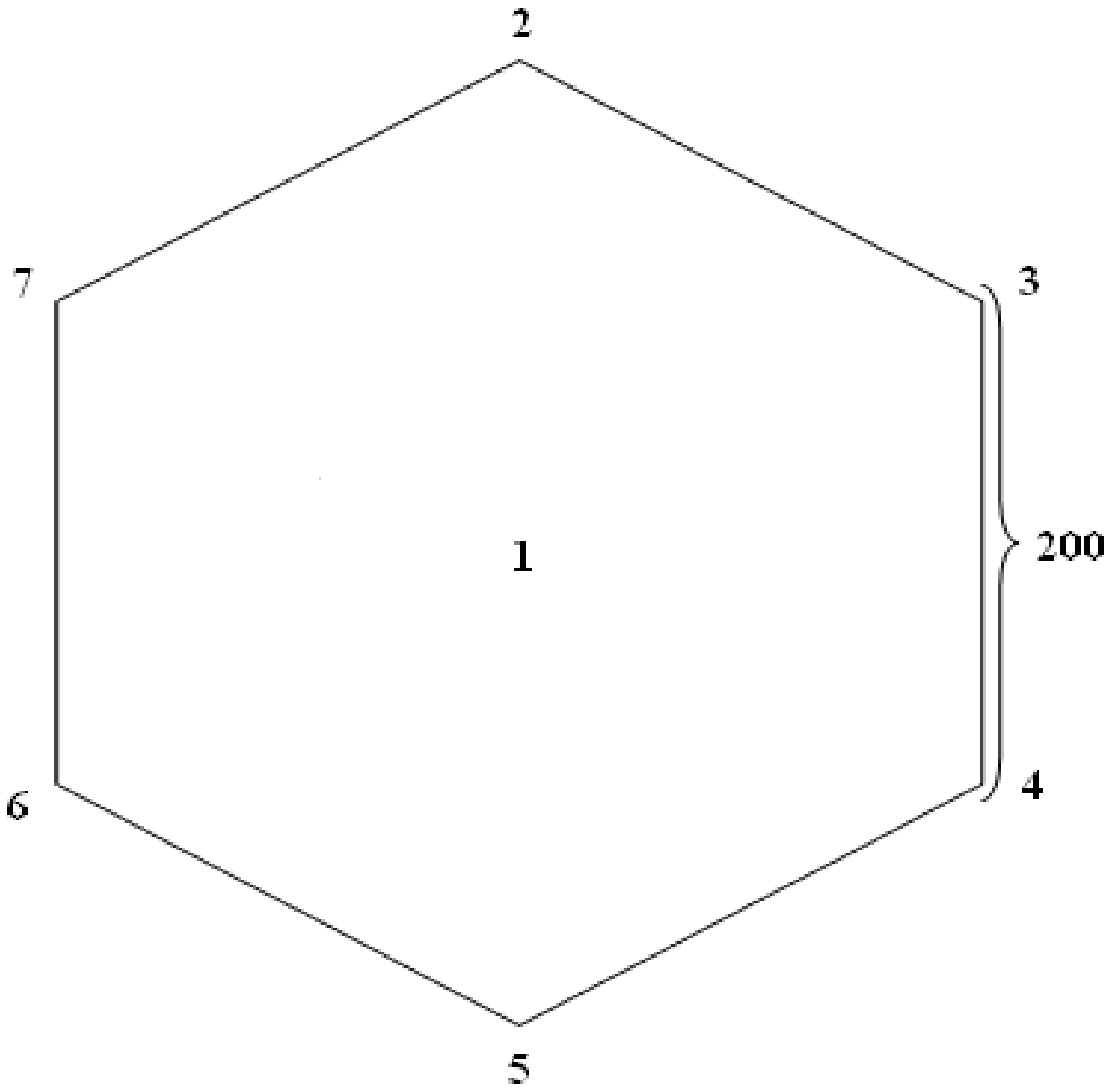
Time				Common Name	Species				# seen	Trans Sect (1-40)		Act.	ID	Comments

Time= Military time. Transect sections: 0-10m=1; 11-20m=2; 21-30m=3; 31-40m=4; 41-50m=5; 51-60m=6; etc.
Act.: Activity: N=nectaring; F=flying; R=resting; O=ovipositing; B=basking; C=courtship; M=mating.
ID: Relative certainty of ID: V=voucher; PV=photo voucher CR=capture & release; GS=good sight; S=sight; PS= poor sight. If photo voucher be sure to include appropriate photo number(s) in comments. If specimen taken be sure to appropriately label.

Corresponding #:____________ Date entered:________ by:_____ Date checked:_______by:___

BUTTERFLY VES LOCATIONS:
PROPERTY:_____OBSERVERS:_____DATE:_____

Draw approximate locations of areas where butterfly VES took place. Be sure to include labels for points of interest such as remnant prairies, fields, woodland clearing, etc.



BUTTERFLY VES: pg____ of_____

Property:_________________________ Observer(s):________________ Date:____________ Visit #:___
Start time:________ End time:_________ Start Temp (F):_______ End Temp (F):_______ %clouds:___
Rain: (Yes/No) Wind spd:____ Disturbance:______________________ Comments: ________________

Habitat area name (Location)	Species				Common Name	Tally	Total Count	ID	Comments

ID: Relative certainty of ID: V=voucher; PV=photo voucher CR=capture & release; GS=good sight; S=sight; PS= poor sight. If photo voucher be sure to include appropriate photo number(s) in comments. If specimen taken be sure to appropriately label. Please fill out approximate location(s) of VES on corresponding map.

Corresponding #:_____________ by:___ Date data entered:_______ Date checked:_______by:___

FIELD CHECKLIST OF IOWA BUTTERFLIES

Date _____ Time _____

Temperature _____ Weather _____

Observers _____

Notes _____

_____

Locations:

1. _____
2. _____
3. _____
4. _____
5. _____

SWALLOWTAILS

___ ___ ___ ___ ___ Pipevine Swallowtail *Battus philenor*
___ ___ ___ ___ ___ Zebra Swallowtail *Eurytides marcellus*
___ ___ ___ ___ ___ **Black Swallowtail** *Papilio polyxenes*
___ ___ ___ ___ ___ **Giant Swallowtail** *Papilio cresphontes*
___ ___ ___ ___ ___ **Eastern Tiger Swallowtail***Papilio glaucus*
___ ___ ___ ___ ___ Spicebush Swallowtail *Papilio troilus*

WHITES AND SULPHURS

___ ___ ___ ___ ___ Checkered White *Pontia protodice*
___ ___ ___ ___ ___ **Cabbage White** *Pieris rapae*
___ ___ ___ ___ ___ Olympia Marble *Euchloe olympia*
___ ___ ___ ___ ___ **Clouded Sulphur** *Colias philodice*
___ ___ ___ ___ ___ **Orange Sulphur** *Colias eurytheme*
___ ___ ___ ___ ___ Southern Dogface *Colias cesonia*
___ ___ ___ ___ ___ Cloudless Sulphur *Phoebis sennae*
___ ___ ___ ___ ___ Mexican Yellow *Eurema mexicana*
___ ___ ___ ___ ___ **Little Yellow** *Eurema lisa*
___ ___ ___ ___ ___ Sleepy Orange *Eurema nicippe*
___ ___ ___ ___ ___ **Dainty Sulphur** *Nathalis iole*

HARVESTERS, COPPERS, HAIRSTREAKS AND BLUES

___ ___ ___ ___ ___ Harvester *Feniseca tarquinius*
___ ___ ___ ___ ___ American Copper *Lycaena phlaeas*
___ ___ ___ ___ ___ **Gray Copper** *Lycaena dione*
___ ___ ___ ___ ___ **Bronze Copper** *Lycaena hyllus*
___ ___ ___ ___ ___ Purplish Copper *Lycaena helloides*
___ ___ ___ ___ ___ Coral Hairstreak *Satyrium titus*
___ ___ ___ ___ ___ Acadian Hairstreak *Satyrium acadica*
___ ___ ___ ___ ___ Edwards' Hairstreak *Satyrium edwardsii*
___ ___ ___ ___ ___ **Banded Hairstreak** *Satryium calanus*
___ ___ ___ ___ ___ Hickory Hairstreak *Satyrium caryaevorum*
___ ___ ___ ___ ___ Striped Hairstreak *Satyrium liparops*
___ ___ ___ ___ ___ Henry's Elfin *Callophrys henrici*
___ ___ ___ ___ ___ Juniper Hairstreak *Callophrys gryneus*
___ ___ ___ ___ ___ White M Hairstreak *Parrhasius m-album*
___ ___ ___ ___ ___ **Gray Hairstreak** *Strymon melinus*
___ ___ ___ ___ ___ Marine Blue *Leptotes marina*
___ ___ ___ ___ ___ Reakirt's Blue *Hemiargus isola*
___ ___ ___ ___ ___ **Eastern Tailed-Blue** *Everes comyntas*
___ ___ ___ ___ ___ **Spring Azure** *Celastrina ladon*
___ ___ ___ ___ ___ Silvery Blue *Glaucopsyche lygdamus*
___ ___ ___ ___ ___ Melissa Blue *Lycaeides melissa*
___ ___ ___ ___ ___ Swamp Metalmark *Calephelis mutica*

BRUSHFOOTS

___ ___ ___ ___ ___ **American Snout** *Libytheana carinenta*
___ ___ ___ ___ ___ Gulf Fritillary *Agraulis vanillae*
___ ___ ___ ___ ___ Variegated Fritillary *Euptoieta claudia*
___ ___ ___ ___ ___ **Great Spangled Fritillary** *Speyeria cybele*
___ ___ ___ ___ ___ Aphrodite Fritillary *Speyeria aphrodite*
___ ___ ___ ___ ___ Regal Fritillary *Speyeria idalia*
___ ___ ___ ___ ___ Silver-bordered Fritillary *Boloria selene*
___ ___ ___ ___ ___ **Meadow Fritillary** *Boloria bellona*
___ ___ ___ ___ ___ Gorgone Checkerspot *Chlosyne gorgone*
___ ___ ___ ___ ___ Silvery Checkerspot *Chlosyne nycteis*
___ ___ ___ ___ ___ **Pearl Crescent** *Phyciodes tharos*
___ ___ ___ ___ ___ Tawny Crescent *Phyciodes batesii*
___ ___ ___ ___ ___ Baltimore Checkerspot *Euphydryas phaeton*
___ ___ ___ ___ ___ **Question Mark** *Polygonia interrogationis*
___ ___ ___ ___ ___ **Eastern Comma** *Polygonia comma*
___ ___ ___ ___ ___ Green Comma *Polygonia faunus*
___ ___ ___ ___ ___ Gray Comma *Polygonia progne*
___ ___ ___ ___ ___ Compton Tortoiseshell *Nymphalis vau-album*
___ ___ ___ ___ ___ **Mourning Cloak** *Nymphalis antiopa*
___ ___ ___ ___ ___ Milbert's Tortoiseshell *Nymphalis milberti*
___ ___ ___ ___ ___ **American Lady** *Vanessa virginiensis*
___ ___ ___ ___ ___ **Painted Lady** *Vanessa cardui*
___ ___ ___ ___ ___ **Red Admiral** *Vanessa atalanta*
___ ___ ___ ___ ___ **Common Buckeye** *Junonia coenia*
___ ___ ___ ___ ___ **Red-spotted Purple** *Limenitis arthemis*
___ ___ ___ ___ ___ **Viceroy** *Limenitis archippus*
___ ___ ___ ___ ___ Common Mestra *Mestra amymone*
___ ___ ___ ___ ___ Goatweed Leafwing *Anaea andria*
___ ___ ___ ___ ___ **Hackberry Emperor** *Asterocampa celtis*
___ ___ ___ ___ ___ **Tawny Emperor** *Asterocampa clyton*
___ ___ ___ ___ ___ **Northern Pearly-eye** *Enodia anthedon*
___ ___ ___ ___ ___ Eyed Brown *Satyrodes eurydice*
___ ___ ___ ___ ___ **Little Wood-Satyr** *Megisto cymela*
___ ___ ___ ___ ___ Common Ringlet *Coenonympha tullia*
___ ___ ___ ___ ___ **Common Wood-Nymph** *Cercyonis pegala*
___ ___ ___ ___ ___ **Monarch** *Danaus plexippus*
___ ___ ___ ___ ___ Queen *Danaus gilippus*

SKIPPERS

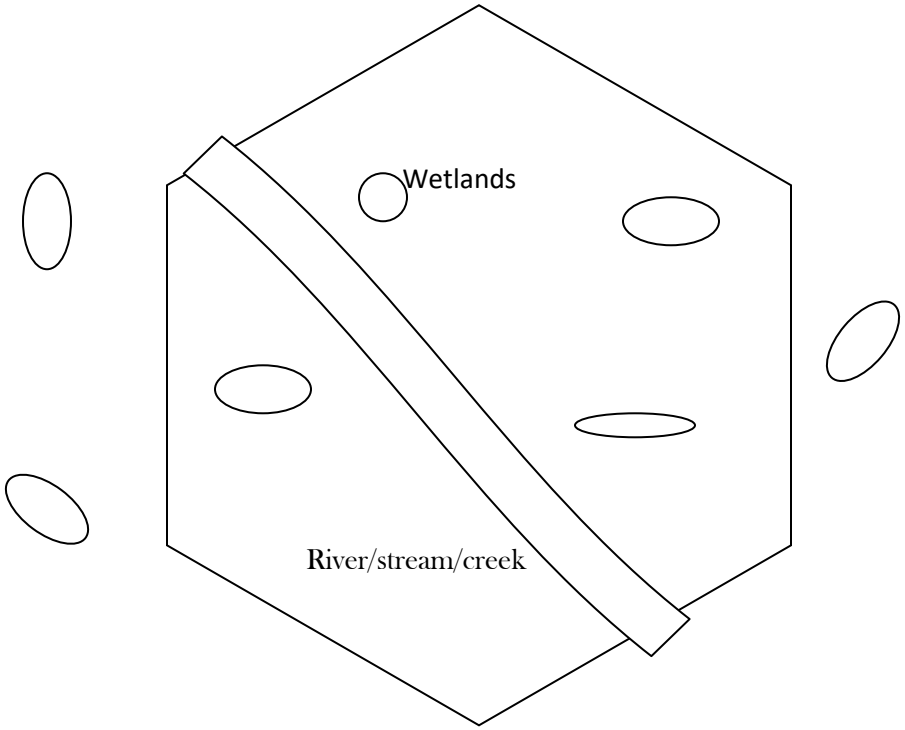
___ ___ ___ ___ ___ **Silver-spotted Skipper** *Epargyreus clarus*
___ ___ ___ ___ ___ Hoary Edge *Achalarus lyciades*
___ ___ ___ ___ ___ Southern Cloudywing *Thorybes bathyllus*
___ ___ ___ ___ ___ Northern Cloudywing *Thorybes pylades*
___ ___ ___ ___ ___ Hayhurst's Scallopwing *Staphylus hayhurstii*
___ ___ ___ ___ ___ Dreamy Duskywing *Erynnis icelus*
___ ___ ___ ___ ___ Sleepy Duskywing *Erynnis brizo*
___ ___ ___ ___ ___ Juvenal's Duskywing *Erynnis juvenalis*
___ ___ ___ ___ ___ Horace's Duskywing *Erynnis horatius*
___ ___ ___ ___ ___ Mottled Duskywing *Erynnis martialis*
___ ___ ___ ___ ___ Columbine Duskywing *Erynnis lucilius*
___ ___ ___ ___ ___ Wild Indigo Duskywing *Erynnis baptisiae*
___ ___ ___ ___ ___ Persius Duskywing *Erynnis persius*
___ ___ ___ ___ ___ Com. Checkered-Skipper *Pyrgus communis*
___ ___ ___ ___ ___ **Common Sootywing** *Pholisora catullus*
___ ___ ___ ___ ___ **Least Skipper** *Ancyloxypha numitor*
___ ___ ___ ___ ___ Poweshiek Skipperling *Oarisma poweshiek*
___ ___ ___ ___ ___ **European Skipper** *Thymelicus lineola*
___ ___ ___ ___ ___ **Fiery Skipper** *Hylephila phyleus*
___ ___ ___ ___ ___ Ottoe Skipper *Hesperia ottoe*
___ ___ ___ ___ ___ Leonard's Skipper *Hesperia leonardus*
___ ___ ___ ___ ___ Dakota Skipper *Hesperia dacotae*
___ ___ ___ ___ ___ Indian Skipper Hesperia sassacus
___ ___ ___ ___ ___ **Peck's Skipper** *Polites peckius*
___ ___ ___ ___ ___ Tawny-edged Skipper *Polites themistocles*
___ ___ ___ ___ ___ Crossline Skipper *Polites origenes*
___ ___ ___ ___ ___ Long Dash *Polites mystic*
___ ___ ___ ___ ___ Whirlabout *Polites vibex*
___ ___ ___ ___ ___ Northern Broken-Dash *Wallengrenia egeremet*
___ ___ ___ ___ ___ Little Glassywing *Pompeius verna*
___ ___ ___ ___ ___ **Sachem** *Atalopedes campestris*
___ ___ ___ ___ ___ Arogos Skipper *Atrytone arogos*
___ ___ ___ ___ ___ **Delaware Skipper** *Anatrytone logan*
___ ___ ___ ___ ___ Byssus Skipper *Problema byssus*
___ ___ ___ ___ ___ Mulberry Wing *Poanes massasoit*
___ ___ ___ ___ ___ Hobomok Skipper *Poanes hobomok*
___ ___ ___ ___ ___ Zabulon Skipper *Poanes zabulon*
___ ___ ___ ___ ___ Broad-winged Skipper *Poanes viator*
___ ___ ___ ___ ___ Dion Skipper *Euphyes dion*
___ ___ ___ ___ ___ Black Dash *Euphyes conspicua*
___ ___ ___ ___ ___ Two-spotted Skipper *Euphyes bimacula*
___ ___ ___ ___ ___ **Dun Skipper** *Euphyes vestris*
___ ___ ___ ___ ___ Dusted Skipper *Atrytonopsis hianna*
___ ___ ___ ___ ___ Pepper and Salt Skipper *Amblyscirtes hegon*
___ ___ ___ ___ ___ Com. Roadside-Skipper *Amblyscirtes vialis*
___ ___ ___ ___ ___ Eufala Skipper *Lerodea eufala*

Chapter Thirteen
Damselfly and Dragonfly Monitoring Protocol

DRAGONFLY AND 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. Approriate habitats within the property should be surveyed. This area may be much larger than that usually incorporated into a VES, 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-km^2 (~250 ac) area around the center point. Stay within the property boundaries.

SURVEY METHODS:

Field Methods

All wetland habitats should be searched for both adult odonates and discarded exuviae. 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). 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 September 11, 2017) (http://www.iowaodes.org) 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 microhabitat (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 micro-habitat, 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 plot, but it is important to create a record of relative abundance indices based on the smaller microhabitat locations in addition to the overall site abundance. However, we tally the number of individuals seen, attempting to make every effort NOT to count the same individual more than once.

In addition to the adult abundance counts, 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.

