Iowa Multiple Species Inventory and Monitoring Program Technical Manual

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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 13 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 is 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 to address your question.

This document will be known as version 1. While the main components of the field protocols are not expected to change often, they may change every 3-5 years, or more frequently should problems be encountered which need to be addressed. Therefore, minor adjustments may be made on a yearly basis, including adding in definitions, changing the bait used for specific trapping regimes, nocturnal bird survey timing, etc.

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Chapter 1
Introduction

This manual was written in response to the need for a monitoring plan 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). The state of Iowa chose 296 species as those SGCN in the IWAP. 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):

<u>Census</u> – A complete count of individuals, objects, or items within a specific area and time period.

<u>Survey</u> – An incomplete count of individuals, objects, or items within a specified area and time period.

<u>Inventory</u> – 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).

<u>Monitoring</u> – 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 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 will 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 statewide.

This program incorporates 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 will be implemented on additional areas. The 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 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 if 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 8-18 of this manual describe the taxonomic protocols in detail. In addition to the faunal protocols, habitat data collection is described in Chapters 19 and 20. The protocols require various numbers of visits to each site per season.

OBJECTIVES:

The first stage for implementation of a monitoring program in Iowa is to inventory a random sampling of public and private lands through surveys. The inventory is conducted following the same procedures used in the monitoring program and will 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. Is more habitat needed or is it adequate?
        ii. Is the habitat high-quality or marginal?
        iii. Are there restoration opportunities or other management options?
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. Determine if there are additional stresses not specified by the IWAP.

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 BBS) 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.

Chapter 2
Sampling Design & Plot Establishment

The strength of the monitoring program design is based upon the random 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 included. 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 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. Ideally, federal entities and county conservation boards will be willing to partner with the IDNR to monitor lands in their ownership. It is expected that funding will be available from the State and Tribal Wildlife Grants program to aid both the DNR and other groups with funding for monitoring. It may also be possible that the IDNR would conduct the monitoring on the federal or CCB lands, depending on funding and personnel available.

Of the < 1% of Iowa land in DNR ownership, all areas 247 acres or larger (and some smaller areas within target wetland classes) were classified according to the 19 habitats outlined in the IWAP (Zohrer 2005). These habitats with their definitions 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 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 follows the ensuing procedure:

1. Areas were listed in Excel and assigned numbers using the random number generator in Excel 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.
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.

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.

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.

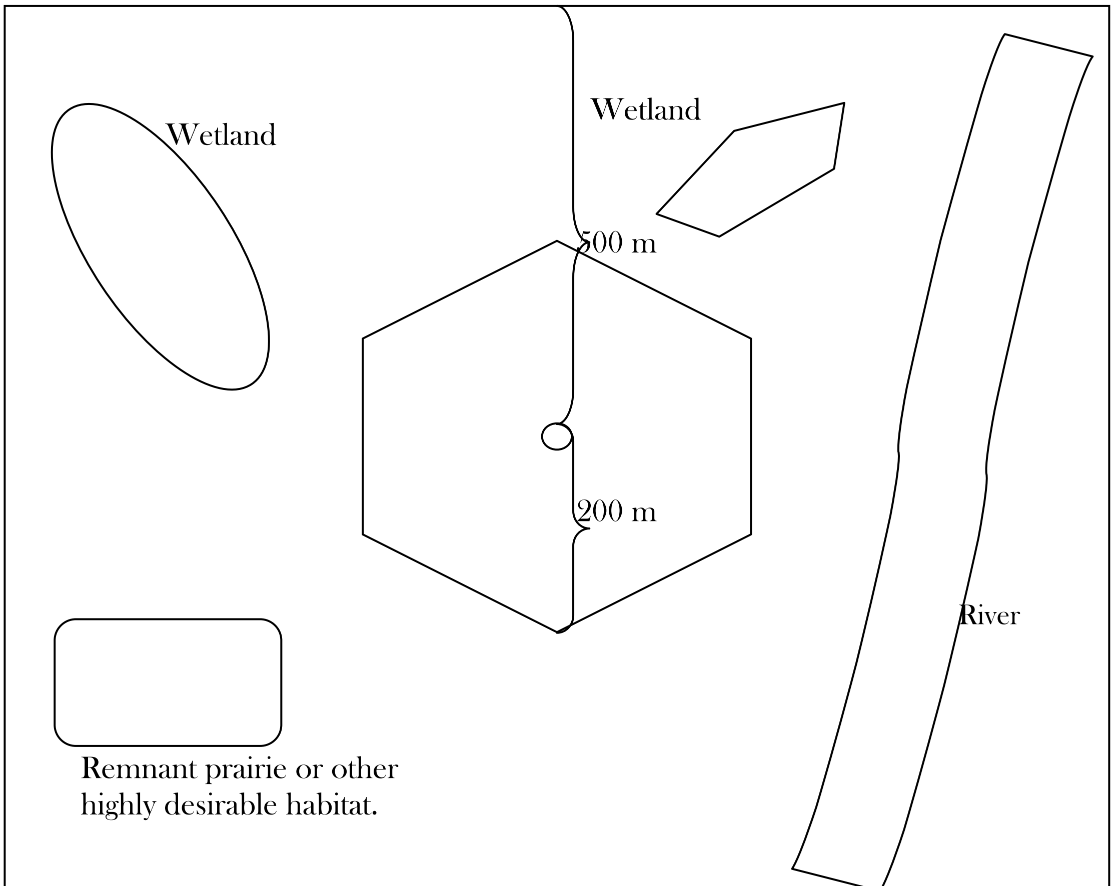


Illustration of a hexagon plot and associated sampling area. The distance from the center point to each outer point of the hexagon is 200 m, the distance from the center point to the outer edge of the additional sampling area is 500 m, or 1000 m centered on the middle point. Additional areas to be searched 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 18 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 this 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 trackplates 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 should 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.

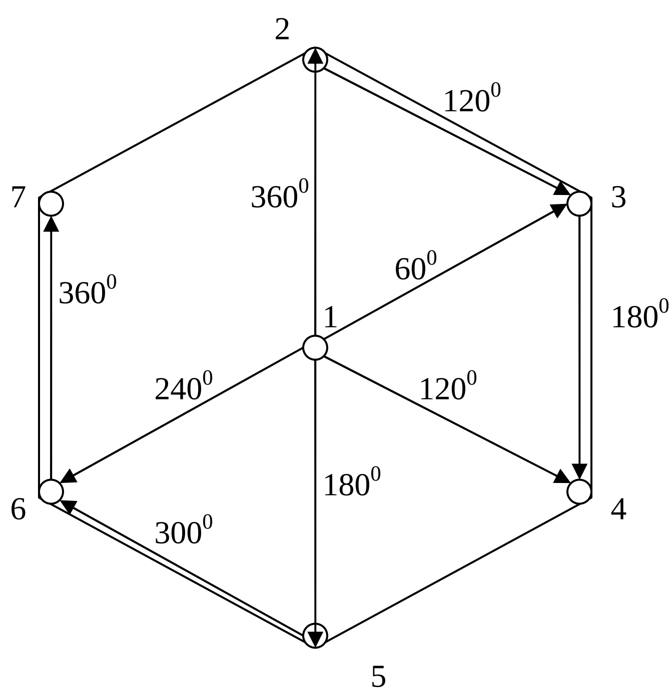
Remember that these protocols were designed so that organizations with limited resources and specific taxonomic interests can choose which protocols to implement should it be infeasible to employ all protocols.