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 10 seconds (or longer). 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. The acetone does more than kill the insects; it dries them out to preserve them. Place them in acetone and leave them overnight. Some of the larger dragonflies may need to be left a little longer. After about 5 minutes (long enough to make sure the individual is dead), straighten the body and wings so the specimen is in a good shape. In the

morning, pull them out of the acetone and let them dry during the day. Use an envelope of paper triangle to keep their wings flat. Individuals with pruinessence (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).

Lab Methods

The exuviae collected in the field need to be identified to species in the lab using field guides or keys and a dissecting scope. Any larval odonates that were collected during the aquatic invertebrate sampling can be identified at this time as well.

EQUIPMENT NEEDED:

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 digital cameras, 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 PRESENCE or MARK. The data collected with this technique will be used to compute abundance indices when possible.

SAFETY ISSUES & CONSIDERATIONS:

The odonate technicians may 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 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 www.iowaodes.org . Appendix 1 contains a list of additional, more common, odonate species which may be encountered during the monitoring efforts. An "*" indicate odonates designated as Data Deficient.

Target dragonflies & damselflies species:

Common Name	Scientific Name	Habitat
Spotted Spreadwing	*Lestes congener*	Edge of pools, marsh
Amber-winged spreadwing	*Lestes eurinus*	Fishless shallow ponds and wetlands
Sweetflag spreadwing	*Lestes forcipatus*	Marsh, pond edge
Paiute dancer	*Argia alberta*	Small streams, road ditches
Springwater dancer	*Argia plana*	Small, shallow streams with canopy cover and clay substrate
*Prairie bluet	*Coenagrion angulatum*	Lakes, ponds
*Taiga bluet	*Coenagrion resolutum*	Ponds & wetlands
*Boreal bluet	*Enallagma boreale*	Marsh
*Alkali bluet	*Enallagma clausum*	Pond edges without vegetation
*Western forktail	*Ischnura perparva*	Heavily vegetated ponds, lakes, and slow flow streams with mud substrate
Sedge sprite	*Nehalennia irene*	Ponds & sedge meadows
*Canada darner	*Aeshna canadensis*	Marsh, pond edge
*Variable darner	*Aeshna interrupta*	Lakes, ponds, streams
Midland clubtail	*Gomphus fraternus*	Creaks & rivers with rock & mud
Sulpher-tipped clubtail	*Gomphus militaris*	Artificial ponds, lakes
*Rapids clubtail	*Gomphus quadricolor*	Rocky creeks
Rusty snaketail	*Ophiogomphus rupinsulensis*	Sandy, rocky creeks
*Pale snaketail	*Ophiogomphus severus*	Privers & streams with fast flow & cobble substrate

Sioux snaketail	*Ophiogomphus smithi*	Sand-bottomed creeks
*Westfall's snaketail	*Ophiogomphus westfalli*	Clear forest streams with strong riffles & cobble substrate
*Brimstone clubtail	*Stylurus intricatus*	Sandy streams
Elusive clubtail	*Stylurus notatus*	Creeks & rivers with sandy substrate
*Arrow clubtail	*Stylurus spiniceps*	Rivers with sandy substrate
Stream cruiser	*Didymops transversa*	Medium to large streams & rivers
Royal river cruiser	*Macromia taeniolata*	Lakes, rivers
Slender baskettail	*Epithera costalis*	Lakes, ponds, and backwaters
Smoky shadowdragon	*Neurocordulia molesta*	Large rivers
*Stygian shadowdragon	*Neurocordulia yamaskanensis*	Mississippi River
Plains emerald	*Somatochlora ensigera*	Prairie streams
Carolina saddlebags	*Tramea Carolina*	Marsh

ADDITIONAL METHODS FOR SPECIAL LOCATIONS:

None

DRAGONFLY AND DAMSELFLY VES:

pg____ of______

Property:________________________ Observer(s):________________ Date:___________ Visit #:_____
Start time:________ End time:_________ Start Temp (F):_______ End Temp (F):______ %clouds:_____Rain:
(Yes/No) Wind spd:____ Disturbance:_____________________ Comments: ____________________

Habitat area name (Location)	Species				Common Name	Tally	Total Count	ID	Comments

ID: Relative certainty of ID: V=voucher; PV=photo voucher CR=capture & release; GS=good sight; S=sight; PS= poor sight. If photo voucher be sure to include appropriate photo number(s) in comments. If specimen taken be sure to appropriately label. This information should be entered in the comments section of the data base.
Corresponding #:_____________ by:___ Date data entered:_______ Date checked:_______by:___

Chapter Fourteen
Terrestrial Snail Monitoring Protocol

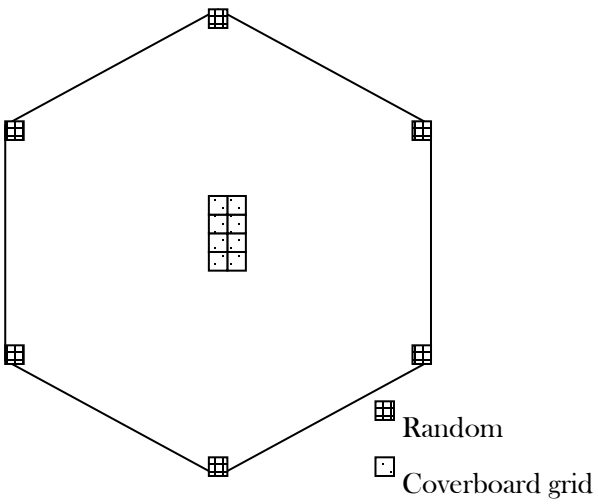
There are no protocols for these species in the USFS MSIM. However, the US Fish and Wildlife Service has developed a protocol for monitoring the Iowa Pleistocene snail (Henry et al. 2003) which has been adapted below to find terrestrial snails of other species in additional habitats. The Iowa WAP (Zohrer 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 can 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 hectare) 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).

Figure 2: Grid pattern on each of the coverboards placed at each hexagonal point.

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 as seen in Figure 3. All coverboards should be soaked in water before being deployed – preferably in water on the sampling site (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 NEEDED:

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 species occurrence data can be analyzed using Program PRESENCE (MacKenzie et al. 2002) or the 'occupancy estimation' or 'robust design occupancy' data type choices in Program MARK (White and Burnham 1999) which will calculate probability of detection estimates and proportion of points occupied. Given the distance between the coverboards (about 200 m), the

Figure 2: 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

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 meters 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. This can be done with Program MARK as well. See Chapter 5 (Data Analysis) for additional information on these techniques.

SAFETY ISSUES & CONSIDERATIONS:
Proper hygiene should be followed after handling snails.

TARGET SPECIES:
Target snail species:

Common Name	Scientific Name	Habitat
Iowa Pleistocene snail	*Discus macclintocki*	Algific slopes
Frigid ambersnail	*Catinella gelida*	Algific slopes
Minnesota Pleistocene snail	*Novasuccinea n. Sp. minnesota a*	Moderate slopes
Iowa Pleistocene succinea	*Novasuccinea n. Sp. minnesota b*	Moderate slopes
Briarton Pleistocene snail	*Vertigo brierensis*	Algific slopes
Hubricht's vertigo	*Vertigo hubrichti*	Algific slopes
Iowa Pleistocene vertigo	*Vertigo iowaensis*	Algific slopes
Bluff vertigo	*Vertigo occulta*	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.

TERRESTRIAL SNAIL DATASHEET:

DATE:__________________ OBS:____ pg____of____

PROPERY:________________________ VISIT #:_______ Start time:_______ End time:_______

Air Temp (°F):______ Soil Temp (°F):______ %clouds:_______ Rain:________ Wind spd:_______

Comments:____

Lo=Location=center point (C) or outer edge (O) of hexagon. Bo #=Board #. Rc=Recaptute=Y or N. Aperture = shell opening (where snail body goes). All measurements are in mm.

Lo	Bo #	Grid #	Species				Mark color and number				Rc	Shell height (mm)		Shell width (mm)		Aperture height (mm)		Aperture width (mm)		Comments

Data entered: date:_____by:___Record #s:_________date checked:_____by:____

Chapter 15
Fish Monitoring
Wade-able 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 AND 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 is also 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. 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.

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 meters or more will need to be sampled.

Regardless of the amount of stream occurring within the plot, a 150 meter reach is the minimum that should be sampled. So, if only 50 meters 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.

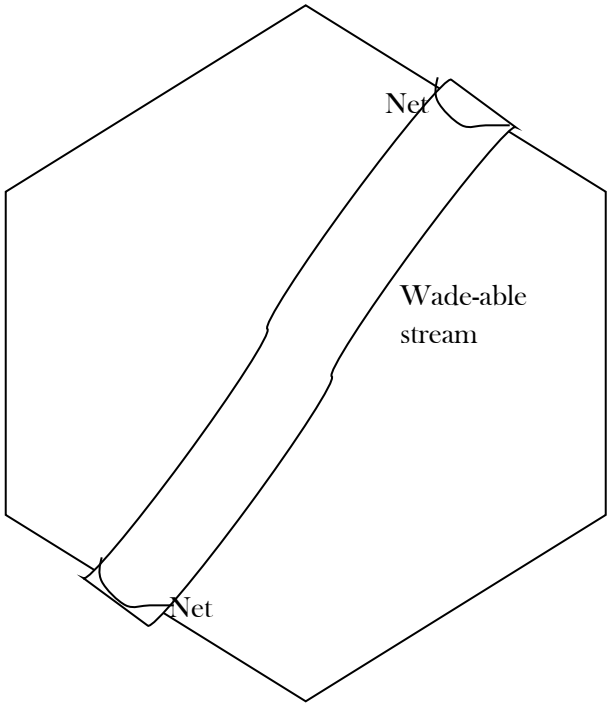
SURVEY METHODS:

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 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.
2). Collect water samples for physicochemical water quality parameters.
3). Collect semi-quantitative benthic macroinvertebrate samples.
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 Hygenics Laboratory.

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.

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.

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 electro-fishing 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. 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.

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.

Seining

Seining may be the most efficient method to sample small fishes (e.g. redfin 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). This issue can be addressed during the first few years of the monitoring program. 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 meters. 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 site, 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.

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

ENVIRNMENTAL 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 (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 NEEDED:

GPS unit
Water collection jars
Binoculars
Dip nets

Block nets
Twine for repairs to blocknets and seine nets
Backpack electrofishing units
Extra batteries and gas:oil mix for Backpack units
Tow boat if needed
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

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

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. 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 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.

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 points occupied using program PRESENCE or program MARK. 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. The sampling

design for fish in wadeable streams may affect the potential analysis of the data. For additional information on the PAO techniques, see Chapter 5 (Data Analysis).

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:**

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

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

SAFETY 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) and also in Iowa AQUATIC GAP (http://www.cfwru.iastate.edu/IAGAP_final_report.pdf). Appendix 1 contains a list of all fish species known to occur in Iowa which may also be encountered during the monitoring efforts. An "*" indicated fish designated as Data Deficient.

Target species:

Common Name	Scientific Name	Habitat
Chestnut lamprey	*Ichthyomyzon castaneus*	Mississippi and Chariton rivers
Northern brook lamprey	*Ichthyomyzon fossor*	Northeast 1/4
Silver lamprey	*Ichthyomyzon unicuspis*	Mississippi River
American brook lamprey	*Lampetra appendix*	Northeast 1/4
Lake sturgeon	*Acipenser fulvescens*	Mississippi River
Pallid sturgeon	*Scaphirhynchus albus*	Missouri River
Shovelnose sturgeon	*Scaphirhynchus platorynchus*	Mississippi and Missouri Rivers
Paddlefish	*Polydon spathula*	Mississippi, Missouri, Des Moines, Iowa, Cedar, and Skunk rivers
American eel	*Anguilla rostrata*	Mississippi and Missouri Rivers & larger tributaries
Skipjack herring	*Alosa chrysochloris*	Mississippi and Missouri Rivers
Largescale stoneroller	*Campostoma oligolepsis*	NE 2/3
Gravel chub	*Erimystax x-punctatus*	Large rivers and streams
Western silvery minnow	*Hybognathus agryritis*	Missouri drainage
Mississippi silvery minnow	*Hybognathus nuchalis*	Mississippi drainage
Plains minnow	*Hybognathus placitus*	Missouri drainage
Pallid shiner	*Hybopsis annis*	Upper Mississippi River
Redfin shiner	*Lythurus umbratilis*	Northeast 1/4
Shoal chub	*Macrhybopsis hyostomus*	Large, interior rivers, statewide
Sturgeon chub	*Macrhybobsis gelida*	Eastern ½ Missouri River
Sicklefin chub	*Macrybopsis meeki*	Missouri River
Pearl dace	*Margariscus margarita*	Worth county
Golden shiner	*Notemigonus crysoleucas*	
Pugnose shiner	*Notropis anogenus*	West Lake Okojobi
River shiner	*Notropis blennius*	
Silverband shiner	*Notropis shumardii*	Mississippi & Missouri(?) River
Ghost shiner	*Notropis buchanani*	Mississippi River
Blacknose shiner	*Notropis heterolepis*	NW
Ozark minnow	*Notropis nubilus*	NE ¼
Carmine shiner	*Notropis percobromus*	NE ¼
Weed shiner	*Notropis texanus*	Cedar & Mississippi Rivers
Topeka shiner	*Notropis topeka*	W ¾

Target species continued:

Common Name	Scientific Name	Habitat
Mimic shiner*	*Notropis volucellus*	
Channel shiner*	*Notropis wickliffi*	Iowa River
Pugnose minnow	*Opsopoeodus emiliae*	Mississippi River
Suckermouth minnow	*Phenacobius mirabilis*	Upper Des Moines basin
Southern redbelly dace	*Phoxinus erythrogaster*	NE 1/3, NW ¼
Flathead chub	*Platygobio gracillis*	Missouri drainage
Longnose dace	*Rhinichthys cataractae*	NE corner
Blue sucker	*Cycleptus elongates*	Mississippi and Missouri Rivers & larger tributaries
Lake chubsucker*	*Erimyzon succetta*	
Black buffalo	*Ictiobus niger*	Mississippi River & large tributaries
Spotted sucker	*Minytrema melanops*	Mississippi River
Silver redhorse	*Mocostoma anisurum*	
River redhorse	*Moxostoma carinatum*	Upper pools of Mississippi
Black redhorse	*Moxostoma duquesnei*	Turkey & upper Iowa river drainages
Brown bullhead	*Ameiurus nebulosus*	N 1/3
Blue catfish	*Ictalurus furcatus*	Lower Mississippi & Missouri Rivers
Slender madtom	*Noturus exilis*	Mississippi River tributaries
Tadpole madtom	*Noturus gyrinus*	Statewide
Freckled madtom	*Noturus gyrinus*	Mississippi River & large tributaries
Grass (Redfin) pickerel	*Esox americanus*	Missouri River & tributaries
Northern pike	*Esox lucius*	
Central mudminnow	*Umbra limi*	N 1/3
Brook trout	*Salvelinus fontinalis*	NE corner
Trout perch	*Percopsis omiscomycus*	NW ¼; Upper Mississippi River, Grand & Chariton Rivers
Pirate perch	*Aphredoderus sayanus*	Mississippi River & large tributaries
Burbot	*Lota lota*	Missouri River, Mississippi River & tributaries
Brook silverside	*Labidesthes sicculus*	East
Banded killifish	*Fundulus diaphanous*	Natural lakes in NW; Missouri River
Starhead topminnow	*Fundulus dispar*	L. Sioux, Iowa Rivers
Blackstripe topminnow	*Fundulus notatus*	E 1/3
Plains topminnow	*Fundulus sciadicus*	Rock River Basin
Mottled sculpin	*Cottus bairdi*	Lower Bear Creek
Slimy sculpin	*Cottus cognatus*	NE corner
Rock bass	*Ambloplites rupestris*	
Longear sunfish[t]	*Lepomis megalotis*[t]	
Northern sunfish[t]	*Lepomis peltastes*	
Western sand darter	*Annicrypta clara*	Mississippi River
Crystal darter	*Crystallaria asprella*	Mississippi & Turkey Rivers
Mud darter	*Etheostoma asprigene*	Mississippi River & tributaries

Target species continued:

Common Name	Scientific Name	Habitat
Rainbow darter	*Etheostoma caeruleum*	Cedar River basin
Bluntnose darter*	*Etheostoma chlorosomum*	Mississippi River
Iowa darter	*Etheostoma exile*	Northern ½
Least darter	*Etheostoma microperca*	Maquoketa, tributary to Otter Creek
Orangethroat darter*	*Etheostoma spectabile*	SE ¼
Banded darter	*Etheostoma zonale*	NE ¼
Northern logperch	*Percina caprodes*	Mississippi drainage, Clear Lake
Blackside darter	*Percina maculate*	Mississippi River
Slenderhead darter	*Percina phoxocephala*	Mississippi drainage
River darter	*Percina shumardi*	Mississippi River

[1]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 breath. Traps should be checked daily and left in the water for 3 to 5 days.

Iowa DNR MSIM **Backpack Electrofishing Datasheet** **Page** _____ **of** _____

LOCATION _____ **WATER BODY** _____

Survey Type E F **Taxa Group** F I S H **Observers** _____

Survey Date ☐☐ / ☐☐ / ☐☐☐☐ **Start Time** _____ : _____

End Time _____ : _____

% Cloud Cover ☐☐☐ **Rain (Y/N)** ___ **Wind Speed (mph)** ☐☐

WYPT	UTMN	UTME
Start		
Stop		

Conductivity (micro S/cm) _____

Secchi (m) _____ . _____

Flow (m/s) _____ . _____

Volts ☐☐☐ **Amps** ☐ . ☐ **Waveform** P U L S E D ☐ D C

Anomaly Codes: D = defdormities, E=eroded or frayed fins, L=Lesions or ulcers, T=tumors, M=multiple DELTS on same fish, AL=anchor worm-light, AH=anchor worm-heavy, BL=blackspot-light, BH=blackspot-heavy, CL=leeches-light, CH=leeches-heavy, F=fungus, I-Ich, N=blind, S=emaciated, P=external parasites, Y=popeye, W=swirled scales, Z=wound, other (describe).

	Count (Tally)									
Species	**0-3"**	**4-6"**	**7-9"**	**10-12"**	**13-15"**	**16-18"**	**19-21"**	**22"+**	**Anomaly Code**	**# Affected**

Date data entered ☐☐ / ☐☐ / ☐☐☐☐ **Date Checked** _____ / _____ / _____

Entered by ☐☐ **Record #** ☐☐☐☐ **Checked by** ☐☐

Comments:

Anomaly Codes: D = deformities, E=eroded or frayed fins, L=Lesions or ulcers, T=tumors, M=multiple DELTS on same fish, AL=anchor worm-light, AH=anchor worm-heavy, BL=blackspot-light, BH=blackspot-heavy, CL=leeches-light, CH=leeches-heavy, F=fungus, I-Ich, N=blind, S=emaciated, P=external parasites, Y=popeye, W=swirled scales, Z=wound, other (describe).

Photo Voucher List

Species	Photo Numbers/Range/Description

Date data entered ☐☐ / ☐☐ / ☐☐☐☐ **Date Checked** _____ / _____ / _____

Entered by ☐☐ **Record #** ☐☐☐☐☐ **Checked by** ☐☐

Chapter Sixteen
Fish Monitoring
Lakes

The Fisheries Section 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.

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 (http://www.innovativenetsystems.com/) 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 NEEDED:

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 points occupied using program PRESENCE

or Program MARK. For additional information on the PAO techniques, see Chapter 5 (Data Analysis).

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

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

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

SAFETY 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 Seventeen
Fish Monitoring
Large Rivers

The Fisheries Section 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.

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) (Angardi 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 (http://www.innovativenetsystems.com) 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 Neilsen 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 6. 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 NEEDED:

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:

8. Species composition
9. Species relative abundance (i.e., the number of fish of each species as a percentage of the total number of captured fish)
10. Fish abundance (i.e., catch per unit effort)
11. Proportion of fish with external abnormalities.

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

SAFETY 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.

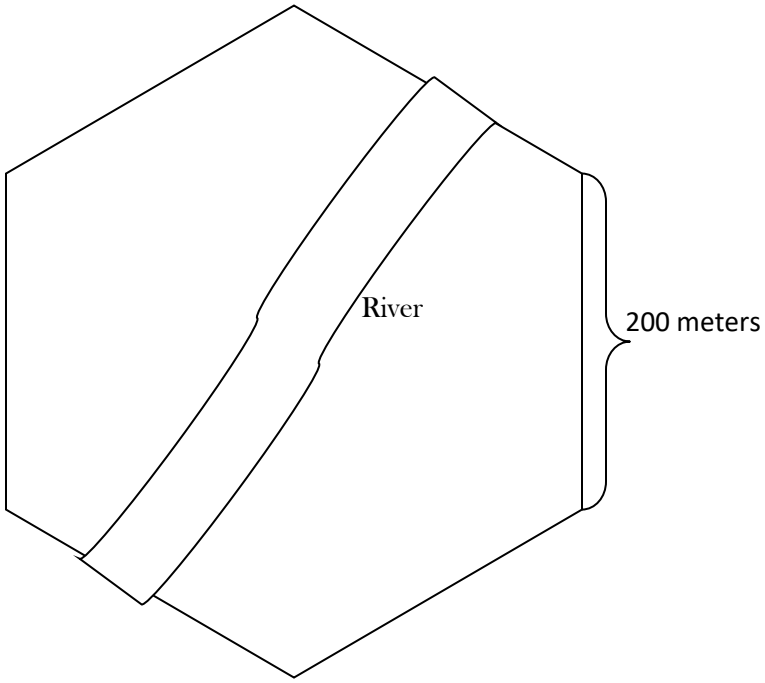
TARGET SPECIES:

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 Eighteen
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.



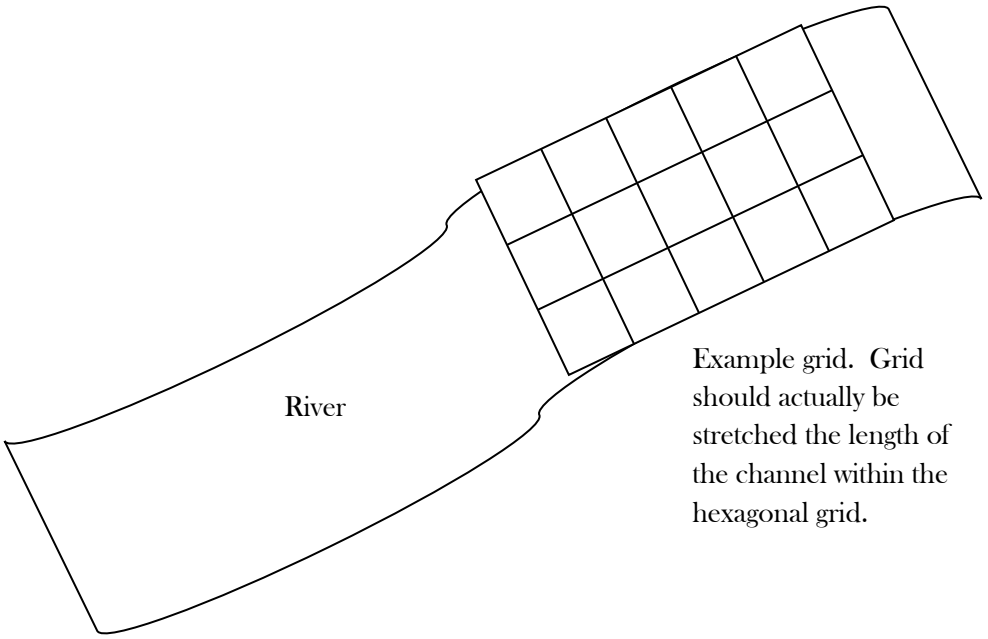
SURVEY METHODS:

Surveys are to be conducted between the beginning of August and the end of September (8 weeks and 5 days). This time frame will allowed 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 m^2. 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-meter increments along the stream side as a reference guide. Marking 1-meter 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 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 meter 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 meters 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. 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:

$$q = (L*W) \,/\, (n/k)$$

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	25	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 m^2 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, or Spectaclecase 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 m^2 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 meters upstream or >40 meters 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.

ENVIRNMENTAL 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 malecologist. 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 sites occupied using program PRESENCE or program MARK. For additional information on the PAO techniques, 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 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.

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

*Federally listed species.

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.

Mussel quadrat data sheet. DATE:_______ OBS:____________ LOCATION:________________ WATER BODY NAME:__________
GPS Coordinates of downstream starting point:____ START TEMP:______END TEMP:______
Rain:______ % CLOUDS:______ STREAM SECTION (1 or 2):_________________ Always have at least 2 data sheets (1/stream section)
Quad #__:Start Time:____End time:____ Q#__:ST:____ET:____ Q#__:ST:____ET:____ Q#__:ST:____ET:____ Q#__: ST:____ ET:____
Q#__:ST:____ET:____ Q#__:ST:____ET:____ Q#__:ST:____ET:____ Q#__:ST:____ET:____ Q#__:ST:____ET:____
Quad=quadrat #; length in mm; S=Sex

Quad			Species						Length			S

Quad			Species						Length			S

Quad			Species						Length			S

Date entered:______by:_____Rec.#:____

Date checked:______by:__

Chapter Nineteen
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.

Crayfish contribute to the biodiversity of aquatic habitats, particularly in the southeastern United States where a number of species are endemic. Approximately 77% of the world's 500-plus species of crayfish occur in North America (Taylor et al. 2007). Crayfish are important both ecologically and economically, serving as predators in aquatic food webs, bio-processors of aquatic biomass, and as a critical food resource for fishes and other vertebrates (Dorn et al 2005), as well as supporting the human food industry (Huner 2002). Little is known about crayfish in Iowa, thus emphasizing the importance of developing monitoring protocols for this taxon.

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 passive sampling methods for crayfish and, overall, providing 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 monitoring (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. Properties will potential habitat for crayfish will be trapped once during spring, summer, and fall survey seasons.

Minnow traps will be set in water bodies near aquatic habitats suitable for crayfish (e.g. near obvious burrow colonies, dense aquatic vegetation, or areas with mud substrate). 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 meters 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. Traps will be baited with fish (i.e. sardines, carp, or other raw fish) because we currently use bait when trapping amphibians and reptiles and because fish is an effective bait for crayfish (Somers and Stechy 1986).

Burrowing crayfish nets are constructed from discarded avian mist nets and measure approximately 20 by 150 cm (Welch and Eversole 2006). Mish 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 by 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 (Figure 1). 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).

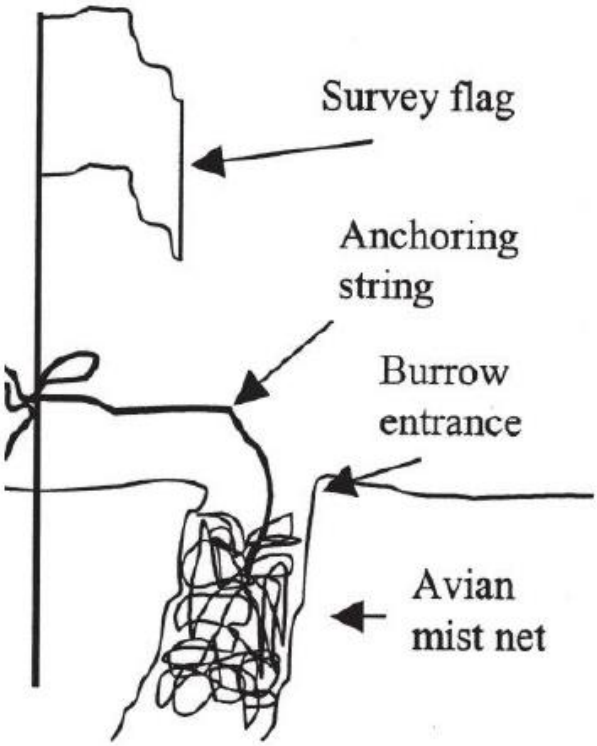


Figure 1. Schematic of the burrowing crayfish net from Welch and Eversole (2006).

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. If burrows and burrow colonies are located within 3 meters or a shoreline, both minnow traps and burrowing crayfish nets will be deployed in the area. If burrows and burrow colonies are located >3 m from a shoreline, only burrowing crayfish nets will be deployed and minnow traps will be deployed in the nearest aquatic habitat. 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 and the carapace length will be measured to the nearest 0.1 mm using a dial caliper. 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 received and store sperm. If a crayfish cannot be identified in the field, it will be collected and preserved in 10% formalin or 70% ethanol solution for later identification. If more than one individual of a presumed species is captured, only 1 should be collected for identification purposes.

Each individual crayfish will be marked when first captured to determine capture efficiency and recapture rates. Crayfish will 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; figure 2). 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. See appendices for a list of all possible mark combinations using the telson/uropod marking method.

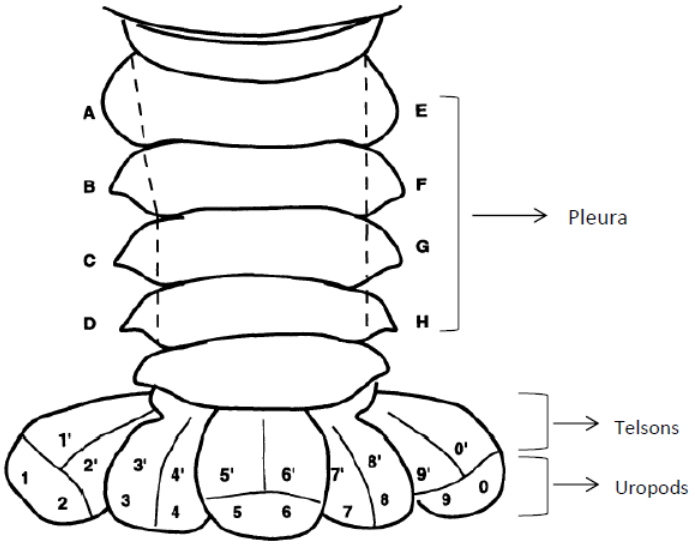


Figure 2: Diagram of the telsons, uropods, and pleura used for individually marking crayfish (Guan 1997).

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 (^{o}F), wind speed (miles/hour), and water temperature (^{o}F). Thes measurements are recorded on the datasheet.

EQUIPMENT LIST:

Weather kestrel temperature and wind gauge
Water thermometer
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*	*Cambarus diogenes*	Streams & creeks
Calico Crayfish*	*Orconectes immunis*	Sloughs & floodplains
Golden Crayfish*	*Orconectes luteus*	Rivers, streams, creeks
Northern Clearwater Crayfish*	*Orconectes propinquus*	Cool water streams
Virile Crayfish*	*Orconectes virilis*	Rivers, streams, and ponds lacking predatory fish
White River Crayfish*	*Procambarus acutus*	
Prairie Crayfish*	*Procambarus gracilis*	Grasslands

CRAYFISH TRAP DATA SHEET:

Property:_____ Date:_____ Observer(s):_____ Visit #:_____ pg____ of____
Start time:_____ End time:_____ Start temp:_____ End temp:_____ %clouds:_____Rain: (Yes/No) Wind speed:_____ Water Temp:_____

Area corresponds to the map of the location being surveyed. Each map should have wetlands numbered. If the area being trapped or where an animal is caught is not marked on the map, record approximate location. Trap type= BN=burrowing crayfish net; MM=metal minnow trap.
Sex: M/F/U. Photo #=corresponding photo number/s for voucher.

Area	Trap type		Trap #	Species				Common name	# caught	Mark ID	Recap (Y/N)	Carapace length (0.1 mm)	Sex	Age	Photo #	Comments

Please remember to list any incidental animal sightings on the back or in the margins of this data sheet.
Corresponding #:_____ Date data entered:_____ Entered by:____ Date checked:_____ Checked by:____

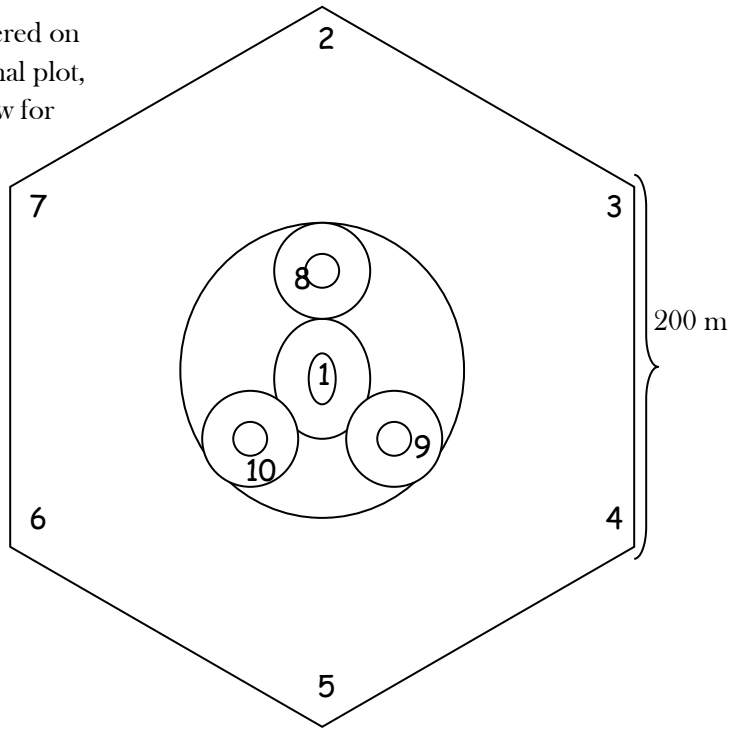
Chapter Twenty
Terrestrial Plant Species and Habitat Classification Monitoring

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 AND MONITORING:

Within each of the permanent hexagonal sampling sites, 4 plots could be established in the center of the site as diagramed below. Additional plots will be established at each of the bird point count locations, i.e. at each point of the hexagon. This would result in a total of 10 possible vegetative plots per hexagon/sampling site. Eight of the plots will be sampled each year. 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 with hexagon is centered.

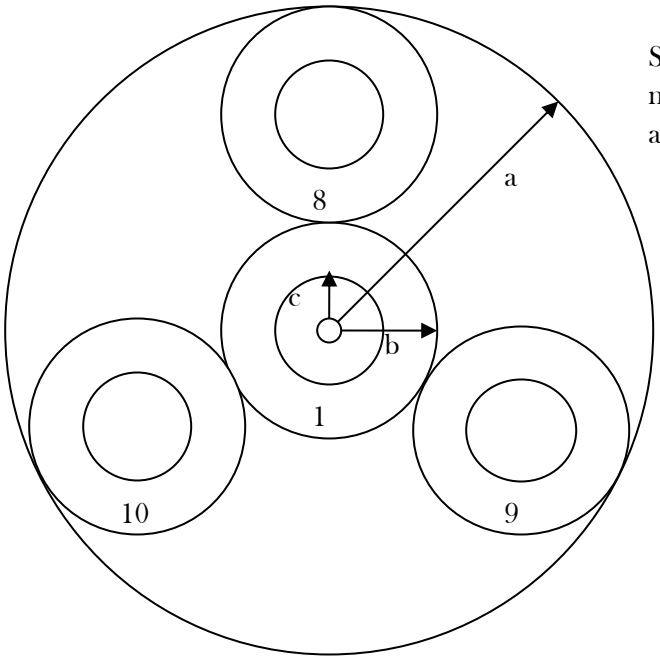
Nested plots centered on middle of hexagonal plot, see diagrams below for additional information.



To find with GIS, the computations for finding the center of plots labeled 8, 9, and 10 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 subplots centered around the center of the hexagonal sampling plot.



Subplots are numbered 1, 8, 9, and 10.

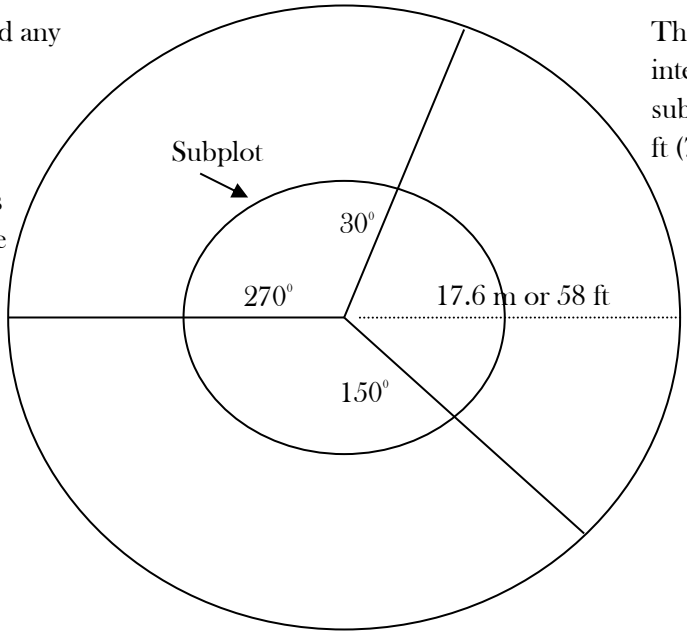
Distances:

a = 56.4 m

Within & around any given subplot

---- woody debris transects, choose

1 per any given subplot.



The radius of the interior (true subplot) plot is 24 ft (7.3 m).

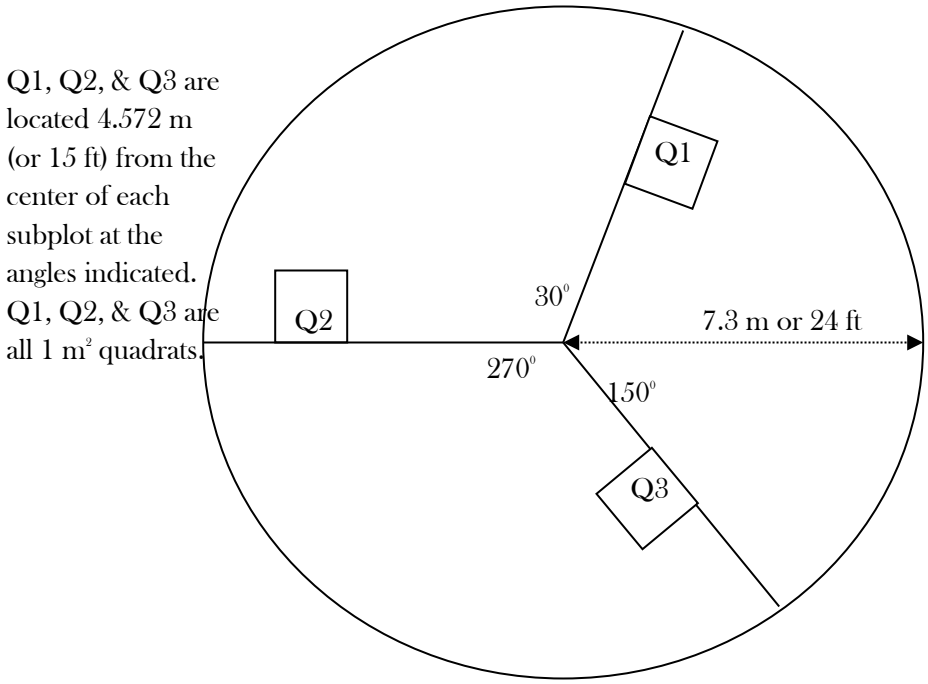


Diagram of the interior of any one of the subplots associated with the larger plant composition monitoring area.