Chapter Three
Landscape Characteristics Protocol
GPS/GIS

MONITORING:
Once the randomly chosen sites 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.

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. Use the following formulas to determine the locations of the outer 6 points that form the shape of the hexagon:

	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

If, however, the property is small (or the hexagon is placed) such that the outer points are < 10 meters from adjacent land which we do not have permission to be on, please move the effected points in to allow a 10 meter buffer from other property. This will most likely be an issue on private land areas.

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.

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 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.

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. 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:

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

Keep a small notebook with the camera – record the number of the photo with the 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. 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.

PHOTO/MAP BOOK:
During this stage of the program, a 3-ring binder 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. 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 GIS software and GPS unit during the 2-3 weeks of training at the beginning of the field season. This training should include practice surveys to ensure that proper procedures are followed. Ideally, the technician(s) hired will be proficient in the use of GIS.

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 in Appendix 2.

Chapter Four
Data Entry & Database Maintenance

<u>THIS CHAPTER IS STILL UNDER CONSTRUCTION. It is expected to change during the 2007 field season.</u>

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. If a program manager wished to share data with another program manager (i.e. if an Iowa county conservation board wished to share information with the Iowa DNR) then the databases should be constructed using software that will allow them to be merged or to be imported/exported to the other software application.

IDNR MSIM DATABASE:
The information in this chapter is specific to the database created by Stephanie Shepherd and maintained within the IDNR Wildlife Diversity Program. Other database programs could be used and this is left to the discretion of the programs other than IDNR that may be using these protocols. All information collected should be entered into the database. As multiple protocols may be implemented on the same day at the same site (thereby sharing the same weather information), it may be advisable to use a relational database which allows different tables to be connected to each other as opposed to entering the same weather information repeatedly.

For data to be inserted into the IDNR MSIM database it should be either entered directly into the database or input into a computer program in such a manner to allow it to be imported into the MSIM database. Currently, the MSIM database is in Microsoft Access. Data entered in Microsoft Excel can be imported into the MSIM database if the correct fields are included. Contact the IDNR WDP for additional information.

The following information 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.

UNIVERSAL FIELDS:
These are visible in a variety of forms.

ID Autonumber fields – these should be hidden for the most part.

Observers – This field should be entered using the initials provided separated by commas for multiple observers.

Species code – This field should be a drop down menu and restricted to the taxa being entered. The exception to this is the Incidental Report Form. New species can be added using the provided button on each of the forms. The new value should appear in the drop down list immediately. Please do a thorough search of the table before adding a new species to ensure it is not under a different name or spelling. This field is sorted in different ways (i.e. Latin name, species code, common name) depending on the taxa.

Time – Time should be entered as military time. The colon should be automatically inserted by the computer program. This field works easiest using the tab key.

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.

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 "Property-Entire" (E.g. "Jackson Property – Entire") that can be chosen as a site location. Choose this and enter UTMs (if known) for the survey site in comments.

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.

Habitat Data

The forms should flow (or be moved through) as follows:

1) *Wetland*: Data Entry Home – Habitat Data Entry Home – Habitat Survey Data – Wetland Switchboard – Lotic Habitat Plots Or Lentic Habitat Plots – then BACK TO Habitat Survey Data (in order to start a new record on the same property) or BACK TO Habitat Data Entry Home (in order to start a new record for a different property).

2) *Terrestrial*: Data Entry Home – Habitat Data Entry Home – Habitat Survey Data – Terrestrial Plot (this consists of a main page with several tabs of subforms) – then BACK TO Habitat Survey Data (in order to start new record on the same property) or BACK TO Habitat Data Entry Home (in order to start a new record for a different property).

Terrestrial Plot Form

1) Setup is a main form and then a series of tabs for all the subforms.

2) This form may give you some trouble with the tabs disappearing from view. Should this occur, just scroll back up to reveal the tabs.

3) Ground Cover and Tree Snag have a toggle switch as to whether the data being entered is "Within the interior plot" or not. These data are collected both within a 7.3

meter plot (within interior plot) and in a larger 17.6 meter plot (NOT within interior plot).

4) There are several fields that are setup with a default value, for these fields a value will already be entered in the field. To keep this value simply tab through the field.

5) The quadrat plant survey form has a subform within this subform. There are buttons to enter data for several quadrats ("Enter Next Quadrat") for each site within a property.

6) For Unknown or Incomplete Plant Species Record (i.e. records that need to be reviewed), there is an Unknown plant species option in the dropdown. Choose this and put all notes in the comments.

Species Survey Data

For the correct species button to appear at the bottom of the page you must click on a taxa in the taxa box. If you are trying to edit a record and need to get the button to come up it is all right to click the already highlighted taxa to get the button to appear.

Mammal Form

This form should flow as follows: Data Entry Home – Main Survey Form – Choosing "Mammals" in the Taxa box will make the mammal button appear.

Mammal Form – choose "Enter Next Site" button to enter a new record for a different site on the same property and the same day (using the same environmental data) – then BACK TO Main Survey Page ("Enter Next Survey" Button) (in order to change properties or to enter a new survey period (i.e. with different date & environmental information).

Notes on the mammal form -

1) SMT: Each visit is allocated a visit number (i.e. Mon. am = 1, Mon. pm = 2, Tues am = 3, Tues pm = 4, etc…).

2) Measurements are in mm or grams.

3) Peromyscus sp. – Choose this as the species for all mouse records which will need to be reexamined for species.

4) It is important to pay attention such that the 1)gender and 2) breeding status fields are not inconsistent with the 3)breeding condition details.

Additional information on forms will be placed here

GENERAL NOTES:

Helpful Shortcuts or Functions

a. Ctrl-F for find
b. Ctrl-' (Apostrophe) to copy field contents from previous record
c. If a data is in the current year, there is no need to enter year. E.g. If "6/26" is entered, Access converts this to "6/26/2006"
d. Drop down menus:
    i. Use the arrow to drop and click or
    ii. Start typing and the selection should pop up
    iii. Ctrl-' can still be used in these boxes

e. Instructions or descriptions for many of the fields should pop up in the bottom left corner of the screen when the cursor is in that field. This will sometimes provide instructions on the information to be entered.

WARNINGS:

1. Make sure when entering a new record that you are on a blank page. Changing a value in a drop down box does NOT create a new record. It changes an existing record.
2. Pay attention to the yellow box in the header as this tells you what property and site you for which the data being entered belongs. If it says “#Name?” then there is an error and the record will not be valid.
3. Use the buttons to navigate through the forms rather than the “X” in the right corner. Only use the “X” when adding a new species. In this case, the button will open a table to add a record and once finished, the “X” must be used to close the table and return to the form.
4. Remember the Main Form-Sub Form structure as it should help in maintaining the correct flow of the forms.
5. Read the instructions on each page carefully.
6. In you need additional codes, contact the program leader or database manager.