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.

Plot Layout

The first subplot 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 subplot 1 at angles of 120⁰, 240⁰, and 360⁰, respectively. Within each of the 10 subplots, 2 of the potential 3 - 1 m² quadrats are sampled approximately 4.572 m (15 ft) from the center of the subplot. The location of each quadrat should be permanently marked to facilitate future measurements using a GPS unit. Within each hexagonal sampling area, a total of 16 to 30 quadrats will be measured for plant composition depending on time constraints.

Within each of the 8 or 10 subplots, 1 woody debris transect (extending 17.6 m or 58 ft) should be established in 1 of the 3 pre-determined aspects (30⁰, 150⁰, 270⁰). Each transect is first marked in a straight line (using a hip chain or a surveyors tape) as it is critical not to bias the measurements by moving the line to include (or exclude) logs.

If time is limited, data should still be collected from at least 2 of the 3 quadrats for 8 of the 10 possible subplots (always subplots 1-7 & one of 8, 9, or 10). Additional information should be collected in the remaining quadrats as time allows. In each subplot location, the decision as to which of the 2 quadrats are to be surveyed should be random and should change between plots.

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 mid-August).

INTERIOR SUBPLOT MEASUREMENTS: (within each 0.017 ha (7.3 m or 23 ft) subplot):

Litter Depth

At 3 locations (2.5, 5, & 7.5 m) along one of the transects, litter depth is measured and recorded. Care should be taken to ensure areas that have been disturbed by the animal trapping and searching efforts is avoided. Please record the direction of the transect. This direction should be either 30^0, 150^0, or 270^0 and should be chosen from among these 3 at random.

General Plant List

Within each 0.08 acre (0.3 hectare) 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 in 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 in such a way as to allow the specimen collected and pressed to be easily matched to the data recorded on the data sheet.

Quadrats

For the quadrat measurements, the 1-m^2 frame is positioned on the ground at the correct location. 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 all vascular plants that are within each quadrat. Plants that are living and plants that have died within the given year should be included. Quadrat cover could only exceed 100% if plant 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% or < 1%. Other categorical percentage classes are: 1-5%, 6-25%, 26-50%, 50-75%, 75-99%, and 100%. If bare soil or water or rock is located within the quadrat, this should be noted on the data sheet. All matter within the quadrat should be included in the summation to 100%.

Also within each quadrat, a 'trampling code' is assigned to quantify damage by humans or wildlife. A trampling code 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 A. Unknown plants should be pressed in a plant press before leaving the property to ensure that all of the unknowns are there (not accidentally dropped in the field).

Canopy Cover

Using a densitometer, 4 canopy cover estimates (yes or no for canopy cover) are made around the perimeter of the 0.017 hectare plot in the 4 cardinal directions.

SUBPLOT (within each 0.1 hectare (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

Tree species, diameter at breast height (cm), and height to nearest m are recorded for all trees and snags $\geq$ 12.5 cm (5 in) in diameter. Decay class is also estimated 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
3	Limb stubs	Broken	Varies
4	Few or no stubs	Broken	Varies
5	None	Broken	< 20%

Vertical Vegetation/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-m^2 quadrat location. The other 2 1-m^2 quadrats were used to estimate the amount of individual vegetative species covers, this 3rd plot will be used for vertical obstruction. Place the pole in the middle of the plot and move to

the end of the attached rope (approximately 4 meters). Squat down until the eye is 1 meter 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.

Ground Cover Percent

As a comparison for the plant composition subplot estimates, every 5 m along the woody debris transects, the ground cover percent will be estimated for a 1 m stretch. 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 m^2 plot centered at the point on the transect corresponding to 5, 10, and 15 m from the center point of the subplot.

EQUIPMENT LIST:

Plant press, cardboard, and newspaper to collect unknowns (this could be left in truck, but plants should be labeled and pressed before site is left).
Unknown/collected plant labels
1 m^2 quadrat sampling frame
Hand lens
Field guides & species lists
Stopwatch
Folding hand trowel
Hip chain or surveyors tape
Robel pole
Meter stick
Masking tape
Bags (garbage, shopping, or gallon) to carry voucher plants
Standard field kit: Clip board, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets.
Dissecting scope at lab or office

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, 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. However, as PAO methods are concerned with detecting the proportions of area occupied as well as trends in occupancy rates, we could use program PRESENCE to determine occupancy probabilities for plant species of interest, depending on the quantity of the data collected.

SAFETY 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:

<u>To Monitor Vegetation</u> (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.

Suggested labels for unknown and collected plants:

Species ID in field:_________________ Hexagon plot ID:__________________ Subplot ID:_______________________ Quadrat ID:______________________ Habitat code (for quadrat):___________ Percent cover:_____________________ Collector's initials:_________________	Species ID in field:_________________ Hexagon plot ID:__________________ Subplot ID:_______________________ Quadrat ID:______________________ Habitat code (for quadrat):___________ Percent cover:_____________________ Collector's initials:_________________
Species ID in field:_________________ Hexagon plot ID:__________________ Subplot ID:_______________________ Quadrat ID:______________________ Habitat code (for quadrat):___________ Percent cover:_____________________ Collector's initials:_________________	Species ID in field:_________________ Hexagon plot ID:__________________ Subplot ID:_______________________ Quadrat ID:______________________ Habitat code (for quadrat):___________ Percent cover:_____________________ Collector's initials:_________________
Species ID in field:_________________ Hexagon plot ID:__________________ Subplot ID:_______________________ Quadrat ID:______________________ Habitat code (for quadrat):___________ Percent cover:_____________________ Collector's initials:_________________	Species ID in field:_________________ Hexagon plot ID:__________________ Subplot ID:_______________________ Quadrat ID:______________________ Habitat code (for quadrat):___________ Percent cover:_____________________ Collector's initials:_________________

INTERIOR SUBPLOT (radius = 7.3 m) DATA SHEETS:
DATE:_____OBS:_____PG:___of____ PROPERTY:_____
SUBPLOT LOCATION:_____

Litter depth: azimuth direction:_____

Distance along transect	Depth (cm)
2.5 m	
5.0 m	
7.5 m	

Canopy cover at 7.3 m from center of plot:

Direction	Cover (Y/N)
0^0	
90^0	
180^0	
270^0	

Quadrats are 4.6 m (15 ft) from plot center
Quadrat 1 Circle azimuth: 30^0, 150^0, or 270^0

Plant species	Percent cover

Quadrat 2 Azimuth: 30^0, 150^0, or 270^0

Plant species	Percent cover

Trampling codes: 1=0-10% of quadrat trampled; 2=11-50% of quadrat trampled; 3=51-100% of quadrat trampled.

Quadrat	Trampling

Visual obstruction: (Robel pole @ 4-m distance & 1-m height). Placed in 3rd quadrat location. Record Azimuth_____

Direction	Height (cm)
0^O	
90^O	
180^O	
270^O	

5 minute area search for plant species:

Plant species

PLANT COMPOSITION SUBPLOT (radius = 17.3 m) DATA SHEETS:
DATE:________OBS:__________________PG:___of____ LOCATION:________________
SUBPLOT LOCATION:____________________________TRANSECT AZIMTH:______

Ground cover% (% add to 100%):

Type	<1m	5 m	10 m	15 m
Litter				
Vegetation				
Rock				
Soil / sand				
Water				
Other (explain)				

Woody debris along all 17.6 m:
Azimuth direction:_____

Dist. From center (m)	Small end diameter (cm)	Large end diameter (cm)	Length (m)	Decay class

Trees species & snags along 17.6 m (> 12.5 cm DBH) check if alive

Species	Alive/ dead	DBH	Decay?

Notes on incidental sightings:

Date entered:______by:_____#_________
Date checked:________by______

Chapter Twenty-one
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 AND 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, or dropping a depth gauge or sounding line from a boat in deeper water. In some waters, a SONAR depth finder may be necessary to determine the water depth. 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 m^2 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 needs to be known prior to data collection to allow 30 0.75 m^2 rectangular plots to be spaced

evenly. It may be best to decide on exact locations based on GIS coverage and use a surveyor's tape to ensure proper spacing of pre-selected locations in the field.

Substrate Measurements

Within each 0.75 m^2 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 (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). 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. In deep water, a heavyweight deep water bottom dredge (e.g. 6 x 6 inch samples) may be needed, although a lightweight shallow water bottom dredge may be better for shallower areas.

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 meters, 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 plot 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 (m^3/sec).

Water temperature

Water pH: The amount of acidity in the water.

Conductivity: The amount of ions (e.g. salts) dissolved in the water.

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 rivers, this information may need to be collected with sounding lines or SONAR.

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 for smaller channels 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.

EQUIPMENT LIST:

Appropriate sampling frames made from PVC pipes and ropes
Compass
Hip chain or surveyors tape
Meter stick
Notes and maps of GIS coverage
pH & conductivity meter
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 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 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. Technicians should also take proper precautions around water, i.e. avoiding fast, deep flowing water.

ADDITIONAL METHODS FOR SPECIAL LOCATIONS:
None.

ADDITIONAL READING:

Platts, Megehan, and Minshell. 1983. Methods for Evaluating Stream, Riparian, and Biotic Conditions. Gen. Tech. Rep. INT-138.

Simonsen, Lyons, and Kehehl. 1994. Guidelines for Evaluating Fish Habitat in Wisconsin Streams. Gen. Tech. Rep. NC-164.

AQUATIC MEASUREMENTS OF LOTIC (running water) HABITAT SAMPLING PLOTS.

DATE:_____OBS:_____PROPERTY:_____PG:___of___

Plot	1	2	3	4	5	6	7	8	9	10	11
Distance	0										
Channel type											
Wetted width											
Bankfull width											
Bankfull height											
Incised height											
Stream discharge											
Water temperature											
Water pH											
Water conductivity											
Riparian veg. width											
0 m from shore 0.25mx0.25m plots											
Depth											
% silt											
%sand											
% gravel											
% cobbles											
% boulders											
% bedrock											
% emergent veg.											
% submergt veg.											
_____ m from shore											
Depth											
% silt											
%sand											
% gravel											
% cobbles											
% boulders											
% bedrock											
% emergent veg.											
% submergt.veg.											
_____ m from shore											
Depth											
% silt											
% sand											
% gravel											
% cobbles											
% boulders											
% bedrock											
% emergent veg.											
% submergt. veg											
_____ m from shore											
Depth											
% silt											
% sand											
% gravel											
% cobbles											
% boulders											
% bedrock											
% emergent veg.											
% submergt veg											

Approximate starting pt (GPS coordinates from GIS): E_____ N_____

Actual downstream starting pt (acquired in field): E_____ N_____

Plot	1	2	3	4	5	6	7	8	9	10	11
Water depth											
Location 1											
Location 2											
Location 3											
Location 4											
Location 5											
Location 6											
Location 7											
Location 8											
Location 9											
Location 10											
Spacing between measures:											

Spacing b/t measures – divide wetted width by 10.

Backside of data sheet for:
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 (m^3/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.

Date entered:_____by:_____Rec#:_____checked by:_____date:_____

AQUATIC MEASUREMENTS OF LOTIC (running water) HABITAT SAMPLING PLOTS.
DATE:_____OBS:_____PROPERTY:_____PG:___of___

POOLS data:

Number	Maximum depth	Surface area	Cause

Number: May be obtained from GIS cover or in the field. Either check with numbered map or mark number and location on map in field for addition to the GIS database.
Cause: Why is the water pooling? Examples include log, rocks, soil, dam.

Additional notes on wildlife encountered today:

Woody debris:

Distance from downstream starting pt.	Small end diameter	Large end diameter	Length	Decay class

Date entered:_____by:_____
Record#:_____
Checked by:_____date:_____

AQUATIC MEASUREMENTS OF LENTIC (standing water) HABITAT SAMPLING PLOTS.
DATE:_____OBS:_____PROPERTY:_____PG:___of___
UTM coordinates (GPS acquired in field): E_____ N_____
Pond #/name:_____ LAB UTMS E:_____N:_____Perimeter:_____

Plot	Distance	Max. Depth	In edge depth	% silt	% sand	% gravel	% cobble	% boulders	% bedrock	% emerg. Veg.	% submer veg
1	0										
2											
3											
4											
5											
6											
7											
8											
9											
10											
11											
12											
13											
14											
15											
16											
17											
18											
29											
20											
21											
22											
23											
24											
25											
26											
27											
28											
29											
30											

Distance can be filled out in the lab based on GIS coverage and used to find sampling plot.
In edge depth=depth at the edge of the plot furthest into the water from shore.
% classifications: 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

Date entered:_____by:_____
Record#:_____
Date checked:_____by:_____

Literature Cited

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Anderson, T. (2004, September 16). Callused Vertigo (*Vertigo authuri*): a technical conservation assessment. [Online]. USDA Forest Service, Rocky Mountain Region. Available: http://www.fs.fed.us/r2/projects/scp/assessments/callusedvertigo.pdf [Last accessed December 6, 2006].

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APPENDIX 1

TABLES OF IOWA WILDLIFE

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

Checklist of Iowa mammals (Bowles 1975, Kane et al. 2003) continued.

Order	Family	*Scientific name*	Common name
Rodentia	Zapodidae	*Zapus hudsonius*	Meadow jumping mouse
	Capromyidae	*@Myocaster coypus*	Nutria
	Erethizontidae	*Erethizon dorsatum*	Porcupine
Carnivora	Canidae	*Canis latrans*	Coyote
		Canis lupus	**[E] Gray wolf
		Urocyon cinereoargenteus	Gray fox
		Vulpes vulpes	Red fox
	Ursidae	*Ursus americanus*	Black bear
	Procyonidae	*Procyon lotor*	Raccoon
	Mustelidae	*Gulo gulo*	**Wolverine
		Lutra canadensis	River otter
		Martes pennanti	Fisher
		Mephitis mephitis	Striped skunk
		Mustela erminea	Ermine
		Mustela frenata – 2 subspecies in Bowles: *M.f. primulina & m.f. spadix*	Long-tailed weasel
		Mustela nivalis	Least weasel
		Mustela vison	Mink
		Spilogale putorius	[A] Spotted Skunk
		Taxidea taxus	Badger
	Felidae	*Felis concolor*	**Mountain lion
		Lynx canadensis	**Lynx
		Lynx rufus	Bobcat
Artiodactyla	Cervidae	*Cervus elaphus canadensis*	**Wapiti
		Odocoileus hemionus	**Mule deer
		Odocoileus virginianus	White-tailed deer
	Antilocapridae	*Antilocapra americana*	**Pronghorn
	Bovidae	*Bison bison*	**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.
[A] Iowa endangered species.
[B] Iowa threatened species.
[C] Iowa species of special concern.
[E] Federally endangered species.

Checklist of 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	*Myotis septentrionalis*	**[D]Northern myotis
		Myotis lucifugus	Little brown bat
		Myotis sodalis	[AE]Indiana bat
		Lasionycteris noctivagans	Silver-haired bat
		Pipistrellus subflavus	Eastern pipistrelle
		Eptesicus fuscus	Big brown bat
		Lasiurus borealis	Red bat
		Lasiurus cinereus	Hoary bat
		Nycticeius humeralis	Evening bat
	Molossidae	*Tadarida macrotis*	*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).

[A] Iowa endangered species.

[B] Iowa threatened species.

[C] Iowa species of special concern.

[D] Federally threatened species.

[E] Federally endangered species.

Checklist of 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	*Necturus maculosus*	[B] Mudpuppy
	Salamandridae	*Notophthalmus viridescens*	[B] Central newt
	Ambystomatidae	*Ambystoma tigrinum*	Tiger salamander
		Ambystoma texanum	Smallmouth salamander
		Ambystoma laterale	[A] Blue-spotted salamander
Anura	Ranidae	*Rana pipiens*	Northern leopard frog
		Rana blairi	Plains leopard frog
		Rana utricularia	Southern leopard frog
		Rana palustris	Pickerel frog
		Rana areolata	[A] **Crawfish frog
		Rana clamitans	Green frog
		Rana catesbeiana	Bullfrog
		Rana sylvatica	*Wood frog
	Hylidae	*Hyla chrysoscelis*	Cope's gray treefrog
		Hyla versicolor	Gray treefrog
		Pseudacris crucifer	Spring peeper
		Pseudacris triseriata	Western chorus frog
		Acris crepitans	Northern cricket frog
	Bufonidae	*Bufo americanus*	American toad
		Bufo woodhousii woodhousii	Woodhouse's toad
		Bufo woodhousii fowleri	Fowler's toad
		Bufo cognatus	Great plains toad
	Pelobatidae	*Spea bombifrons*	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).

[A] Iowa endangered species.

[B] Iowa threatened species.

[C] Iowa species of special concern.

Checklist of 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	*Terrapene ornata*	[B] Ornate box turtle
		Chrysemys picta	Painted turtle
		Trachemys scripta	Red-eared turtle
		Graptemys geographica	Map turtle
		Graptemys pseudogeographica	False map turtle
		Emydoidea blandingi	[B] Blanding's turtle
		Clemmys inscuplta	[A] Wood turtle
	Chelydridae	*Chelydra serpentina*	[**] Snapping turtle
		Macroclemys temmincki	[**] Alligator snapping turtle
	Kinosternidae	*Kinosternon flavescens*	[A] Yellow mud turtle
		Sternotherus odoratus	[B] Stinkpot
	Trionychidae	*Apalone spinifera*	Spiny softshell turtle
		Apalone mutica	Smooth softshell turtle
Squamata	Scincidae	*Eumeces septentrionalis*	Northern prairie skink
		Eumeces fasciatus	Five-lined skink
		Eumeces obsoletus	[A] Great plains skink
	Anguidae	*Ophisaurus attenuatus*	[B] Slender glass lizard
	Teiidae	*Cnemidophorus sexlineatus*	Six-lined racerunner
	Colubridae	*Nerodia sipedon*	Northern water snake
		Nerodia rhombifera	[B] Diamondback water snake
		Nerodia erythrogaster	Yellowbelly water snake
		Nerodia erythrogaster neglecta	[A, D] Copperbelly water snake
		Regina grahami	Graham's crayfish snake
		Storeria dekayi	Brown snake
		Storeria occipitomaculata	Northern redbelly snake
		Virginia valeriae	Smooth earth snake
		Tropidoclonion lineatum	Northern lined snake
		Thamnophis sirtalis	Eastern garter snake
		Thamnophis radix	Plains garter snake
		Thamnophis proximus	Western ribbon snake
		Carphophis amoenus	[B] Western worm snake
		Opheodrys vernalis	[C] Smooth green snake
		Opheodrys aestivus	[**] Rough green snake
		Diadophis punctatus	Prairie ringneck snake
		Coluber constrictor	Racer
		Lampropeltis triangulum	Milk snake
		Lampropeltis calligaster	Prairie kingsnake

Checklist of Iowa reptiles (Christiansen and Bailey 1990 & 1998, Kane et al. 2003). These animals represent 31 genera, 9 families, and 2 orders (continued).