EDITING:

Note that all forms open on a new, blank record. There are several ways to find and edit an existing record.

<u>To Find a Record</u>

1. Use the back arrow. On most forms, the back arrow will allow you to scroll through each of the previous records.
2. If there are many records, the find tool may be faster. Place the cursor in the field to be searched and either press ctrl-F or go to Edit-Find and type in the value under scrutiny.
3. You may also filter records based on a particular field value (E.g. date or property name). Find the value you are looking for and place the cursor in that field. Press the button that looks like a funnel with a lightning bolt and then the filtered records should be able to be scrolled through. To unfilter the records, press the button with the funnel but no lightning bolt.

<u>Editing a Record on a Separate Subform</u>

For all Species Surveys and from the Main Habitat Page, first find the appropriate record on the Main Form and then go to the subform (remember it will open on a blank/new record) and use the scroll back button use the find tool here.

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

If one ignores the probability of detecting a species (and assumes that this probability is equal to 1 – meaning it is always found 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 analysis.

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. Also, this information could be used to help prioritize areas for conservation. If an area has been shown to have good population persistence for a certain species it might be ranked higher on a land acquisition list than an area with a larger extinction probability, perhaps.

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 (l) – 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 systemically placed. Typically an observer stays in the center of the point for a specified amount of time (e.g. 2 minutes) before beginning the data collection and remains standing at that location throughout the timed count (e.g. 10-12 minutes).

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

Requirements & Assumptions:

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

Advantages:

Since the locations are known prior to the start of data collection, distances at each location can be flagged, if necessary to aid in correct distance measuring. Other advantages (compared to line transect sampling) include that radial distances are easier to measure and that point counts are easier to 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 this 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, however, could be calculated on the number of visits needed per site, per year, IF the animals were marked, which may or may not be done 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.

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 with some of these techniques.

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 an animal.

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 may 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 areas 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 in 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
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 early 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.

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 sites 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 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 implementer to serve on the review board 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 review board, and monitoring partners. The report should be written and the program reviewed every 5 years. Again, responsibility for the report belongs to the program scientist with help from appropriate staff and monitoring partners.

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

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

SYMPOSIA, WORKSHOPS, AND CONFERENCES:
The purpose of participating would be to review and summarize the information collected on a specific species. 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 as suggested by the USGS 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.

Reviews should be conducted periodically and should include review of individual protocols, sampling location selection procedures, 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 review board 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 review board 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, sites, how the data was collected and analyzed?
3) Are the conclusions logical?

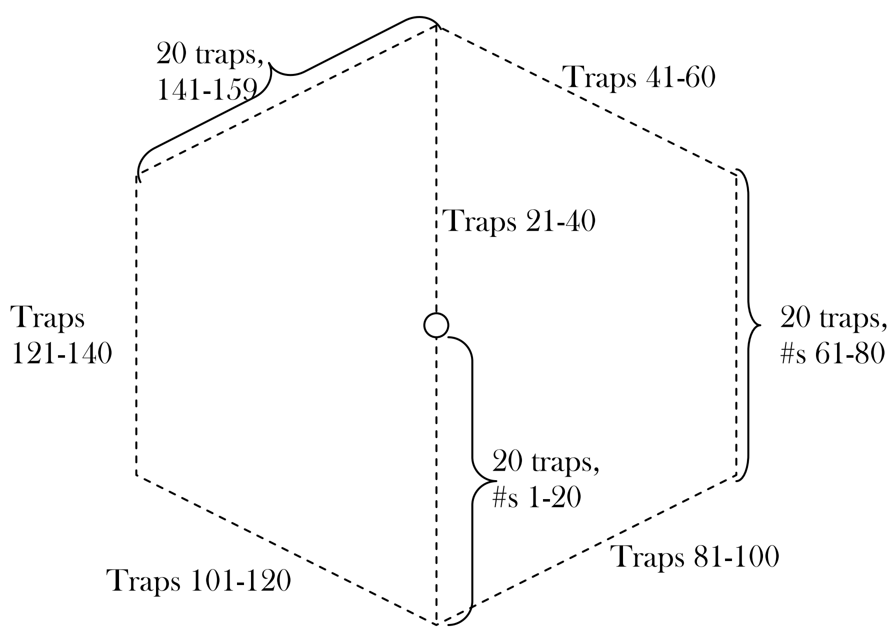
The files maintained as part of the review process should be in the possession of the program scientist (or a copy of these files), with another copy (or the originals) being maintained by the review coordinator. For Iowa, it would be appropriate for the IWAP implementer to fill the role of MSIM review coordinator, if the implementer 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, are located in Appendix 2 at the end of the data sheets.

Chapter Eight
Mammal Monitoring Protocol for Small, Medium, and Large Mammals

IOWA MAMMAL MONITORING:

Small Mammals

At each hexagonal sampling plot, the spacing between the traps will be 10 meters with the total number of traps to 159 per site, arranged around the arms of the hexagon and along the dividing transect. 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 traps for two reasons. The first is to allow this data to be compared to other studies that have followed the same protocol (although currently (2005) the only known studies are in California, but this is expected to change, Patricia Manley, personal communication). The second reason is that some research has indicated that the transect method is more efficient for a basic species inventory when density or abundance data is 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 more 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 is conducted between the last week of April and the middle of July over a 12 week period, with an additional 2 weeks in early to mid April being used for training. When the sites are visited in future years, care should be taken to ensure that they are trapped during similar times of the season to help standardize environmental conditions. Trap locations should be permanently marked or 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.

Traps are opened for 4 nights and checked twice daily, once in the morning and again in the late afternoon. 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. Given that only 12 weeks of the year will be monitored (as this is the time most small mammals are expected to be active), 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 12 weeks, 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.

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. 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.

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 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.

<u>Medium & Large Mammals</u>

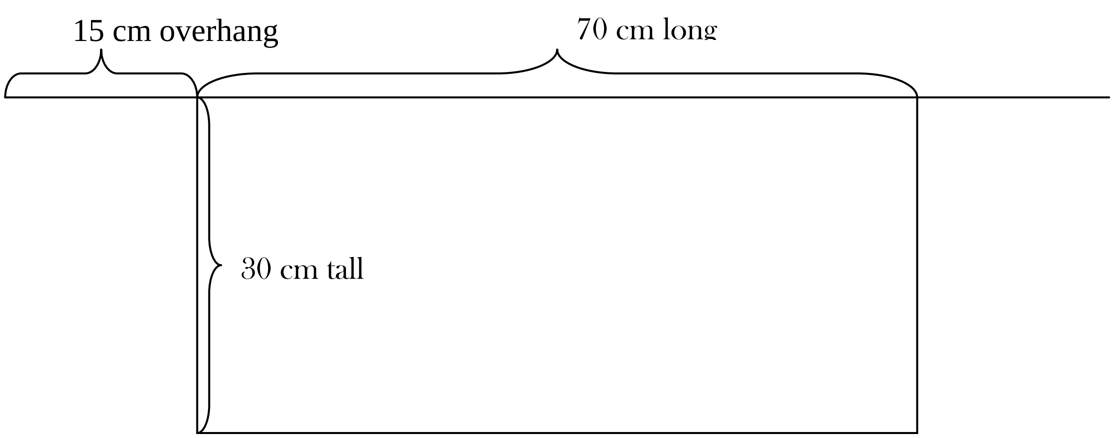
For medium and large mammals, trackplates and camera surveys will be the primary method of detection. The same protocols as those outlined in the FS MISM program will be followed, such that 3 track plate stations and 3 camera stations are arranged anywhere within the larger, 101 ha area of the property. Locations should be determined beforehand with the use of aerial photos and GIS.

The cameras are attached to a tree. If no suitable trees are found, then cameras (and bait if used) can be attached to stakes. Stakes MUST be able to withstand weather and animal activity. Until the first few years of the study have been completed, no additional methods will be used to track mammals unless the landowner requests such effort. If the landowner requests additional efforts, then those will be implemented dependent upon current funding situations.

Each survey encompasses a 10 day period during the summer. Each site is visited every other day for a total of 5 visits to replace bait, trackplate paper and ink, and film (if not using digital cameras).

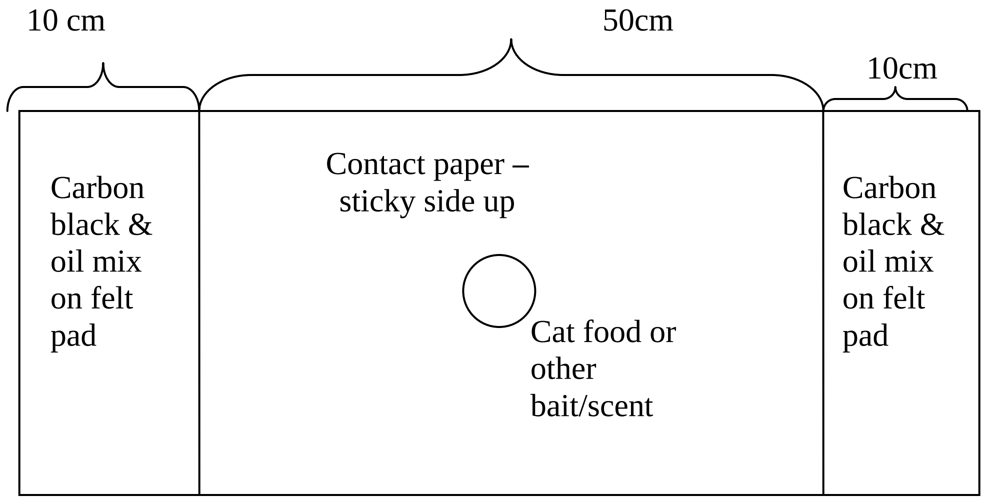
In addition, visual encounters for medium and large mammals, such as additional tracks, scat, foraging marks, or the actual animal, should also be noted on the data sheets along with the location.

The trackplate covers are made from 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 thesis 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.

THIS WILL CHANGE DEPENDING ON THE EQUIPMENT PURCHASED OR BORROWED: Camera stations include a 35mm camera with a Trail Master TM550 dual sensor, passive infra-red detector (Goodson & Associates, Inc, Lenexa, KS). Film is 35 mm ISO 400 and a flash is also used. Settings for the passive infra-red ***(Trail master? We used active for the trial plot)*** should be P = 5 & Pt = 2.5, which requires 5 full windows to be interrupted for at least 2.5 seconds before a photo is taken, and there is a 2 minute delay between shots. The camera and Trail master 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 an adjacent tree. Cameras and detectors are attached to the tree using a tripod, wires, nylon straps, and duct tape. A large feather should be hung 1.5 m above the ground to act as a visual cue. Film is checked and replaced as needed (Manley et al. 2004).

An additional attractant *could be* used for both the track plate and camera stations. This attractant is 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 can be purchased.

HABITAT AND PLANT SPECIES COMPOSITION 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 faunal monitoring data sheet. A habitat data collection plot should be established at every bird point count location which is the same as the end of every transect for small mammal traps.

See Chapter 19 for information on terrestrial habitat and plant composition measurements, and Chapter 20 for information on aquatic measurements. As the same areas will be searched for all species of greatest conservation need, habitat data collection instructions are included in these chapters. However, all data collection technicians should coordinate with other crews to ensure that all needed habitat data is collected.

EQUIPMENT NEEDED:
Per site:

Small mammal protocol:	159 Sherman traps, plus replacements, per site	
	Bait (rolled oats and birdseed or peanut butter)	
	Surveyor's tape	
	Compass	
	Polystyrene batting (or cotton balls in non-zip sandwich bags)	
	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 for traps	
	2 hand lenses (shrew ID)	
	Dust masks	
	Hand sanitizer	
	GPS unit to record trap locations	
Meso- & large mammals:		
Track plate stations:	3 bottom trays (track plates with contact paper)	
	6 binder clips	
	Duct tape	
	Acetylene torch/carbon black mix/carpenter's chalk	
	Contact paper	
	Putty or poster gum	
	Bait	
	3 tbs Gusto mixture	
	Roll of clear tape	
Camera stations:	3 cameras	or Deercams
	3 Trail masters	or Deercams
	3 wires	
	100 feet of 22 gauge bailing wire OR airplane cables	

Turkey feathers
3 tbs Gusto mixture
3 rolls ISO 400 35mm film
Camera batteries

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

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
Large area for trap drying
Garbage bags for garbage

STAFF & TRAINING:

Two weeks of training is recommended and should include 1) field guide use and id, 2) trips to University museums to discuss 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) soot, carbon, or chalk application, 6) set up and maintenance of track plate and camera stations. Crew will need a reference guide of local species' tracks for the clipboards.

DATA QUALITY & MANAGEMENT:

Small mammal data can be affected by:

- Trap placement: Should be checked periodically by supervisor.
- Observer handling care: Mortalities can be monitored through data, and should be <1%.
- Error in species ID: Difficult to monitor, therefore, could switch observer crews during week of trapping.

At the end of each trapping day, field crew pairs 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, trap function, and legibility.

The track plate and camera station data are 'independently verifiable and the data are subject to very little interpretation' (Manley et al. 2004, MELP 1998). This means that there are very few sources for technician errors in this protocol design, therefore there is no need for separate quality assurance teams. However, the set up of the track stations is critical and should be spot-checked by the crew supervisor periodically throughout the season.

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. (Chapter 5 Data Analysis).

Using a closed population model, such as a Lincoln Petersen estimation method, will allow for the estimation of population size, provided that animals are recaptured during the primary sampling period. For more information, see Chapter 5.

Photos, track-casts, and CONTACT paper will be archived for species ID 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 track plate or trail master stations per site should be adjusted.

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.

SAFETY CONSIDERATIONS:

Small mammals may carry many diseases which can be transferred to humans. Technician crews should be aware of the potential diseases and the associated symptoms. Gloves and dust masks should 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. Crews should work in teams of two and carry radios or 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 Iowa Wildlife Action Plan (Zohrer et al. 2005). Bats will be 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.