Order	Family	*Scientific name*	Common name
		Lampropeltis getulus	[B] Speckled kingsnake
		Pituophis melanoleucus	[C] Bullsnake
		Elaphe vulpina	Fox snake
		Elaphe obsoleta	Black rat snake
		Heterodon platyrhinos	Eastern hognose snake
		Heterodon nasicus	* * [A] Western hognose snake
	Viperidae	*Sistrurus catenatus*	[A] Massasauga
		Sistrurus catenatus catenatus	*Eastern massasauga
		Crotalus horridus	Timber rattlesnake
		Crotalus viridis	[A] Prairie rattlesnake
		Agkistrodon contortrix	[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.
[A] Iowa endangered species.
[B] Iowa threatened species.
[C] Iowa species of special concern.
[D] Not in GAP or Christiansen and Bailey (1998), but on the T&E list.

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

Checklist of Iowa breeding birds (Kent and Dinsmore 1996, Kane et al. 2003) continued.

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

Checklist of Iowa breeding birds (Kent and Dinsmore 1996, Kane et al. 2003) continued.

Order	Family	*Scientific name*	Common name
		Sterna antillarum	[AE] Least tern
		Chlidonias niger	[C] Black tern
Columbiformes	Columbidae	*Columa livia*	Rock pigeon
		Streptopelia decaocto	Eurasian collared dove
		Zenaida macroura	Mourning dove
		Ectopistes migratorius	Passenger pigeon (extinct)
Psittaciformes	Psittacidae	*Conuropsis carolinensis*	Carolina parakeet (extinct)
Cuculiformes	Cuculidae	*Coccyzus erythrophthalmus*	Black-billed cuckoo
		Coccyzus americanus	Yellow-billed cuckoo
Strigiformes	Tytonidae	*Tyto alba*	[A] Barn owl
	Strigidae	*Otus asio*	Eastern screech-owl
		Bubo virginianus	Great horned owl
		Speotyto cunicularia	Burrowing owl
		Strix varia	Barred owl
		Asio otus	[B] Long-eared owl
		Asio flammeus	[A] Short-eared owl
Caprimulgiformes	Caprimulgidae	*Chordeiles minor*	Common nighthawk
		Caprimulgus carolinensis	Chuck-will's-widow
		Caprimulgus vociferus	Whip-poor-will
Apodiformes	Apodidae	*Chaetura pelagica*	Chimney swift
	Trochilidae	*Archilochus colubris*	Ruby-throated hummingbird
Coraciiformes	Alcedinidae	*Ceryle alcyon*	Belted kingfisher
Piciformes	Picidae	*Melanerpes erythrocephalus*	Red-headed woodpecker
		Melanerpes carolinensis	Red-bellied woodpecker
		Sphyrapicus varius	Yellow-bellied sapsucker
		Picoides pubescens	Downy woodpecker
		Picoides villosus	Hairy woodpecker
		Colaptes auratus	Northern flicker
		Dryocopus pileatus	Pileated woodpecker
Passerformes	Tyrannidae	*Contopus virens*	Eastern wood-pewee
		Empidonax virescens	Acadian flycatcher
		Empidonax traillii	Willow flycatcher
		Empidonax minimus	Least flycatcher
		Sayornis phoebe	Eastern phoebe
		Sorornis saya	Say's phoebe
		Myiarchus crinitus	Great crested flycatcher

Checklist of Iowa breeding birds (Kent and Dinsmore 1996, Kane et al. 2003) continued.

Order	Family	*Scientific name*	Common name
		Tyrannus verticalis	Western kingbird
		Tyrannus tyrannus	Eastern kingbird
		Tyrannus fortificatus	Scissor-tailed flycatcher
	Alaudidae	*Eremophila alpesteris*	Horned lark
	Hirundinidae	*Progne subis*	Purple martin
		Tachycineta bicolor	Tree swallow
		Stelgidopteryx serripennis	Northern rough-winged swallow
		Riparia riparia	Bank swallow
		Hirundo pyrrhonota	Cliff swallow
		Hirundo rustica	Barn swallow
	Corvidae	*Cyanocitta cristata*	Blue jay
		Pica pica	Black-billed magpie
		Corvus brachyrhynchus	American crow
	Paridae	*Parus atricapillus*	Black-capped chickadee
		Parus bicolor	Tufted titmouse
	Sittidae	*Sitta canadensis*	Red-breasted nuthatch
		Sitta carolinensis	White-breasted nuthatch
	Certhiidae	*Certhia americana*	Brown creeper
	Troglodytidae	*Salpinctes obsoletus*	Rock wren
		Thryothorus ludovicianus	Carolina wren
		Thryomanes bewickii	Bewick's wren
		Troglodytes aedon	House wren
		Troglodytes troglodytes	Winter wren
		Cistothorus platensis	Sedge wren
		Cistothorus palustris	Marsh wren
	Muscicapidae	*Polioptila caerulea*	Blue-gray gnatcatcher
		Sialia sialis	Eastern bluebird
		Catharus fuscescens	Veery
		Hylocichla mustelina	Wood thrush
		Turdus migratorius	American robin
	Mimidae	*Dumetella carolinensis*	Gray catbird
		Mimus polyglottos	Northern mockingbird
		Toxostoma rufum	Brown thrasher
	Bombycilla	*Bombycilla cedrorum*	Cedar waxwing
	Laniidae	*Lanius ludovicianus*	Loggerhead shrike
	Sturnidae	*Sturnus vulgaris*	European starling
	Vireonidae	*Vireo flavifrons*	Yellow-throated vireo
		Vireo gilvus	Warbling vireo
		Vireo olivaceus	Red-eyed vireo

Checklist of Iowa breeding birds (Kent and Dinsmore 1996, Kane et al. 2003) continued.

Order	Family	*Scientific name*	Common name
		Vireo griseus	White-eyed vireo
		Vireo bellii	Bell's vireo
	Emberizidae	*Vermivora pinus*	Blue-winged warbler
		Vermivora chrysoptera	Golden-winged warbler
		Parula americana	Northern parula
		Dendroica petechia	Yellow warbler
		Dendroica pensylvanica	Chestnut-sided warbler
		Dendroica dominica	Yellow-throated warbler
		Dendroica discolor	Prairie warbler
		Dendroica cerulea	Cerulean warbler
		Mniotilta varia	Black-and-white warbler
		Setophaga ruticilla	American redstart
		Protonotaria citrea	Prothonotary warbler
		Helmitheros vermivorus	Worm-eating warbler
		Seiurus aurocapillus	Ovenbird
		Seiurus motacilla	Louisiana waterthrush
		Oporornis formosus	Kentucky warbler
		Geothlypis trichas	Common yellowthroat
		Wilsonia citrina	Hooded warbler
		Icteria virens	Yellow-breasted chat
		Piranga rubra	Summer tanager
		Piranga olivacea	Scarlet tanager
		Cardinalis cardinalis	Northern cardinal
		Pheucticus ludovicianus	Rose-breasted grosbeak
		Guiraca caerulea	Blue grosbeak
		Passerina cyanea	Indigo bunting
		Spiza americana	Dickcissel
		Pipilo erythrophthalmus	Eastern towhee
		Spizella passerina	Chipping sparrow
		Spizella pallida	Clay-colored sparrow
		Spizella pusilla	Field sparrow
		Pooecetes gramineus	Vesper sparrow
		Chondestes grammacus	Lark sparrow
		Passerculus sandwichensis	Savannah sparrow
		Ammodramus savannarum	Grasshopper sparrow
		Ammodramus henslowii	[B] Henslow's sparrow
		Melospiza melodia	Song sparrow
		Melospiza georgiana	Swamp sparrow
		Dolichonyx oryzivorus	Bobolink

Checklist of Iowa breeding birds (Kent and Dinsmore 1996, Kane et al. 2003) continued.

Order	Family	*Scientific name*	Common name
		Agelaius phoeniceus	Red-winged blackbird
		Sturnella magna	Eastern meadowlark
		Sturnella neglecta	Western meadowlark
		Xanthocephalus xanthocephalus	Yellow-headed blackbird
		Quiscalus mexicanus	Great-tailed grackle
		Quiscalus quiscala	Common grackle
		Molothrus ater	Brown-headed cowbird
		Icterus spurius	Orchard oriole
		Icterus galbula	Baltimore oriole
	Fringillidae	*Carpodacus mexicanus*	House finch
		Loxia curvirostra	Red crossbill
		Carduelis pinus	Pine siskin
		Carduelis tristis	American goldfinch
	Passeridae	*Passer domesticus*	House sparrow
		Passer montanus	Eurasian tree sparrow

* Indicates species in Kent and Dinsmore (1996), but not in GAP (Kane et al. 2003).
[A] Iowa endangered species.
[B] Iowa threatened species.
[C] Iowa species of special concern.
[E] Federally endangered species.
[T] Federally threatened species.

Checklist of 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	*Gavia stellata*	Red-throated loon
		Gavia pacifica	Pacific loon
		Gavia adamsii	*Yellow-billed loon
Podicipediformes	Podicipedidae	*Podiceps auritus*	Horned grebe
		Aechmophorus clarkii	Clark's grebe
Pelicaniformes	Pelicanidae	*Pelcanus occidentalis*	Brown pelican
	Phalacrocoracidae	*Phalacrocorax brasilianus*	Neotropic cormorant
	Anhingidae	*Ahinga anhinga*	Anhinga
	Frigatidae	*Fregata magnifiscens*	Magnificent frigatebird
Ciconiiformes	Ardeidae	*Egretta thula*	Snowy egret
		Egretta tricolor	Tricolored heron
		Egretta rufescens	Reddish egret
	Threskiornithidae	*Eudocimus albus*	White ibis
		Plegadis falcinellus	Glossy ibis
		Ajaia ajaja	Roseate spoonbill
	Ciconiidae	*Mycteria americana*	Wood stork
Anseriformes	Anatidae	*Dendrocygna autumnalis*	Black-bellied whistling duck
		Cygnus columbianus	Tundra swan
		Anser fabalis	Bean goose
		Anser albifrons	Greater white-fronted goose
		Chen caerulescens	Snow goose
		Chen rossii	Ross's goose
		Branta bernicla	Brant
		Anas querquedula	Garganey
		Anas cyanoptera	Cinnamon teal
		Anas penelope	Eurasian wigeon
		Aythya marila	Greater scaup
		Somateria mollissima	Common eider
		Somateria spectabilis	King eider
		Histronicus histronicus	Harlequin duck
		Clangula hyemalis	Long-tailed duck
		Melanitta nigra	Black scoter
		Melanitta perspicillata	Surf scoter
		Melanitta fusca	White-winged scoter
		Bucephala clangula	Common goldeneye
		Bucephala islandica	Barrow's goldeneye
		Mergus merganser	Common merganser
		Mergus serrator	Red-breasted merganser
Falconiformes	Cathartidae	*Coragyps atratus*	Black vulture

Checklist of Iowa's migratory birds and vagrants (Kent and Dinsmore 1996, Kane et al. 2003). These animals represent 116 genera, 36 families, and 12 orders continued.

Order	Family	*Scientific name*	Common name
	Accipitridae	*Accipiter gentilis*	Northern goshawk
		Buteo regalis	Ferruginous hawk
		Buteo lagopus	Rough-legged hawk
		Aquila chrysaetos	Golden eagle
	Falconidae	*Falco rusticolus*	Gyrfalcon
		Falco mexicanus	Prairie falcon
Gruiformes	Rallidae	*Coturnicops noveboracensis*	Yellow rail
		Laterallus jamaicensis	Black rail
		Porphyrula martinica	Purple gallinule
Charadriiformes	Charadriidae	*Pluvialis squatarola*	Black-bellied plover
		Pluvialis dominicus	American golden-plover
		Charadrius alexandrinus	Snowy plover
		Charadrius semipalmatus	Semipalmated plover
	Recurvirostridae	*Himantopus mexicanus*	Black-necked stilt
		Recurvirostra americana	American avocet
	Scolopacidae	*Tringa melanoleuca*	Greater yellowlegs
		Tringa flavipes	Lesser yellowlegs
		Tringa solitaria	Solitary sandpiper
		Catoptrophorus semiplamatus	Willet
		Numenius borealis	[E] Eskimo curlew
		Numenius phaeopus	Whimbrel
		Limosa haemastica	Hudsonian godwit
		Arenaria interpres	Ruddy turnstone
		Calidris canutus	Red knot
		Calidris alba	Sanderling
		Calidris pusilla	Semipalmated sandpiper
		Calidris mauri	Western sandpiper
		Calidris minutilla	Least sandpiper
		Calidris fuscicollis	White-rumped sandpiper
		Calidris bairdii	Baird's sandpiper
		Calidris melanotos	Pectoral sandpiper
		Calidris acuminata	Sharp-tailed sandpiper
		Calidris alpina	Dunlin
		Calidris ferruginea	Curlew sandpiper
		Calidris himantopus	Stilt sandpiper
		Tryngites subruficollis	Buff-breasted sandpiper

Checklist of Iowa's migratory birds and vagrants (Kent and Dinsmore 1996, Kane et al. 2003). These animals represent 116 genera, 36 families, and 12 orders continued.

Order	Family	*Scientific name*	Common name
		Philomachus pugnax	Ruff
		Limnodromus griseus	Short-billed dowitcher
		Limnodromus scolopaceus	Long-billed dowitcher
		Phalaropus lobatus	Red-necked phalarope
		Phalaropes fulicaria	Red phalarope
	Laridae	*Stercorarius pomarinus*	Pomarine jaeger
		Stercorarius parasiticus	Parasitic jaeger
		Stercorarius longicaudus	Long-tailed jaeger
		Larus atricilla	Laughing gull
		Larus minutus	Little gull
		Larus ridibundus	Black-headed gull
		Larus philadelphia	Bonaparte's gull
		Larus canus	Mew gull
		Larus californicus	California gull
		Larus argentatus	Herring gull
		Larus thayeri	Thayer's gull
		Larus glaucoides	Iceland gull
		Larus fuscus	Lesser black-backed gull
		Larus schistisagus	Slaty-backed gull
		Larus hyperboreus	Glaucous gull
		Larus marinus	Great black-backed gull
		Rissa tridactyla	Black-legged kittiwake
		Rhodostethia rosea	Ross's gull
		Xema sabini	Sabine's gull
		Pagophila eburnea	Ivory gull
		Sterna caspia	Caspian tern
		Sterna hirundo	Common tern
		Sterna pardisaea	*Artic tern
	Alcidae	*Uria lomvia*	Thick-billed murre
		Brachyramphus marmoratus or B.m. perdix	Long-billed murrelet
		Synthliboramphus antiquus	Ancient murrelet
	Columbidae	*Zenaida asiatica*	*White-winged dove
		Columbina passerina	Common ground-dove
Cuculiformes	Cuculidae	*Crotophaga sulcirostris*	Groove-billed ani
Strigiformes	Strigidae	*Nyctea scandiaca*	Snowy owl

Checklist of Iowa's migratory birds and vagrants (Kent and Dinsmore 1996, Kane et al. 2003). These animals represent 116 genera, 36 families, and 12 orders continued.

Order	Family	*Scientific name*	Common name
		Surnia ulula	Northern hawk owl
		Strix nebulosa	Great gray owl
		Aegolius acadicus	Northern saw-whet owl
	Trochilidae	*Selasphorus rufus*	Rufous hummingbird
Piciformes	Picidae	*Melanerpes lewis*	Lewis's woodpecker
		Picoides arcticus	Black-backed woodpecker
Passerformes	Tyrannidae	*Contopus borealis*	Olive-sided flycatcher
		Contopus sordidulus	Western wood-pewee
		Empidonax flaviventris	Yellow-bellied flycatcher
		Empidonax alnorum	Alder flycatcher
		Empidonax species	"Western" flycatcher
		Pyrocephalus rubinus	Vermillion flycatcher
	Corvidae	*Perisoreus canadensis*	Gray jay
		Gymnorhinus cyanocephalus	Pinyon jay
		Nucifraga columbiana	Clark's nutcracker
		Corvus ossifragus	Fish crow
		Corvus corax	Common raven
	Paridae	*Parus hudsonicus*	Boreal chickadee
	Sittidae	*Sitta pygamaea*	Pygmy nuthatch
	Muscicapidae	*Regulus satrapa*	Golden-crowned kinglet
		Regulus calendula	Ruby-crowned kinglet
		Sialia currucoides	Mountain bluebird
		Myadestes townsendi	Townsend's solitaire
		Catharus ustulatus	Gray-cheeked thrush
		Catharus ustulatus	Swainson's thrush
		Catharus guttatus	Hermit thrush
		Ixoreus naevius	Varied thrush
	Mimidae	*Oreoscoptes montanus*	Sage thrasher
		Toxostoma curvirostre	Curve-billed thrasher
	Motacillidae	*Anthus rubescens*	American pipit
		Anthus spragueii	*Sprague's pipit
	Bombycillidae	*Bombycilla garrulus*	Bohemian waxwing
	Laniidae	*Lanius excubitor*	Northern shrike
	Sturnidae	*Vireo solitarius*	Blue-headed vireo
		Vireo philadelphicus	Philadelphia vireo
	Emberizidae	*Vermivora peregrina*	Tennessee warbler
		Vermivora celata	Orange-crowned warbler

Checklist of Iowa's migratory birds and vagrants (Kent and Dinsmore 1996, Kane et al. 2003). These animals represent 116 genera, 36 families, and 12 orders continued.

Order	Family	*Scientific name*	Common name
		Vermivora ruficapilla	Nashville warbler
		Dendroica magnolia	Magnolia warbler
		Dendroica tigrina	Cape May warbler
		Dendroica caerulescens	Black-throated blue warbler
		Dendroica coronata	Yellow-rumped warbler
		Dendroica nigrescens	Black-throated gray warbler
		Dendroica townsendii	Townsend's warbler
		Dendroica virens	Black-throated green warbler
		Dendroica fusca	Blackburnian warbler
		Dendroica pinus	Pine warbler
		Dendroica palmarum	Palm warbler
		Dendroica castanea	Bay-breasted warbler
		Dendroica striata	Blackpoll warbler
		Seiurus noveboracensis	Northern waterthrush
		Oporornis agilis	Connecticut warbler
		Oporornis philadelphia	Mourning warbler
		Oporornis tolmiei	MacGillivray's warbler
		Wilsonia pusilla	Wilson's warbler
		Wilsonia canadensis	Canada warbler
		Piranga ludoviciana	Western tanager
		Pheucticus melanocephalus	Black-headed grosbeak
		Passerina amoena	Lazuli bunting
		Passerina ciris	Painted bunting
		Pipilo chlorurus	Green-tailed towhee
		Pipilo maculatus	Spotted towhee
		Spizella arborea	American tree sparrow
		Amphispiza bilineata	Black-throated sparrow
		Calamospiza melanocorys	Lark bunting
		Ammodramus leconteii	Le Conte's sparrow
		Ammodramus nelsoni	Nelson's sharp-tailed sparrow
		Passerella iliaca	Fox sparrow
		Melospiza lincolnii	Lincoln's sparrow
		Zonotrichia albicollis	White-throated sparrow
		Zonotrichia atricapilla	Golden-crowned sparrow
		Zonotrichia leucophrys	White-crowned sparrow
		Zonotrichia querula	Harris's sparrow
		Junco hyemalis	Dark-eyed junco

Checklist of Iowa's migratory birds and vagrants (Kent and Dinsmore 1996, Kane et al. 2003). These animals represent 116 genera, 36 families, and 12 orders continued.