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
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
Red squirrel	*Tamiasciurus hudsonicus*	Forest
Plains pocket mouse	*Perognathus flavescens*	Prairie, sand & loess
Prairie vole	*Microtus ochrogaster*	Upland prairie
Red-backed vole	*Clethrionomys gapperi*	Forest
Southern bog lemming	*Synaptomys cooperi*	Moist grassland
Woodland vole	*Microtus pinetorum*	Forest
River otter	*Lutra canadensis*	Rivers, streams, & lakes
Spotted skunk	*Spilogale putoris*	Grassland, forest, farmsteads
Bobcat	*Lynx rufus*	Forest, woodland, grassland

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>Sherman Trap Array Augmentation</u>

2) Trap for longer duration.
3) Increase the number of traps (and therefore, the size of) the trapping area.
4) Arboreal small mammals are best trapped in trees, so in forested areas, could place additional Sherman traps in a trees.

<u>Track Plate & Camera Array Augmentation</u>

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 trackplate 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 would still be visited every other day and a cast of the tracks would 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 4^{th} Sherman trap for a total of 40 traps with 5 being on each transect. 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 ID'd, 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 small mammal crew if tomahawks are used. All employees involved with the tomahawk trapping will need to 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. 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.

DATA SHEETS:

Data sheets for this protocol are located in Appendix 2.

Chapter Nine
Bat Monitoring Protocol

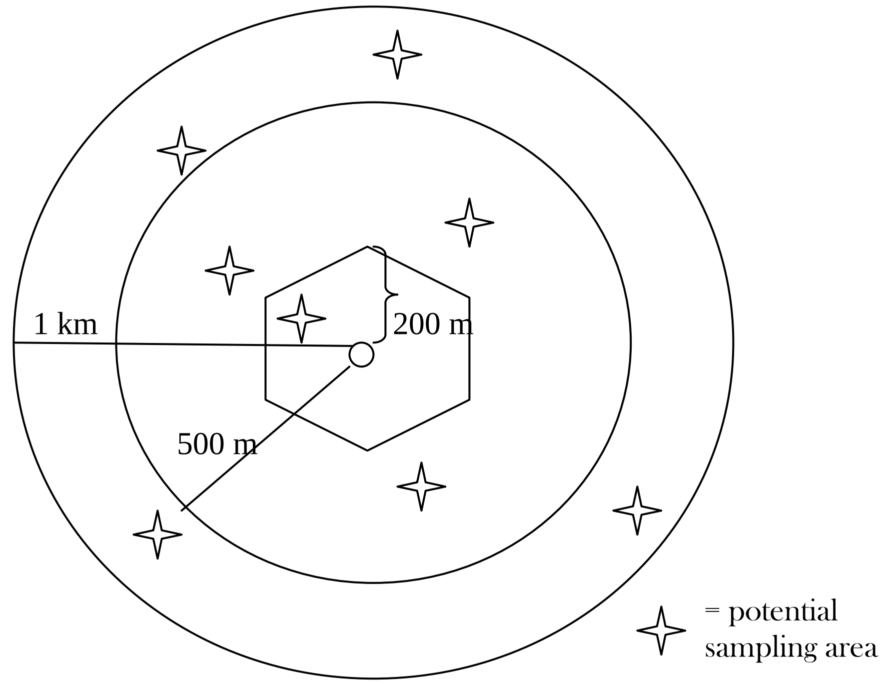
IOWA BAT MONITORING:
Bat detections will be done primarily using ANABAT (or other recording devices) detectors. At least 2 detectors should be deployed at suitable habitat locations for 3 occasions during the season, for a total of 6-9 nights of recording bat calls per year per detector.

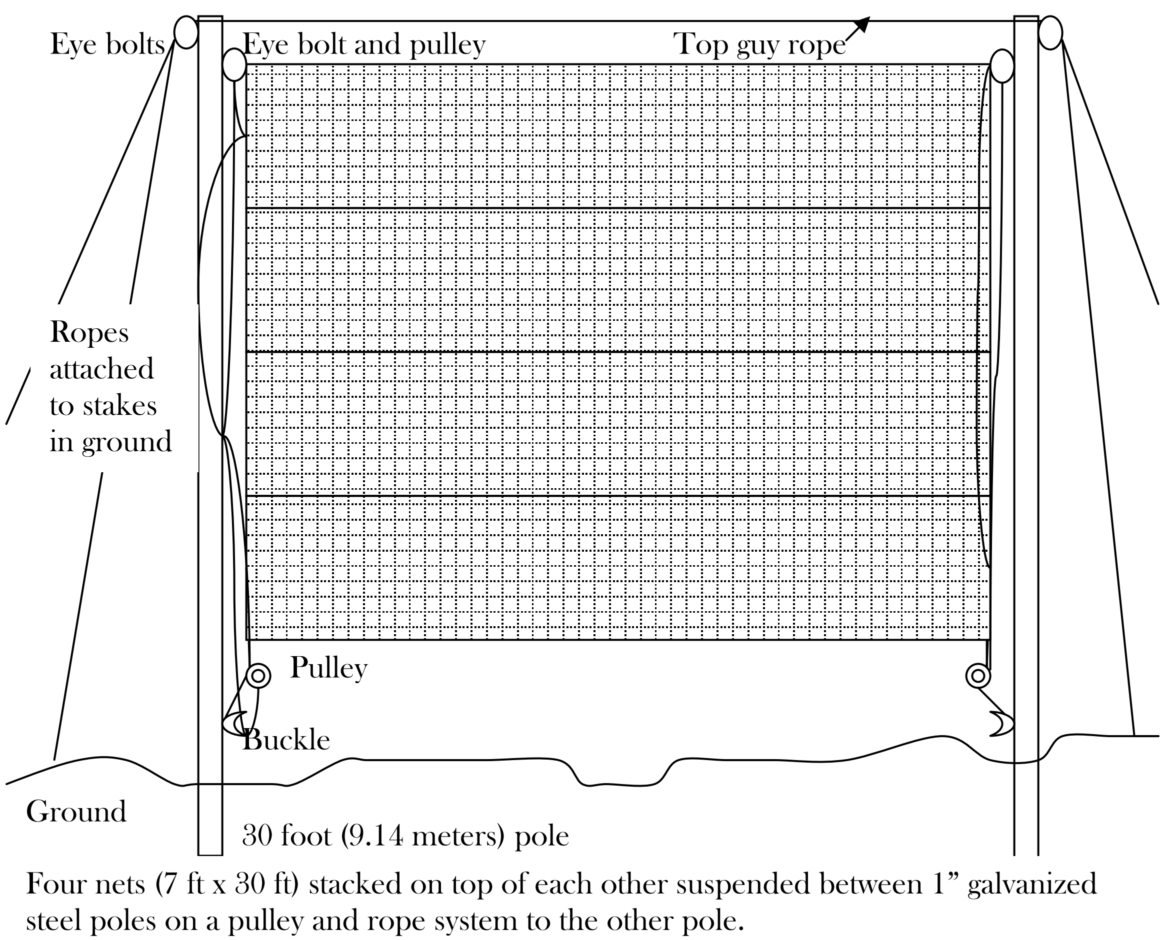
To establish regional keys for the ANABAT detectors, bats will be captured at different locations and the calls recorded using the detectors. Calls should be recorded both directly into the detector and also through the weather protection covers for the detectors. Mist nets will be the primary method used to capture bats. Suitable habitats will be chosen from aerial photos or GIS database within a 1 km^2 area with the center of the area also being the center of the permanent hexagonal sampling plot. 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:
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). Two to three ANABAT detectors should be deployed for 1-3 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 1-3 night timeframe. This process should be repeated 3 times during the summer season resulting in at least 6 nights and up to 9 nights of ANABAT deployment.

During the first year(s) of the program, trapping with mist nets will be conducted on a regular basis to ensure that adequate calls are collected from bats of known species. Throughout the remainder of the program, trapping will be done as necessary to determine species when calls cannot be identified.





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.

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 nets stacked such that the nets reach a height of 7 m.

Inclement weather conditions, including temperature below 10°C, 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.

HABITAT AND PLANT SPECIES COMPOSITION 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 data sheet. A habitat data collection plot should be established at every hexagonal point location.

See Chapter 19 for information on terrestrial habitat and plant composition measurements, and Chapter 20 for information on aquatic measurements. As the same areas will be searched for all species of greatest conservation need, habitat data collection instructions are included in these chapters. All data collection technicians should coordinate with other crews to ensure that all needed habitat data is collected.

However, in addition to the above data, potential roosting sites seen on the property should be noted on the data sheet. 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
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
3 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 (e.g. small cotton, GSA 'mailing bags' 8x10 inches)
Sunrise/sunset chart
Batting or gold gloves (leather)
Field guides
Night vision scope
Digital camera

STAFF & TRAINNING:
ANABATs can be deployed 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 id, and 2) trips to University museums to discuss defining species characteristics. Since these are delicate animals, technicians with mist net experience or bat experience should be given preference for hiring.

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. Alternatively, entire crews could be rotated among sites (so no 1 crew surveys the same site twice) to reduce potential bias in net placement within sampling areas.

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. Field supervisors should accompany crews on a rotating basis to check techniques.

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 sites occupied using program 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 bat 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 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 locations and status.

TARGET SPECIES:
The following list of target species represents the species of greatest conservation concern as chosen by the Steering committee for the Iowa Comprehensive Wildlife Conservation Plan (Zohrer et al. 2005). 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.

Target bat species:

Common Name	Scientific Name	Habitat
Evening bat	*Nycticeius humeralis*	Forest, riparian
Indiana bat	*Myotis sodalis*	Forest, upland, & riparian
Northern myotis	*Myotis septentrionalis*	Forest

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.

Roost Site Monitoring

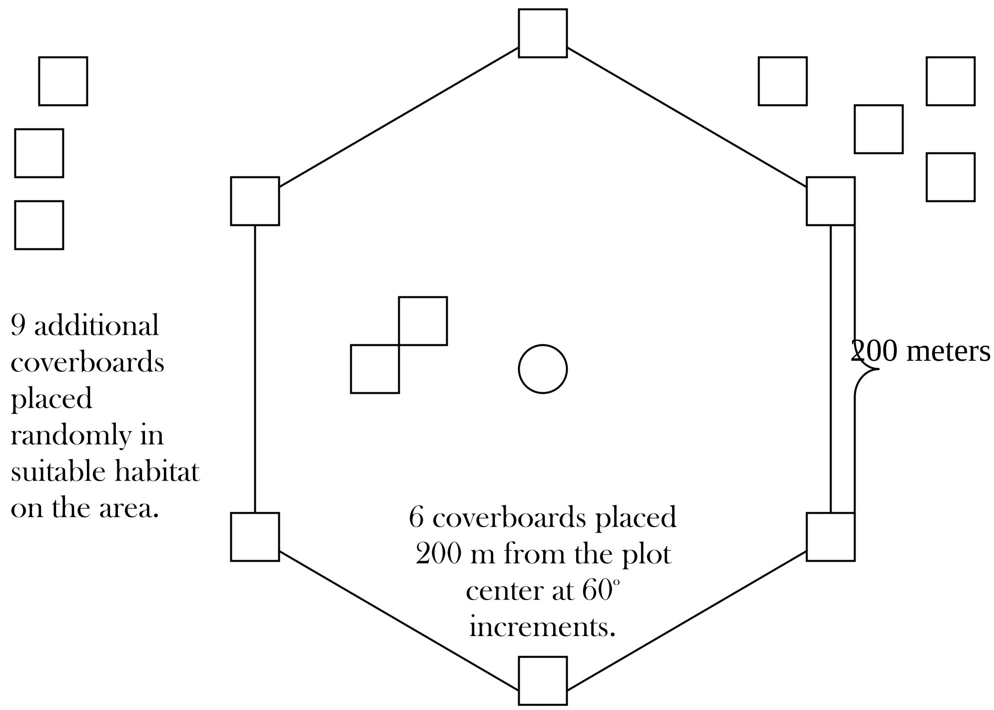
If the target species have potential roost sites (bridges, caves, etc) within the sampling unit, it may be beneficial to monitor these in addition to mist netting at habitat locations. Field visits will be needed to search for potential roost sites and determine the best location to watch bats exit the 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.

DATA SHEETS:

Data sheets for this protocol are located in Appendix 2.

Chapter Ten
Amphibian & Reptile Monitoring Protocol

IOWA 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 minnow traps. The diagram above shows the placement of cover boards. Minnow traps should be deployed in wetlands for 3 days (Kolozsvary and Swihart 1999) and checked daily.

SURVEY METHODS:
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 at least 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 transect, 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.

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. 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°, 60°, 120°, 180°, 240°, and 300°), another board should be placed 200 m from the plot center. Incidentally, this coincides with the placement of the poles to mark the bird point count locations. Place the board 1-2 meters from the pole to prevent 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. Locations of these boards should be recorded with a GPS unit and marked to allow them to be found in the future.

Minnow traps may be an effective way to find additional tadpoles (Shaffer et al. 1994). Minnow 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. 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.

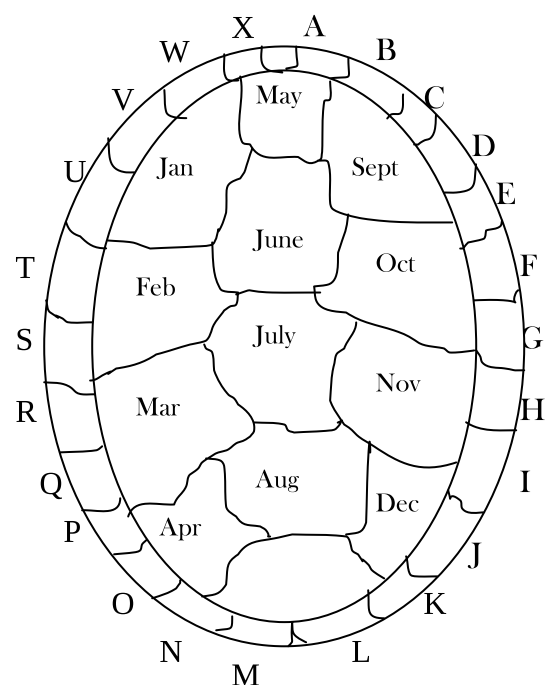
To protect the amphibian, it is critical that the technician's hands be free of any chemicals or lotions. Insect repellent can be absorbed through the skin of the amphibian, resulting in the animal's death.