Order	Family	*Scientific name*	Common name
		Calcarius lapponicus	Lapland longspur
		Calcarius pictus	Smith's longspur
		Calcarius ornatus	Chestnut-collared longspur
		Plectrophenax nivalis	Snow bunting
		Euphagus carolinus	Rusty blackbird
		Euphagus cyanocephalus	Brewer's blackbird
		Icterus bullockii	Bullock's oriole
	Fringillidae	*Leucosticte tephrocotis*	Gray-crowned rosy finch
		Pinicola enucleator	Pine grosbeak
		Carpodacus purpureus	Purple finch
		Loxia leucoptera	White-winged crossbill
		Carduelis flammea	Common redpoll
		Carduelis hornemanni	Hoary redpoll
		Coccothraustes vespertinus	Evening grosbeak
		Carduelis psaltria	*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).

[A] Iowa endangered species.

[B] Iowa threatened species.

[C] Iowa species of special concern.

[E] Federally endangered species.

Checklist of 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	*Achalarus lyciades*	Hoary edge
		Amblyscirtes hegon	[C] Pepper and salt skipper
		Amblyscirtes vialis	Common roadside skipper
		Ancyloxypha numitor	Least skipper
		Atalopedes campestris	Sachem
		Atrytone arogos	[C] Arogos skipper
		Atrytone logan	Delaware skipper
		Atrytonopsis hianna	[C] Dusted skipper
		Epargyreus clarus	Silver-spotted skipper
		Erynnis baptisiae	[C] Wild indigo duskywing
		Erynnis brizo	[C] Sleepy duskywing
		Erynnis horatius	Horace's duskywing
		Erynnis icelus	[C] Dreamy duskywing
		Erynnis juvenalis	Juvenal's duskywing
		Erynnis lucilius	[C] Columbine duskywing
		Erynnis martialis	Mottled duskywing
		Erynnis persius	Persius duskywing
		Euphyes bimacula	[C] Two-spotted skipper
		Euphyes conspicua	Black dash
		Euphyes dion	[C] Sedge (or dion) skipper
		Euphyes vestris	Dun skipper
		Hesperia dacotae	[AE] Dakota skipper
		Hesperia leonardus	[C] Leonard's skipper
		Hesperia ottoe	[C] Ottoe skipper
		Hesperia uncas	Uncas skipper
		Hylephila phyleus	Fiery skipper
		Lerodea eufala	Eufala skipper
		Oarisma powesheik	[B] Powesheik skipperling
		Pholisora catullus	Common sooty wing
		Poanes hobomok	Hobomok skipper
		Poanes massasoit	[B] Mulberry wing
		Poanes viator	[C] Broad-winged skipper
		Poanes zabulon	[C] Zabulon skipper
		Polites mystic	Long dash
		Polites origenes	Crossline skipper
		Polites peckius	Peck's skipper
		Polites themistocles	Tawny-edged skipper
		Pompeius verna	Little glassywing
		Problema byssus	[B] Byssus skipper

Checklist of Iowa butterflies (Opler and Krezik 1984, Zohrer et al. 2005). These animals represent 70 genera, 6 families, and 1 order continued.

Order	Family	*Scientific name*	Common name
		Pyrgus communis	Common checkered skipper
		Staphylus hayhurstii	Southern sooty wing
		Thorybes bathyllus	Southern cloudwing
		Thorybes pylades	Northern cloudwing
		Thymelicus lineola	European skipper
		Wallengrenia egerement	Northern broken-dash
	Libytheidae	*Libetheana carinenta*	American snout
	Lycaenidae	*Calephelis muticum*	[C] Swamp metalmark
		Callophrys gryneus	Juniper hairstreak
		Callpohrys henrici	Henry's elfin
		Celastrina neglecta	Summer azure
		Everes comyntas	Eastern-tailed blue
		Feniseca tarquinius	Harvester
		Glaucopsyche lygdamus	[B] Silvery blue
		Hemiargus isola	Reakirt's blue
		Leptotes marina	Marine blue
		Lycaeides melissa	Melissa blue
		Lycaena dione	Gray copper
		Lycaena helloides	[C] Purplish copper
		Lycaena hyllus	Bronze copper
		Lycaena phlaeas	American copper
		Parrhasius m-album	White M hairstreak
		Plebejus saepiolus	Greenish blue
		Satyrium acadica	[C] Acadian hairstreak
		Satyrium calanus	Banded hairstreak
		Satyrium caryaevorum	[C] Hickory hairstreak
		Satyrium edwardsii	[C] Edward's hairstreak
		Satyrium liparops	[C] Striped hairstreak
		Satyrium titus	Coral hairstreak
		Strymon melinus	Gray hairstreak
	Nymphalidae	*Agraulis vanillae*	Gulf fritillary
		Anaea andria	Goatweed leafwing
		Asterocampa celtis	Hackberry emperor
		Asterocampa clyton	Tawny emperor
		Boloria bellona	Meadow fritillary
		Boloria selene	Silver-bordered fritillary
		Cercyonis pegala	Common wood nymph
		Chlosyne gorgone	Gorgone checkerspot

Checklist of Iowa butterflies (Opler and Krezik 1984, Zohrer et al. 2005). These animals represent 70 genera, 6 families, and 1 order continued.

Order	Family	*Scientific name*	Common name
		Chlosyne nycteis	Silvery checkerspot
		Coenoympha tullia	[A] Common ringlet
		Danaus plexippus	Monarch or Queen
		Enodia anthedon	Northern pearly eye
		Euphydryas phaeton	[B] Baltimore checkerspot
		Euptoieta claudia	Variegated fritillary
		Junonia coenia	Common buckeye
		Limenitis archippus	Viceroy
		Limenitis arthemis	Red-spotted purple
		Megisto cymela	Little wood satyr
		Nymphalis antiopa	Mourning cloak
		Nymphalis milberti	Milbert's tortoiseshell
		Nymphalis vaualbum	Compton tortoiseshell
		Phyciodes tharos	Pearl crescent
		Phycoides batesii	Tawny crescent
		Polygonia comma	Eastern comma
		Polygonia interrogationis	Question mark
		Polygonia progne	Gray comma
		Satyrodes eurydice	Eyed brown
		Speyeria aphrodite	Aphrodite fritillary
		Speyeria cybele	Great spangled fritillary
		Speyeria idalia	[C] Regal flitillary
		Vanessa atalanta	Red admiral
		Vanessa cardui	Painted lady
		Vanessa virginiensis	American lady
	Papilionidae	*Battus philenor*	[C] Pipevine swallowtail
		Eurytides marcellus	[C] Zebra swallowtail
		Papilio cresphontes	Giant swallowtail
		Papilio glaucus	Eastern tiger swallowtail
		Papilio polyxenes	Black swallowtail
		Papilio troilus	Spicebush swallowtail
	Pieridae	*Colias eurytheme*	Orange sulphur
		Colias philodice	Clouded sulphur
		Euchloe olympia	[C] Olympia white (or marble)
		Eurema lisa	Little yellow
		Eurema nicippe	Sleepy orange
		Nathalis iole	Dainty orange
		Phoebis sennae	Cloudless sulphur

Checklist of Iowa butterflies (Opler and Krezik 1984, Zohrer et al. 2005). These animals represent 70 genera, 6 families, and 1 order continued.

Order	Family	*Scientific name*	Common name
		Pieris rapae	Cabbage white
		Pontia protodice	Checkered white
		Zerene cesonia	Southern dogface

[A] Iowa endangered species
[B] Iowa threatened species
[C] Iowa species of special concern.
[E]Federally endangered

Checklist of 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	*Aeshna canadensis*	Canada darner
Suborder		*Aeshna constricta*	Lance-tipped darner
Anisoptera		*Aeshna interrupta*	Variable darner
		Aeshna multicolor	Blue-eyed darner
		Aeshna mutata	Spatterdock darner
		Aeshna tuberculifera	Black-tipped darner
		Aeshna umbrosa	Shadow darner
		Aeshna verticalis	Green striped darner
		Aeshna verticalis	Green-striped darner
		Anax junius	Common green darner
		Boyeria vinosa	Fawn darner
		Epiaeschna heros	Swamp darner
		Nasiaeschna pentacantha	Cyrano darner
	Corduliidae	*Epitheca cynosura*	Common baskettail
		Epitheca princeps	Prince baskettail
		Neurocordulia molesta	Smoky shadowdragon
		Neurocordulia yamaskanensis	Stygian shadowdragon
		Somatochlora ensigera	Plains emerald
		Somatochlora linearis	Mocha emerald
	Gomphidae	*Arigomphus cornutus*	Horned clubtail
		Arigomphus submedianus	Jade clubtail
		Dromogomphus spoliatus	Flag-tailed spinyleg
		Gomphus externus	Plains clubtail
		Gomphus fraternus	Midland clubtail
		Gomphus graslinellus	Pronghorn clubtail
		Gomphus militaris	Sulpher-tipped clubtail
		Gomphus quadricolor	Rapids clubtail
		Gomphus vastus	Cobra clubtail
		Ophiogomphus rupinsulensis	Rusty snaketail
		Ophiogomphus smithi	Sioux snaketail
		Progomphus obsurus	Common sanddragon
		Stylurus amnicola	Riverine clubtail
		Stylurus intricatus	Brimstone clubtail
		Stylurus notatus	Elusive clubtail
		Stylurus plagiatus	Russet-tipped clubtail
		Stylurus spiniceps	Arrow clubtail
	Libellulidae	*Celithemis elisa*	Calico pennant
		Celithemis eponina	Halloween pennant

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

Order	Family	Scientific name	Common name
		Erythemis simplicicollis	Eastern pondhawk
		Leucorrhinia intacta	Dot-tailed whiteface
		Libellula cyanea	Spangled skimmer
		Libellula incesta	Slaty skimmer
		Libellula luctuosa	Widow skimmer
		Plathemis lydia	Common whitetail
		Libellula pulchella	Twelve-spotted skimmer
		Libellula quadrimaculata	Four-spotted skimmer
		Pachydiplax longipennis	Blue dasher
		Pantela flavescens	Wandering glider
		Pantela hymenaea	Spot-winged glider
		Perithemis tenera	Eastern amberwing
		Sympetrum ambiguum	Blue-faced meadowhawk
		Sympetrum corruptum	Variegated meadowhawk
		Sympetrum costiferum	Saffron-winged meadowhawk
		Sympetrum danae	Black meadowhawk
		Sympetrum internum	Cherry-faced meadowhawk
		Sympetrum obtrusum	White-faced meadowhawk
		Sympetrum occidentale	Western meadowhawk
		Sympetrum rubicundulum	Ruby meadowhawk
		Sympetrum vicinum	Yellow-legged meadowhawk
		Tramea carolina	Carolina saddlebags
		Tramea lacerate	Black saddlebags
		Tramea onusta	Red saddlebags
	Macromiidae	*Macromia illinoiensis*	Illinois river cruiser
		Macromia taeniolata	Royal river cruiser
Odonata	Calopterygidae	*Calopteryx aequabilis*	River jewelwing
Suborder		*Calopteryx maculata*	Ebony jewelwing
Zygoptera		*Hetaerina titia*	Smoky rubyspot
		Hetaeruba americana	American rubyspot
	Coenagrionidae	*Amphiogrion saucium*	Eastern red damsel
		Argia alberta	Paiute dancer
		Argia apicalis	Blue-fronted dancer
		Argia emma	Emma's dancer
		Argia fumipennis	Variable dancer
		Argia moesta	Powdered dancer
		Argia plana	Springwater dancer

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

Order	Family	Scientific name	Common name
		Argia tibialis	Blue-tipped dancer
		Coenagrion angulatum	Prairie bluet
		Coenagrion resolutum	Taiga bluet
		Enallagma anna	River bluet
		Enallagma antennatum	Rainbow bluet
		Enallagma aspersum	Azure bluet
		Enallagma basidens	Double-striped bluet
		Enallagma boreale	Boreal bluet
		Enallagma caruncularum	Tulle bluet
		Enallagma civile	Familiar bluet
		Enallagma clausum	Alkali bluet
		Enallagma cyathigerium	Northern bluet
		Enallagma erbium	Marsh bluet
		Enallagma exsulans	Stream bluet
		Enallagma geminatum	Skimming bluet
		Enallagma hageni	Hagen's bluet
		Enallagma signatum	Orange bluet
		Enallagma traviatum	Slender bluet
		Enallagma vesperum	Vesper bluet
		Ischnura hastata	Citrine forktail
		Ischnura posita	Fragile forktail
		Ischnura verticalis	Eastern forktail
		Nehalennia Irene	Sedge sprite
	Lestidae	*Archilestes grandis*	Great spreadwing
		Lestes congener	Spotted spreadwing
		Lestes disjunctus	Common spreadwing
		Lestes dryas	Emerald spreadwing
		Lestes eurinus	Amber-winged spreadwing
		Lestes inaequalis	Elegant spreadwing
		Lestes forcipatus	Sweetflag spreadwing
		Lestes rectangularis	Slender spreadwing
		Lestes unguiculatus	Lyre-tipped spreadwing

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

Checklist of 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	*Ichthyomyzon castaneus*	[B] Chestnut lamprey
		Ichthyomyzon fossor	Northern brook lamprey
		Ichthyomyzon unicuspis	Silver lamprey
		Lampetra appendix	[B] American brook lamprey
Acipenseriformes	Acipenseridae	*Acipenser fulvescens*	[A] Lake sturgeon
		Scaphirhynchus albus	[AE] Pallid sturgeon
		Scaphirhynchus platorynchus	Shovelnose sturgeon
	Polyodontidae	*Polydon spathula*	Paddlefish
Amiiformes	Amiidae	*Amia calva*	Bowfin
Lepisosteiformes	Lepisosteidae	*Lepirosteus platostomus*	Shortnose gar
		Lepisosteus osseus	Longnose gar
		Lepisosteus oculatus	Spotted gar
Anguilliformes	Anguillidae	*Anguilla rostrata*	American eel
Clupeiformes	Clupeidae	*Dorosoma cepedianum*	Gizzard shad
		Alosa alabamae	Alabama shad
		Alosa chrysochloris	Skipjack herring
Osteoglossiformes	Hiodontidae	*Hiodon tergisus*	Mooneye
		Hiodon alosoides	Goldeye
Salmoniformes	Salmonidae	*Oncorhynchus mykiss*	Rainbow trout
		Salmo trutta	Brown trout
		Salvelinus fontinalis	Brook trout
	Esocidae	*Esox lucius*	Northern pike
		Esox masquinongy	Muskellunge
		Esox americanus	[B] Grass pickerel
	Umbridae	*Umbra limi*	Central mudminnow
Cypriniformes	Cyprinidae	*Campostoma anomalum*	Central stoneroller
		Campostoma oligolepsis	Largescale stoneroller
		Carassius auratus	Goldfish
		Ctenopharyngodon idella	Grass carp
		Cyprinus carpio	Common carp
		Hypophthalmichthys nobilis	Bighead carp
		Hypophthalmichthys molitrix	Silver carp
		Phenacobius mirabelis	Suckermouth minnow

Checklist of Iowa fish (From Zohrer et al. 2005). These animals represent 71 genera, 26 families, and 16 orders continued.

Order	Family	*Scientific name*	Common name
		Pimephales notatus	Bluntnose minnow
		Pimephales promelas	Fathead minnow
		Pimephales vigilax	Bullhead minor
		Hybognathus agryritis	Western silvery minnow
		Hybognathus nuchalis	Mississippi silvery minnow
		Hybognathus hankinsoni	Brassy minnow
		Hybognathus placitus	Plains minnow
		Semotilus atromaculatus	Creek chub
		Couesius plumbeus	Lake chub
		Macrhybopsis aestivalis	Speckled chub
		Macrhybopsis gelida	Sturgeon chub
		Platygobio gracillis	Flathead chub
		Macrybopsis meeki	Sicklefin chub
		Macrybopsis storeriana	Silver chub
		Erimytax x-punctatus	Gravel chub
		Nocomis biguttatus	Horneyhead chub
		Notemigonus crysoleucas	Golden shiner
		**Hybopsis amnis*	*Pallid shiner
		Opsopoeodus emiliae	[C] Pugnose minnow
		Notropis anogenus	[A] Pugnose shiner
		Notropis atherinoides	Emerald shiner
		Notropis blennius	River shiner
		Notropis buchanani	Ghost shiner
		Notropis chalybaeus	Ironcolor shiner
		Luxilus cornutus	Common shiner
		Notropis dorsalis	Bigmouth shiner
		Notropis heterodon	Blackchin shiner
		Notropis heterolepis	[B] Blacknose shiner
		Notropis hudsonius	Spottail shiner
		Cyprinella lutrensis	Red shiner
		Notropis nubilus	Ozark minnow
		Notropis rubellus	Rosyface shiner
		Notropis shumardi	Silverband shiner
		Cyprinella spiloptera	Spotfin shiner
		Notropis texanus	[A] Weed shiner
		Notropis Topeka	[BE] Topeka shiner
		Lythrurus umbratilis	Redfin shiner

Checklist of Iowa fish (From Zohrer et al. 2005). These animals represent 71 genera, 26 families, and 16 orders continued.

Order	Family	*Scientific name*	Common name
		Notropis volucellus	Mimic shiner
		Notropis wickliffi	Channel shiner
		Rhinichthys atratulus	Blacknose dace
		Rhinichthys cataractae	Longnose dace
		Margariscus margarita	[A] Pearl dace
		Phoxinus erythrogaster	Southern redbelly dace
		Clinostomus elongatus	Redside dace
	Catostomidae	*Cycleptus elongates*	Blue sucker
		Erimyzon sucetta	Lake chubsucker
		Ictiobus cyprinellus	Bigmouth buffalo
		Ictiobus bubalus	Smallmouth buffalo
		Ictiobus niger	Black buffalo
		Carpiodes cyprinus	Quillback
		Carpiodes carpio	River carpsucker
		Carpiodes velifer	Highfin carpsucker
		Moxostoma duquesnei	[B] Black redhorse
		Moxostoma erythrurum	Golden redhorse
		Moxostoma anisurum	Silver redhorse
		Moxostoma macrolepidotum	Shorthead redhorse
		Moxostoma carinatum	River redhorse
		Moxostoma valenciennesi	Greater redhorse
		Minytrema melanops	Spotted sucker
		Hypentelium nigricans	Northern hog sucker
		Catostomus commersoni	White sucker
Siluriformes	Ictaluridae	*Ictalurus punctatus*	Channel catfish
		Pylodictus olivaris	Flathead catfish
		Ictalurus furcatus	Blue catfish
		Ameiurus melas	Black bullhead
		Ameiurus natalis	Yellow bullhead
		Ameiurus nebulosus	Brown bullhead
		Noturus exilis	Slender madtom
		Noturus gyrinus	Tadpole madtom
		Noturus flavus	Stonecat
		Noturus gyrinus	[A] Freckled madtom
Percopsiformes	Aphredoderidae	*Aphredoderus sayanus*	[C] Pirate perch
	Percopsidae	*Percopsis omiscomycus*	Trout perch
Gadiformes	Gadidae	*Lota lota*	[B] Burbot

Checklist of Iowa fish (From Zohrer et al. 2005). These animals represent 71 genera, 26 families, and 16 orders continued.

Order	Family	*Scientific name*	Common name
Atheriniformes	Cyprinodontidae	*Fundulus diaphanous*	Banded killifish
		Fundulus notatus	Blackstripe topminnow
		Fundulus dispar	Starhead topminnow
		Fundulus sciadicus	Plains topminnow
	Poeciliidae	*Gambusia affins*	Western mosquito fish
	Atherinidae	*Labidesthes sicculus*	Brook silverside
Gasterosteiformes	Gasterosteidae	*Culaea inconstans*	Brook stickleback
Scorpaeniformes	Cottidae	*Cottus bairdi*	Mottled sulpin
		Cottus cognatus	Slimy sculpin
Perciformes	Percichthyidae	*Morone chysops*	White bass
		Morone mississippiensis	Yellow bass
		Morone saxatilis	Striped bass
	Centrarchidae	*Micropterus salmoides*	Largemouth bass
		Micropterus dolomieu	Smallmouth bass
		Micropterus punctulatus	Spotted bass
		Pomoxis annularis	White crappie
		Pomoxis nigromaculatus	Black crappie
		Ambloplites rupestris	Rock bass
		Lepomis macrochrius	Bluegill
		Lepomis microlophus	Redear sunfish
		Lepomis gulosus	Warmouth
		Lepomis cyanellus	Green sunfish
		Lepomis gibbosus	Pumpkinseed
		Lepomis humilis	Orangespotted sunfish
		Lepomis megalotis	Longear sunfish
	Percidae	*Perca flavenscens*	Yellow perch
		** *Sander vitreus*	** Walleye
		** *Sander canadensis*	** Sauger
		Percina phoxocephala	Slenderhead darter
		Percina maculate	Blackside darter
		Percina evides	Gilt darter
		Percina shumardi	River darter
		Percina caprodes	Northern logperch
		*** *Crystallaria asprella*	*** Crystal darter
		Ammocrypta clara	[B] Western sand darter

Checklist of Iowa fish (From Zohrer et al. 2005). These animals represent 71 genera, 26 families, and 16 orders continued.

Order	Family	*Scientific name*	Common name
		Etheostoma zonale	Banded darter
		Etheostoma nigrum	Johnny darter
		Etheostoma chlorosomum	[A] Bluntnose darter
		Etheostoma asprigene	Mud darter
		Etheostoma caeruleum	Rainbow darter
		Etheostoma exile	Iowa darter
		Etheostoma spectabile	[B] Orangethroat darter
		Etheostoma flabellare	Fantail darter
		Etheostoma microperca	[A] Least darter
	Sciaenidae	*Aplodinotus grunniens*	Freshwater drum

*Genus listed as *Notropsis* 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).
[A] Iowa endangered species.
[B] Iowa threatened species.
[C] Iowa species of special concern.
[E] Federally endangered species.

Checklist of Iowa mussels (Zohrer et al. 2005). These animals represent 35 genera, 5 families, and 2 orders.

Order	Family	*Scientific name*	Common name
Unionoida	Unionidae	*Actinonaias ligmentina*	Mucket
		Alasmidonta marginata	Elktoe
		Alasmidonta viridis	[A] Slippershell
		Amblema plicata	Three ridge
		Anodonta suborbiculata	Flat floater
		Anodontoides ferussacianus	[B] Cylinder
		Arcidens confragosus	Rock poctetbook
		Cyclonaias tuberculata	[B] Purple pimpleback
		Ellipsaria lineolata	[B] Butterfly
		Elliptio crassidens	Elephant ear
		Elliptio dilatata	Spike
		Epioblasma triquetra	Snuffbox
		Fusconaia ebena	Ebony shell
		Fusconaia flava	Wabash pigtoe
		Fusconaia ozarkensis	[A] Ozark pigtoe
		Lampsilis cardium	Plain pocketbook
		Lampsilis higginsi	[AE] Higgin's eye pearlymussel
		Lampsilis siliquoidea	Fatmucket
		Lampsilis teres anodontoides	[A] Yellow sandshell
		Lampsilis teres teres	[A] Slough sandshell
		Lasmigona camplanata	White heelsplitter
		Lasmigona compressa	[B] Creek heelsplitter
		Lasmigona costata	Fluted shell
		Leptodea fragilis	Fragile papershell
		Leptodea leptodon	[E]Scaleshell
		Ligumia recta	Black sandshell
		Ligumia subrostrata	Pondmussel
		Megalonaias nervosa	Washboard
		Obliquaria reflexa	Threehorn wartback
		Obovaria olivaria	Hickorynut
		Plethobasus cyphus	[A] Bullhead (Sheepnose)
		Pleurobema rubrum	Pyramid pigtoe
		Pleurobema sintoxia	[A] Round (Ohio River) pigtoe
		Potamilus alatus	Pink heelsplitter
		Potamilus capax	[E] Fat pockebook
		Potamilus ohiensis	Pink papershell
		Pyganodon grandis	Giant floater
		Quadrula fragosa	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
		Quadrula metanerva	Monkeyface
		Quadrula nodulata	Wartyback
		Quadrula pustulosa	Pimpleback
		Quadrula quadrula	[E] Mapleleaf
		Simpsonaias ambiua	Salamander mussel
		Strophitus undulates	[B] Strange floater (Squawfoot)
		Toxolasma parvus	Lilliput
		Tritogonia verrucosa	[A] Pistolgrip or Buckhorn
		Truncilla donaciformis	Fawnsfoot
		Truncilla truncata	Deertoe
		Uniomerus tetralasmus	Pondhorn
		Utterbackia imbecillis	Paper pondshell
		Venustaconcha ellipsiformis	[B] Ellipse
	Margaritiferidae	*Cumberlandia monodonta*	[A] Spectacle case
Veneroida	Sphaeriidae	*Musculium sp.* *Pisidium sp.* *Sphaerium sp.*	Fingernail clams
	Corbinulidae	*Corbicula fluminea*	Asian clam
	Dreissenidae	*Dreissena polymorpha*	Zebra mussel

[A] Iowa endangered species.
[B] Iowa threatened species.
[E] Federally endangered species.

APPENDIX 2

White-Nose Syndrome Decontamination Protocols For Bat Trapping Equipment

Decontamination Protocols
For Bat Trapping Equipment

Due to White-nosed syndrome, the following protocols are to be used on field equipment used to trap and handle bats within Iowa. Absolutely no equipment used on bats in other states should be used within Iowa's borders. We will follow Level 3 Decontamination Procedures (including everything outlines in Level 2 as well). As adapted (taken almost verbatim) from the Wyoming Strategic Plan for White-Nose Syndrome (Abel and Grenier, 2011):

Level 2 Decontamination Procedures

1. Place a maximum of one bat per cloth holding bag during each survey. Keep used and unused bags separated at all times. Alternatively, place one bat per paper bag or paper cup, dispose of used bags and cups.
2. Maintain several pairs of leather handling gloves. Wipe gloves down between bats with Lysol wipes and allow to dry (switch gloves between bats as much as possible).
3. Wash all cloth bags in a washing machine after soaking in a solution of Lysol for 15 minutes.
    a. Soak all holding bags in Lysol IC Quaternary Disinfectant Cleaner (or Professional Lysol Antibacterial All-purpose Cleaner) and rinse with clean water.
    b. Then wash all bags in washing machine before next use.
4. Clean & decontaminate bat processing equipment (e.g. calipers, rulers, etc) at the end of the survey. Use Lysol Disinfectant Wipes and rinse processing equipment with clean water.
    a. Wipe down calipers, rulers, all other equipment which touches bats, between touching another bat.
5. Clean & decontaminate all bat survey equipment (e.g. processing table, mist-net poles, clip boards, etc.) and personal field gear (e.g. head lamps, etc) after the end of the survey with Lysol Disinfectant Wipes. Rinse all gear with clean water after disinfection.
6. At the end of each survey, soak all mist nets and outer clothing for more than 15 minutes in Lysol IC Quaternary Disinfectant Cleaner, rinse mist nets with fresh water before re-using. Wash clothing in washing machine after soaking.

Level 3 Decontamination Procedures

1. Same procedures as above and:
2. At the end of each survey, soak all mist nets and outer clothing for more than 15 minutes in Lysol IC Quaternary Disinfectant Cleaner, rinse mist nets with fresh water before re-using. Wash clothing in washing machine after soaking.
3. Step in Lysol solution and wipe down boots.

White-Nose Syndrome Decontamination Protocol
U.S. Fish and Wildlife Service – Version 01.25.2011

I. GENERAL INFORMATION:

The US Fish and Wildlife Service (USFWS) strongly recommends, first and foremost, compliance with all cave1 closures, advisories, and regulations on all Federal, State, Tribal, and private lands. However, where such closures are not required or recommended, the following protocol outlines the best known procedures to help reduce the transmission of the fungus *Geomyces destructans (G.d.)*, believed to be the cause of white-nose syndrome (WNS), to important bat habitat and populations. WNS is responsible for significant bat mortality in eastern North America, and threatens bat populations across the continent.

1 The use of the word "cave" in this document includes natural caves, man-made mines, or any other site that may harbor G.d. spores.
2 Use of some products which contain quaternary ammonia, isopropanol, and other potentially harmful chemicals or boiling water in confined spaces needs to be approached carefully due to inhalation or contact risks of the product. Since products/procedures may also cause damage to clothing, gear, and sensitive electronic equipment, all users should be aware of these risks prior to entering cave environments. Use of personal protective equipment to reduce contact with the product is strongly encouraged, particularly if extended contact is anticipated or as recommended by the manufacturer. Always read and follow the MSDS information and all safety/use criteria for every product used.
3 The active ingredient is considered to be at the effective concentrations known to kill the conidia of *Geomyces spp.*; however, the efficacy of field application remains to be demonstrated. Any equipment decontaminated with this product should be used with extra precaution until laboratory results are finalized.

If not properly trained and/or permitted by the appropriate government agency; then please do not handle bats. If you observe live or dead bats (multiple individuals in a single location) that may exhibit signs of WNS, contact a wildlife professional in your state wildlife agency (http://www.fws.gov/offices/statelinks.html) or contact your nearest USFWS Ecological Services Field Office (http://www.fws.gov/offices/). Researchers, contact your state or federal agency for permitting requirements.

II. RECOMMENDED DECONTAMINATION PRODUCTS:

All necessary and appropriate precautionary use, storage, and disposal information should be apparent on each of the product labels. It is critical that all researchers and biologists read and follow all label instructions provided on the products mentioned in this protocol. It would be a violation of federal law to use, store, or dispose of a regulated product in any manner not prescribed on the approved label/MSDS.

The following chemical (a minimum of 0.3% quaternary ammonium compound, unless otherwise denoted) and natural products were tested in the laboratory and determined effective for killing the conidia of *Geomyces* spp.:
1. Lysol® IC Quaternary Disinfectant Cleaner - (A product effective at 1:128 dilution, or 1 ounce of concentrate per gallon of water.) 2

2. Professional Lysol® Antibacterial All-purpose Cleaner (A product effective at 1:128 dilution, or 1 ounce of concentrate per gallon of water.) 2
3. Formula 409® Antibacterial All-Purpose Cleaner (Off-the-shelf concentrations as specified by label) 2
4. A 10% solution of household bleach – (A product effective at 1 part bleach to 9 parts water) 2
5. Lysol® Disinfecting Wipes (0.28 % di-methyl benzyl ammonium chloride) 2 & 3
6. Boiling in water for 15 minutes 2

III. DECONTAMINATION PROCEDURES:

BEFORE EACH CAVE VISIT: In order to effectively reduce the risk for human transfer of *G.d.*, it is imperative that everyone follow these decontamination procedures any time you plan cave visits. **Under no circumstances should clothing, footwear or gear that was used in a WNS-affected state or region be used in a WNS-unaffected state or region.** Clothing or gear that has been or is suspected of being exposed to *G.d.* may be reused in other WNS affected caves; however, the WNS decontamination procedures provided in this document should always be followed for items used in affected caves prior to entering another affected cave or leaving the affected state or region. Used gear that must be transported out of affected states or regions should be decontaminated, contained, and sealed prior to leaving the affected area and should not be stored or transported in close proximity with unexposed equipment. If gear cannot be decontaminated, either for safety reasons or fear that equipment may be damaged, it should not enter subsequent caves but rather be designated for use in that one specific cave. Gear should not be used in multiple caves in the **same day** unless the decontamination procedures below can be performed **between each cave visit or the maximum distance between visited caves is less than 10 miles (see supplements for explanation).**

AFTER EACH CAVE VISIT*:* Thoroughly scrape or brush off any dirt and mud from clothing, boots, and gear. Then place all in a sealed plastic bag or plastic container (with lid) to be cleaned and disinfected off-site. At a minimum, outer clothing should be removed prior to entering a vehicle after/between a cave visit. A clean change of clothing is recommended. **Care should be exercised at all times to prevent contamination of clean clothing, equipment, and/or vehicles.** To decontaminate clothing, footwear and gear, please follow all relevant procedures listed below.

A. Submersible Gear (i.e. clothing and equipment that can be submerged without damage)**:**
Wash all clothing and any appropriate equipment in washing machine or by hand using conventional detergents in cold, warm, or hot water. Woolite® fabric wash has been found to be highly effective for this procedure. Rinse thoroughly, and then follow by soaking for a minimum of 10 minutes in one of the decontaminating products in Section II, then rinse and air dry. Please notice when boiling water is selected as the decontamination method, all gear must be submersed for 15 minutes, then followed by air drying.

1. Footwear:

When safety permits, rubber (wellington-type) caving boots (which withstand harsh decontaminating products and are easily cleaned) are recommended. Boots need to be fully scrubbed and rinsed to remove all soil and organic material. Decontaminate rubber and leather boots, (including soles and leather uppers) with a product listed in Section II for a minimum of 10 minutes, then rinse and air dry.

2. Ropes and Harnesses:

To date, only Sterling rope and webbing have proved to sustain no damage when using the following procedure. Wash rope/webbing in a front loading washing machine on the gentle cycle using Woolite® Extra Delicates detergent. Immerse in a dilution of Lysol IC Quaternary Disinfectant Cleaner for 15 minutes. Rinse twice in clean water and air dry. Brands of rope/webbing other than Sterling have not yet been tested for integrity after decontamination. Brands not tested should be dedicated to a single cave or not used at all.

B. Non-submersible Gear (i.e. equipment that will be damaged by submersion)**:**
Clean thoroughly with soap (i.e. Dawn® antibacterial dish soap) and water, where appropriate, and then decontaminate all equipment by applying one of the recommended chemical products (understanding certain products are tougher on surfaces than others) in Section II to the outside surface for a minimum of 10 minutes, then rinse and air dry.

1. Cameras and Electronic Equipment:

If possible, do not bring electronic equipment into a cave. If practical, cameras and other similar equipment that must be used in a cave may be placed in plastic casing (i.e. underwater camera housing) or wrapped in plastic wrap where only the lens is left unwrapped to allow for photos to be taken. The plastic casing should be decontaminated using one of the appropriate products in Section II. The plastic wrap should be discarded after use and followed up by decontaminating the camera surface with Lysol ® Disinfecting Wipes, realizing this could damage the body of the camera.

2. Vehicles:

In addition to gear, vehicles used to transport equipment can also harbor spores. Keep vehicles as clean as possible by taking extra precautions (e.g. storing gear in clean containers, bringing a change of clothes, conducting all work outside of the vehicle once in the cave) and decontaminating storage containers along with all other clothing, gear, and misc. equipment using the appropriate decontamination products in Section II.

Note: *Protocol updated as of 01.25.2011. Please visit http://www.fws.gov/WhiteNoseSyndrome/ for updated materials and for* ***supplemental documents that detail decontamination procedures for 1. cavers, and 2. researchers****.*

APPENDIX 3

TURTLE MARKING CODES
AND
HERPETOFAUNA PHOTO VOUCHER GUIDELINES

Turtle Marking Codes & Herpetofauna Photo Vouchering

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	HNV	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	LNV	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	KLV	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	AHVW	AILP	AIPW	AJMN	AJVW
ABCP	ABIV	ABLP	ABPW	ACIO	ACLM	ACOX	AHJN	AHMO	AHVX	AILQ	AIPX	AJMO	AJVX
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	AHNW	AIJW	AIMX	AJKO	AJNW	AKMN
ABHO	ABJW	ABMX	ACHL	ACJP	ACMQ	AHIJ	AHKP	AHNX	AIJX	AINO	AJKP	AJNX	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	AMOQ	ANWX	BCHP	BCKN	BCOP	BHJM	BHMQ	BIJN	BIMV	BJKP	VJOV	BKNO
AKNP	ALNP	AMOV	AOPQ	BCHQ	BCKO	BCOQ	BHJN	BHMV	BIJO	BIMW	BJKQ	BJOW	BKNP
AKNQ	ALNQ	AMOW	AOPV	BCHV	BCKP	BCOV	BHJO	BHMW	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	BHNQ	BIJX	BINV	BJLN	BJPX	BKOP
AKOQ	ALOQ	AMPX	AOQX	BCIL	BCLM	BCPW	BHJX	BHNV	BIKL	BINW	BJLO	BJQV	BKOQ
AKOV	ALOV	AMQV	AOVW	BCIM	BCLN	BCPX	BHKL	BHNW	BIKM	BINX	BJLP	BJQW	BKOV
AKOW	ALOW	AMQW	AOVX	BCIN	BCLO	BCQV	BHKM	BHNX	BIKN	BIOP	BJLQ	BJQX	BKOW
AKOX	ALOX	AMQX	AOWX	BCIO	BCLP	BCQW	BHKN	BHOP	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	BHVW	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	HNVW	IJKQ	IJOW	IKNP	ILNP	IMOV	IOPQ	JKMN	JLMN
HJKO	HJOQ	HKMX	HLMX	HMOP	HNVX	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	LNVW	MNPQ	MQWX	OQVW
JMOP	JNVX	KLMX	KMOP	KNVX	LMOP	LNVX	MNPV	MVWX	OQVX
JMOQ	JNWX	KLNO	KMOQ	KNWX	LMOQ	LNWX	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	

HERPETOFAUNA 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 in 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 *Rana* species listed by the photo angle. *Rana* 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. sphenocephala*)
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.

Hyla chrysoscelis:

Adair	Calhoun	Delaware	Howard	Lucas	Scott
Adams	Carroll	Des Moines	Humbolt	Lyon	Shelby
Allamakee	Cass	Dickinson	Ida	Marshall	Tama
Audubon	Cedar	Dubuque	Iowa	Mitchell	Taylor
Benton	Cerro Gordo	Fayette	Jackson	Monroe	Union
BlackHawk	Chickasaw	Floyd	Jasper	Montgomery	Van Buren
Boone	Clarke	Franklin	Johnson	Osceola	Warren
Bremer	Clay	Grundy	Jones	Page	Wayne
Buchanan	Clayton	Hancock	Keokuk	Palo Alto	Winneshiek
Buena Vista	Clinton	Hardin	Kossuth	Pocohontas	Webster
Butler	Dallas	Henry	Linn	Poweshiek	Wright

Hyla versicolor:

Adair	Cerro Gordo	Floyd	Jasper	O'Brien	Story
Adams	Cherokee	Franklin	Jefferson	Osceloa	Tama
Audubon	Chickasaw	Greene	Jones	Page	Taylor
Black Hawk	Clarke	Grundy	Kossuth	Palo Alto	Union
Bremer	Clay	Guthrie	Lee	Plymouth	Van Buren
Buchanan	Clinton	Hamilton	Lucas	Pocahontas	Wapello
Buena Vista	Crawford	Hancock	Lyon	Pottawattamie	Warren
Butler	Dallas	Hardin	Marshall	Poweshiek	Webster
Calhoun	Dickinson	Howard	Mills	Sac	Winnebago
Carroll	Dubuque	Humbolt	Mitchell	Scott	Woodbury
Cass	Emmet	Ida	Monroe	Shelby	Worth
Cedar	Fayette	Iowa	Montgomery	Sioux	Wright

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 4

MACROINVERTEBRATE SAMPLING

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 in^2 (0.094 m^2) (OEPA 1989). Placement of the spacers between the wood plates on a ¼" (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 to 1.5 feet per second (15.2 cm to 45.7 cm per second).
2. Deploy the artificial substrates in runs with depths of 1 to 3 feet (30.8 to 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 and 8 inches (10.2 to 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 to 5 feet (0.91 to 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 in 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 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., Chronomidae 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 *5*

FISH ABNORMALITIES

Appendix 5
Fish External Abnormalities

Methods for Examinations of Fish External Abnormalities – Adopted from the Ohio EPA, and *copied verbatim from IDNR 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 carebaiis*, 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 contact or elongate their body. They may occur almost anywhere on the external surface of the fish, but are most frequently seen on the anterioventral surface of bullheads (*Ictaluras* 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 fished

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 urges 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 (*Icthyophthirus multifilis*).
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).
P	External parasites (other than those already specified).
W	Swirled scales.
Y	Popeye disease.
Z	Wound, other, not included above.

APPENDIX 6

CRAYFISH MARKING CODES

Crayfish Marking Codes

APPENDIX: All possible semi-permanent mark combinations by punching 1-3 holes in the telson and/or uropods of crayfish (obtained from Guan 1997).

1	1’	10	12	13	14	15	16	17	18	19
2	2’	20	23	24	25	26	27	28	29	1’0’
3	3’	30	34	35	36	37	38	39	2’0’	1’2’
4	4’	40	45	46	47	48	49	3’0’	2’3’	1’3’
5	5’	50	56	57	58	59	4’0’	3’4’	2’4’	1’4’
6	6’	60	67	68	69	5’0’	4’5’	3’5’	2’5’	1’5’
7	7’	70	78	79	6’0’	5’6’	4’6’	3’6’	2’6’	1’6’
8	8’	80	89	7’0’	6’7’	5’7’	4’7’	3’7’	2’7’	1’7’
9	9’	90	8’0’	7’8’	6’8’	5’8’	4’8’	3’8’	2’8’	1’8’
0	0’	9’0’	8’9’	7’9’	6’9’	5’9’	4’9’	3’9’	2’9’	1’9’
1’0		1’1	1’2	1’3	1’4	1’5	1’6	1’7	1’8	1’9
2’0		2’1	2’2	2’3	2’4	2’5	2’6	2’7	2’8	2’9
3’0		3’1	3’2	3’3	3’4	3’5	3’6	3’7	3’8	3’9
4’0		4’1	4’2	4’3	4’4	4’5	4’6	4’7	4’8	4’9
5’0		5’1	5’2	5’3	5’4	5’5	5’6	5’7	5’8	5’9
6’0		6’1	6’2	6’3	6’4	6’5	6’6	6’7	6‘8	6’9
7’0		7’1	7’2	7’3	7’4	7’5	7’6	7’7	7’8	7’9
8’0		8’1	8’2	8’3	8’4	8’5	8’6	8’7	8’8	8’9
9’0		9;1	9’2	9’3	9’4	9’5	8’6	9’7	9’8	9’9
0’0		0’1	0’2	0’3	0’4	0’5	0’6	0’7	0’8	0’9
120		123	124	125	126	127	128	129	130	134
135		136	137	138	139	140	145	146	147	148
149		150	156	157	158	159	160	167	168	169
170		178	179	180	189	190	230	234	235	236
237		238	239	240	245	246	247	248	249	250
256		257	258	259	260	267	268	269	270	278
279		280	289	290	340	345	346	347	348	349
350		356	357	358	359	360	367	368	369	370
378		379	380	389	390	450	456	457	458	459
460		467	468	469	470	478	479	480	489	490
560		567	568	569	570	578	579	580	589	590
670		678	679	680	689	690	780	789	790	890
1’10		1’12	1’13	1’14	1’15	1’16	1’17	1’18	1’19	1‘20
1’23		1’24	1’25	1’26	1’27	1‘28	1’29	1’30	1’34	1’35
1’36		1’37	1’38	1‘39	1’40	1’45	1’46	1‘47	1’48	1’49
1’50		1’56	1’57	1’58	1’59	1’60	1’67	1’68	1’69	1’70
1’78		1’79	1’80	1’89	1’90	2’10	2’12	2’13	2’14	2’15
2’16		2’17	2’18	2’19	2’20	2’23	2’24	2’25	2’26	2’27
2’28		2’29	2’30	2’34	2’35	2’36	2’37	2’38	2’39	2’40
2’45		2’46	2’47	2’48	2’49	2’50	2’56	2’57	2’58	2’59
2’60		2’67	2’68	2’69	2’70	2’78	2’79	2’80	2’89	2’90
3’10		3’12	3’13	3’14	3’15	3’16	3’17	3’18	3’19	3’20

APPENDIX continued: All possible semi-permanent mark combinations by punching 1-3 holes in the telson and/or uropods of crayfish (obtained from Guan 1997).

3’23	3’24	3’25	3’26	3’27	3’28	3’29	3’30	3’34	3’35
3’36	3’37	3’38	3’39	3’40	3’45	3’46	3’47	3’48	3’49
3’50	3’56	3’57	3’58	3’59	3’60	3’67	3’68	3’69	3’70
3’78	3’79	3’80	3’89	3’90	4’10	4’12	4’13	4’14	4’15
4’16	4’17	4’18	4’19	4’20	4’23	4’24	4’25	4’26	4’27
4’28	4’29	4’30	4’34	4’35	4’36	4’37	4’38	4’39	4’40
4’45	4’46	4’47	4’48	4’49	4’50	4’56	4’57	4’58	4’59
4’60	4’67	4’68	4’69	‘470	4’78	4’79	4’80	4’89	4’90
5’10	5’12	5’13	5’14	5’15	5’16	5’17	5’18	5’19	5’20
5’23	5’24	5’25	5’26	5’27	5’28	5’29	5’30	5’34	5’35
5’36	5’37	5’38	5’39	5’40	5’45	5’46	5’47	5’48	5’49
5’50	5’56	5’57	5’58	5’59	5’60	5’67	5’68	5’69	5’70
5’78	5’79	5’80	5’89	5’90	6’10	6’12	6’13	6’14	6’15
6’16	6’17	6’18	6’19	6’20	6’23	6’24	6’25	6’26	6’27
6’28	6’29	6’30	6’34	6’35	3’36	6’37	6’38	6’39	6’40
6’45	6’46	6’47	6’48	6’49	6’50	6’56	6’57	6’58	6’59
6’60	6’67	6’68	6’69	6’70	6’78	6’79	6’80	6’89	6’90
7’10	7’12	7’13	7’14	7’15	7’16	7’17	7’18	7’19	7’20
7’23	7’24	7’25	7’26	7’27	7’28	7’29	7’30	7’34	7’35
7’36	7’37	7’38	7’39	7’40	7’45	7’46	7’47	7’48	7’49
7’50	7’56	7’57	7’58	7’59	7’60	7’67	7’68	7’69	7’70
7’78	7’79	7’80	7’89	7’90	8’10	8’12	8’13	8’14	8’15
8’16	8’17	8’18	8’19	8’20	8’23	8’24	8’25	8’26	8’27
8’28	8’29	8’30	8’34	8’35	8’36	8’37	8’38	8‘39	8’40
8’45	8’46	8’47	8’48	8’49	8’50	8’56	8’57	8’58	8’59
8’60	8’67	8’68	8’69	8’70	8’78	8’79	8’80	8’89	8’90
9’10	9’12	9’13	9’14	9’15	9’16	9’17	9’18	9’19	9’20
9’23	9’24	9’25	9’26	9’27	9’28	9’29	9’30	9’34	9’35
9’36	9’37	9’38	9’39	9’40	9’45	9’46	9’47	9’48	9’49
9’50	9’56	9’57	9’58	9’59	9’60	9’67	9’68	9’69	9’70
9’78	9’79	9’80	9’89	9’90	0’10	0’12	0’13	0’14	0’15
0’16	0’17	0’18	0’19	0’20	0’23	0’24	0’25	0’26	0’27
0’28	0’29	0’30	0’34	0’35	0’36	0’37	0’38	0’39	0’40
0’45	0’46	0’47	0’48	0’49	0’50	0’56	0’57	0’58	0’59
0’60	0’67	0’68	0’69	0’70	0’78	0’79	0’80	0’89	0’90
1’0’0	1’0’1	1’0’2	1’0’3	1’0’4	1’0’5	1’0’6	1’0’7	1’0’8	1’0’9
1’2’0	1’2’1	1’2’2	1’2’3	1’2’4	1’2’5	1’2’6	1’2’7	1’2’8	1’2’9
1’3’0	1’3’1	1’3’2	1’3’3	1’3’4	1’3’5	1’3’6	1’3’7	1’3’8	1’3’9
1’4’0	1’4’1	1’4’1	1’4’3	1’4’4	1’4’5	1’4’6	1’4’7	1’4’8	1’4’9
1’5’0	1’5’1	1’5’2	1’5’3	1’5’4	1‘5’5	1’5’6	1’5’7	1’5’8	1’5’9
1’6’0	1’6’1	1’6’2	1’6’3	1’6’4	1’6’5	1’6’6	1’6’7	1’6’8	1’6’9
1’7’0	1’7’1	1’7’2	1’7’3	1’7’4	1’7’5	1’7’6	1’7’7	1’7’8	1’7’9

APPENDIX continued: All possible semi-permanent mark combinations by punching 1-3 holes in the telson and/or uropods of crayfish (obtained from Guan 1997).

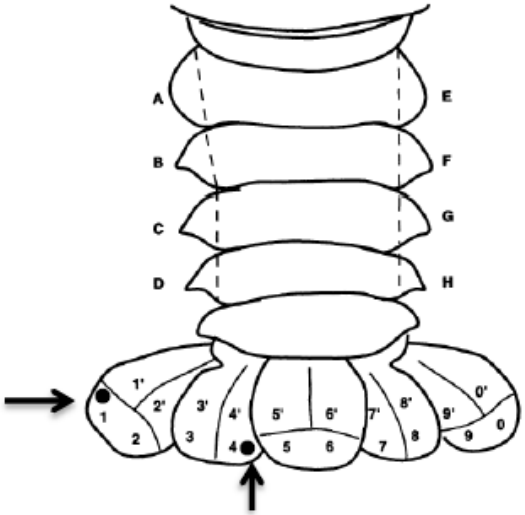
1’8’0	1’8’1	1’8’2	1’8’3	1’8’4	1’8’5	1’8’6	1’8’7	1’8’8	1’8’9
1’9’0	1’9’1	1’9’2	1’9’3	1’9’4	1’9’5	1’9’6	1’9’7	1’9’8	1’9’9
2’0’0	2’0’1	2’0’2	2’0’3	2’0’4	2’5’0	2’0’6	2’0’7	2’0’8	2’0’9
2’3’0	2’3’1	2’3’1	2’3’3	2’3’4	2’3’5	2’3’6	2’3’7	2’3’8	2’3’9
2’4’0	2’4’1	2’4’2	2’4’3	2’4’4	2’4’5	2’4’6	2’4’7	2’4’8	2’4’9
2’5’0	2’5’1	2’5’2	2’5’3	2’5’4	2’5’5	2’5’6	2’5’7	2’5’8	2’5’9
2’6’0	2’6’1	2’6’2	2’6’3	2’6’4	2’6’5	2’6’6	2’6’7	2’6’8	2’6’9
2’7’0	2’7’1	2’7’2	2’7’3	2’7’4	2’7’5	2’7’6	2’7’7	2’7’8	2’7’9
2’8’0	2’8’1	2’8’2	2’8’3	2’8’4	2’8’5	2’8’6	2’8’7	2’8’8	2’8’9
2’9’0	2’9’1	2’9’2	2’9’3	2’9’4	2’9’5	2’9’6	2’9’7	2’9’8	2’9’9
3’0’0	3’0’1	3’0’2	3’0’3	3’0’4	3’0’5	3’0’6	3’0’7	3’0’8	3’0’9
3’4’0	3’4’1	3’4’2	3’4’3	3’4’4	3’4’5	3’’6	3’4’7	3’4’8	3’4’9
3’5’0	3’5’1	3’5’2	3’5’3	3’5’4	3’5’5	3’5’6	3’5’7	3’5’8	3’5’9
3’6’0	3’6’1	3’6’2	3’6’3	3’6’4	3’6’5	3’6’6	3’6’7	3’6’8	3’6’9
3’7’0	3’7’1	3’7’2	3’7’3	3’7’4	3’7’5	3’7’6	3’7’7	3’7’8	3’7’9
3’8’0	3’8’1	3’8’2	3’8’3	3’8’4	3’8’5	3’8’6	3’8’7	3’8’8	3’8’9
3’9’0	3’9’1	3’9’2	3’9’3	3’9’4	3’9’5	3’9’6	3’9’7	3’9’8	3’9’9
4’0’0	4’0’1	4’0’2	4’0’3	4’0’4	4’0’5	4’0’6	4’0’7	4’0’8	4’0’9
4’5’0	4’5’1	4’5’2	4’5’3	4’5’4	4’5’5	4’5’6	4’5’7	4’5’8	4’5’9
4’6’0	4’6’1	4’6’2	4’6’3	4’6’4	4’6’5	4’6’6	4’6’7	4’6’8	4’6’9
4’7’0	4’7’1	4’7’2	4’7’3	4’7’4	4’7’5	4’7’6	4’7’7	4’7’8	4’7’9
4’8’0	4’8’1	4’8’2	4’8’3	4’8’4	4’8’5	4’8’6	4’8’7	4’8’8	4’8’9
4’9’0	4’9’1	4’9’2	4’9’3	4’9’4	4’9’5	4’9’6	4’9’7	4’9’8	4’9’9
5’0’0	5’0’1	5’0’2	5’0’3	5’0’4	5’0’5	5’0’6	5’0’7	5’0’8	5’0’9
5’6’0	5’6’1	5’6’2	5’6’3	5’6’4	5’6’5	5’6’6	5’6’7	5’6’8	5’6’9
5’7’0	5’7’1	5’7’2	5’7’3	5’7’4	5’7’5	5’7’6	5’7’7	5’7’8	5’7’9
5’8’0	5’8’1	5’8’2	5’8’3	5’8’4	5’8’5	5’8’6	5’8’7	5’8’8	5’8’9
5’9’0	5’9’1	5’9’2	5’9’3	5’9’4	5’9’5	5’9’6	5’9’7	5’9’8	5’9’9
6’0’0	6’0’1	6’0’2	6’0’3	6’0’4	6’0’5	6’0’6	6’0’7	6’0’8	6’0’9
6’7’0	6’7’1	6’7’2	6’7’3	6’7’4	6’7’5	6’7’6	6’7’7	6’7’8	6’7’9
6’8’0	6’8’1	6’8’2	6’8’3	6’8’4	6’8’4	6’8’6	6’8’7	6’8’8	6’8’9
6’9’0	6’9’1	6’9’2	6’9’3	6’9’4	6’9’5	6’9’6	6’9’7	6’9’8	6’9’9
7’0’0	7’0’1	7’0’2	7’0’3	7’0’4	7’0’5	7’0’6	7’0’7	7’0’8	7’0’9
7’8’0	7’8’1	7’8’2	7’8’3	7’8’4	7’8’5	7’8’6	7’8’7	7’8’8	7’8’9
7’9’0	7’9’1	7’9’2	7’9’3	7’9’4	7’9’5	7’9’6	7’9’7	7’9’8	7’9’9
8’0’0	8’0’1	8’0’2	8’0’3	8’0’4	8’0’5	8’0’6	8’0’7	8’0’8	8’0’9
8’9’0	8’9’1	8’9’2	8’9’3	8’9’4	8’9’5	8’9’6	8’9’7	8’9’8	8’9’9
9’0’0	9’0’1	9’0’2	9’0’3	9’0’4	9’0’5	9’0’6	9’0’7	9’0’8	9’0’9
1’2’0’	1’2’3’	1’2’4’	1’2’5’	1’2’6’	1’2’7’	1’2’8’	1’2’9’	1’3’0’	1’3’4’
1’3’5’	1’3’6’	1’3’7’	1’3’8’	1’3’9’	1’4’0’	1’4’5’	1’4’6’	1’4’7’	1’4’8’
1’4’9’	1’5’0’	1’5’6’	1’5’7’	1’5’8’	1’5’9’	1’6’0’	1’6’7’	1’6’8’	1’6’9’
1’7’0’	1’7’8’	1’7’9’	1’8’0’	1’8’9’	1’9’0’	2’3’0’	2’3’4’	2’3’5’	2’3’6’

APPENDIX continued: All possible semi-permanent mark combinations by punching 1-3 holes in the telson and/or uropods of crayfish (obtained from Guan 1997).

2’3’7’	2’3’8’	2’3’9’	2’4’0’	2’4’5’	2’4’6’	2’4’7’	2’4’8’	2’4’9’	2’5’0’
2’5’6’	2’5’7’	2’5’8’	2’5’9’	2’6’0’	2’6’7’	2’6’8’	2’6’9’	2’7’0’	2’7’8’
2’7’9’	2’8’0’	2’8’9’	2’9’0’	3’4’0’	3’4’5’	3’4’6’	3’4’7’	3’4’8’	3’4’9’
3’5’0’	3’5’6’	3’5’7’	3’5’8’	3’5’9’	3’6’0’	3’6’7’	3’6’8’	3’6’9’	3’7’0’
3’7’8’	3’7’9’	3’8’0’	3’8’9’	3’9’0’	4’5’0’	4’5’6’	4’5’7’	4’5’8’	4’5’9’
4’6’0’	4’6’7’	4’6’8’	4’6’9’	4’7’0’	4’7’8’	4’7’9’	4’8’0’	4’8’9’	4’9’0’
5’6’0’	5’6’7’	5’6’8’	5’6’9’	5’7’0’	5’7’8’	5’7’9’	5’8’0’	5’8’9’	5’9’0’
6’7’0’	6’7’8’	6’7’9’	6’8’0’	6’8’9’	6’9’0’	7’8’0’	7’8’9’	7’9’0’	8’9’0’

Diagrams demonstrating selected individual mark combinations of crayfish (adapted from Guan 1997):

Mark Number = 14



Mark Number = 2’5