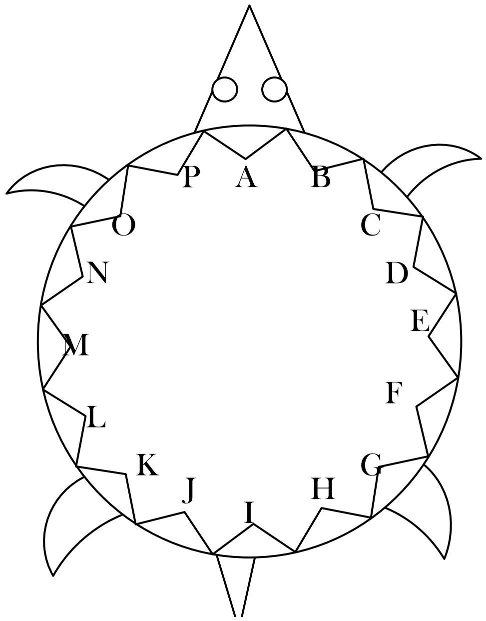
For each observation, record the time, the amount of time that has elapsed from the start of the search, species, detection type (visual, auditory, sign), age class (adult, sub-adult, juvenile), and substrate type (rock, log, bare ground, etc.). In addition, animals that are captured should be measured (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 3 (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 3). 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, following the marking codes in Appendix 3 regardless of family is advisable. Once these marks have all been assigned to individuals, further marks using the scutes labeled D, E, F, G, R, S, T, and U can be designed. 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 expected 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.

From Micheal Dorcas's website Last accessed 11/7/06
http://www.bio.davidson.edu/people/midorcas/research/Contribute/HerpLabProtocols2006-09-22.pdf

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Nocturnal Auditory Amphibian Counts

In addition to the VES, each sample plot should 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.

HABITAT AND PLANT SPECIES COMPOSITION 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.). The sampling plot may be classified as a prairie or lake stratification.

If so, the primary habitat measurements would be acquired following Chapter 19 (Terrestrial Plant Species and Habitat Monitoring) and Chapter 20. 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
Snake tubes for handling venomous snakes
Pair hip waders
15 Coverboards
Minnow traps
Frye nets
Hoop traps for turtles
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:
Two weeks of training is recommended and should include 1) field guide use and id, 2) trips to University museums to discuss 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 could be checked by either the use of digital cameras, or by the field supervisor working periodically with each technician.
- Manley et al. (2004) suggests rotating crew member 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.

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.

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 nocturnal auditory call data collected would also be analyzed using program MARK or PRESENCE to determine the proportion of areas occupied. Current technology lacks the ability to rigorously analyze the call index data, but we believe that advances in methodology will soon allow this 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 will most likely be rare enough that they may 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. If, however, there is reason to mark these animals (probably with passive integrated transponders (PIT–tags)), the safest way to handle a venomous snake is 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 will not need to be handled, but do photograph it (from a safe distance without disturbing it) if possible.

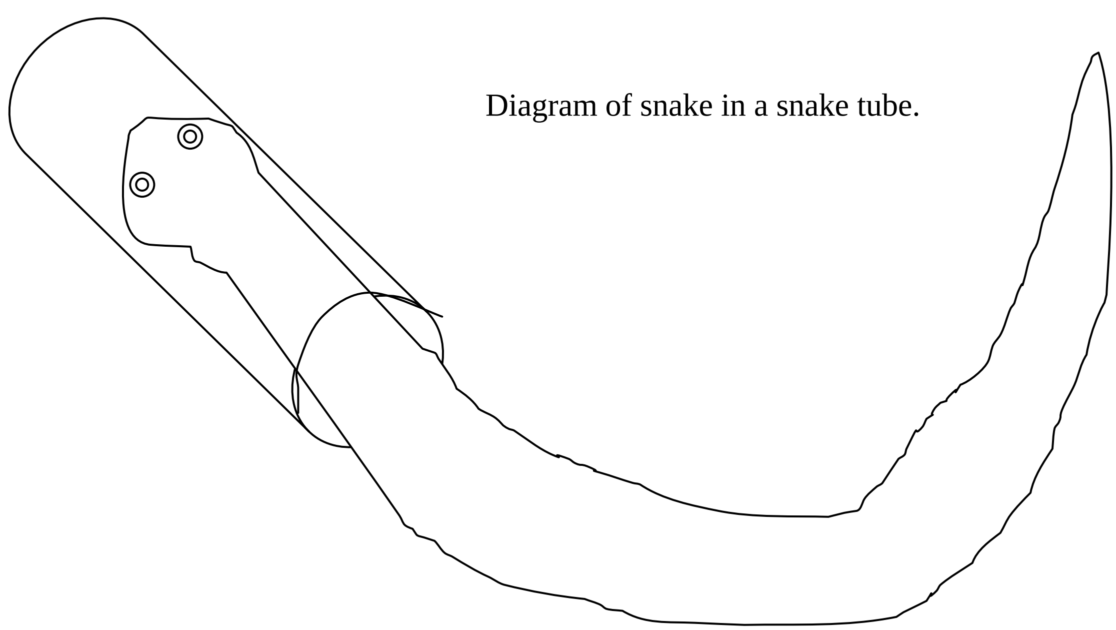


Diagram of snake in a snake tube.

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 the waders 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.

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). 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.

Target amphibian species:

Common Name	Scientific Name	Habitat
Mudpuppy	*Necturus maculosus*	Clean rivers, streams, lakes, reservoirs
Central newt	*Notophthalmus viridescens*	Vegetated woodland ponds, roadside flooded ditches, & adjacent habitat
Smallmouth salamander	*Ambystoma texanum*	Woodland pools, open woods
Blue-spotted salamander	*Ambystoma laterale*	Woodland pools, open woods
Crawfish frog	*Rana areolata*	Prairie marshes, ponds, river floodplains
Cricket frog	*Acris crepitans*	Shallow wetlands & streams
Great plains toad	*Bufo cognatus*	Prairie, nonnative grassland

Target reptile species:

Common Name	Scientific Name	Habitat
Ornate box turtle	*Terrepene ornata*	Sand prairies, savanna
Blanding's turtle	*Emydoidea blandingii*	Shallow, well vegetated wetlands
Wood turtle	*Clemmys insculpta*	Floodplain forest, rivers
Alligator snapping turtle	*Macroclemys temmincki*	Large rivers
Yellow mud turtle	*Kinosternon flavescens*	Shallow, ephemeral pools, adjacent areas with nearly pure sand soils
Common musk turtle	*Sternotherus odoratus*	Backwaters and spring fed ponds adjacent to sandy uplands

Target reptile species, continued:

Common Name	Scientific Name	Habitat
Slender glass lizard	*Ophisaurus attenuatus*	Prairie, pasture, forest edge, savanna
Six-lined racerunner	*Cneimidophorus sexlineatus*	Sand prairies, savanna
Northern prairie skink	*Eumeces septentrionalis*	Sandy prairie-forest edge, wetland edge
Great plains skink	*Eumeces obsoletus*	Rocky prairie, forest edge
Diamondback water snake	*Nerodia rhombifera*	Quiet pools and backwater sloughs
Yellowbelly water snake	*Nerodia erythrogaster flavigaster*	Backwater sloughs and forested wetlands
Copperbelly water snake	*Nerodia erythrogaster neglecta*	Backwater sloughs and forested wetlands
Smooth earth snake	*Virginia valeriae*	Rocky woodland
Western worm snake	*Carphophis amoenus*	Rocky woodland
Smooth green snake	*Opheodrys vernalis*	Old field, savanna, wet prairie, marsh
Prairie kingsnake	*Lampropeltis calligaster*	Woodland edge, open woodland, grassland, savanna
Speckled kingsnake	*Lampropeltis getulus*	Prairie, woodland edge, savanna
Bullsnake	*Pituophis catenifer sayi*	Prairie, deciduous woodland edge, savanna
Western hognose snake	*Heterodon nasicus*	Sand prairie
Eastern massasauga rattlesnake	*Sistrurus catenatus catenatus*	Early successional wetland, upland grassland
Timber rattlesnake	*Crotalus horridus*	Forested areas near rock outcrops, woodland, hill prairie
Prairie rattlesnake	*Crotalus viridis*	Prairie
Copperhead	*Agkistrodon contortrix*	Forested, rocky hillsides

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

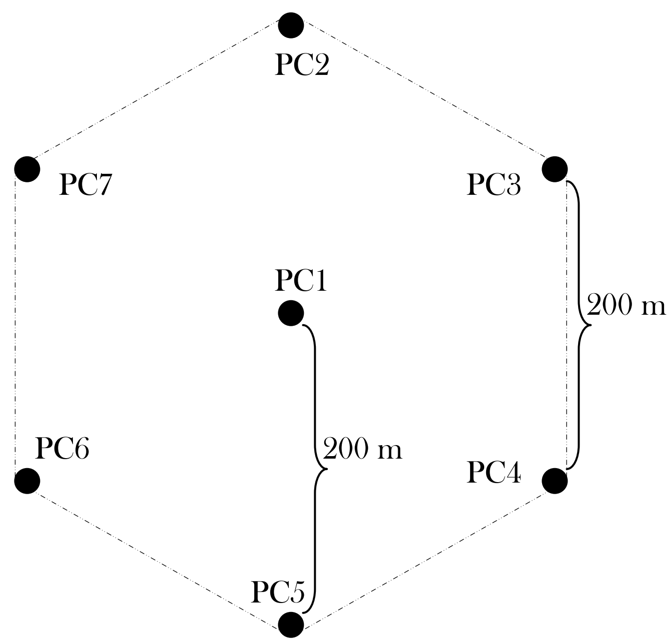
Pitfall traps with (or without) drift fences are time consuming to install. They also only catch species that are not able to jump or crawl out, mostly limiting the use to salamanders, toads, small snakes, and some lizards. They can result in high mortality for small mammals, or herpetofauna if not checked 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.

DATA SHEETS:

Data sheets for this protocol are located in Appendix 2.

Chapter Eleven
Breeding and Migratory Bird Monitoring Protocol

IOWA BIRD MONITORING:
Two primary methods will be used to document birds in Iowa, point counts and nocturnal broadcast surveys. Both of these methods will be implemented in such a way as to be able to be compared to the USFS data, should that program become national. The nocturnal broadcast surveys will cover a larger area which encompasses the hexagonal sampling plot. In addition to the 4 point locations utilized by the USFS (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^{o}, 60^{o}, 120^{o}, 180^{o}, 240^{o}, and 300^{o}, respectively. The point count stations are 200 meters apart. If the station should fall on a dangerous (e.g. cliff) or noisy place (e.g. road), then the station should be moved to the closest available spot with care being taken to keep the station spacing as close to 200 m as possible.

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. 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. Data collection should begin 15 minutes after sunrise and be concluded for the day by 4.5 hours after sunrise. Depending on travel time both within the hexagon between stations and between permanent sampling plot locations, it may or may not be possible for 1 person to cover 2 permanent locations 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 could 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. If possible, each visit should be conducted by a different technician to ensure observer un-bias. During the fall season, however, bird species composition may change quickly; therefore 4 visits per site would be preferred. 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 to 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.

Other data to be collected at every sampling hexagonal plot include the date, cloud cover, wind speed, and start & end times. Tables for sky condition and wind speed are located on the data sheet. 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 such. In addition, birds seen or heard as the technician moves through the sampling plot should be recorded. 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 may not be used in the density estimates.

During the summer season (May and June) 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.

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 could 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 4 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 moonlight (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 tape (or CD) player and if needed, amplified with a megaphone such that calls are 100-110 dB at 1 m in front of the technician holding the speaker or megaphone.

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 tape 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 tape), and bird location.

HABITAT & PLANT COMPOSITION 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. A habitat data collection plot should be established at every bird point count location. See Chapter 19 for information on terrestrial habitat and plant composition measurements, and Chapter 20 for information on aquatic measurements. As the same areas will be searched for all species of greatest conservation need, habitat data collection instructions are included in these chapters. However, all data collection technicians should coordinate with other crews to ensure that all needed habitat data is collected.

EQUIPMENT LIST:

Day point count surveys:
- Binoculars
- Small tape recorder and blank tapes (to record unrecognized bird calls)
- Stopwatch
- Range finder (if observer needs assistance in determining distance)
- Standard field backpack with clipboard, datasheets, pencils, notebook, and field guides
- Bird call tapes to leave in truck for ID help

Nocturnal calling surveys:
- Correct calling tape or CD for that area
- Tape or CD player
- Megaphone
- Batteries
- Headlamps
- 1 million candle watt spotlight
- Compass
- Topographic maps
- Aerial photos (leave in truck)
- Flagging (reflective)
- Stopwatch
- Standard field backpack with clipboard, datasheets, pencils, notebook, and field guides

STAFF & TRAINING:

Point count survey technicians should be hired based upon their ability to already be able to ID birds by call and sight (at least most birds). They can gain experience on the job but should have at least limited prior experience. There should be one person per site per visit and technicians should rotate through sites so no site is visited by the same technician during the same 'season' unless there is no other choice. 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 2-3 weeks of training at the beginning of the season, including field trials and museum visits. Each person will be provided a list of potential sightings and a notebook to record unknowns and details. 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. Most likely the bird technicians will be needed to do both the early point count surveys and the nocturnal callback surveys. Nocturnal callback survey technicians should work in teams of at least 2 for safety concerns as these surveys are conducted after dark (between sunset and midnight). Ideally, one of the 2 people would have prior nocturnal birding experience.

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.

Things that the crew 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. Is the data legible?

Similarly, the nocturnal survey crews should also be 'tested' by a more experienced crew leader periodically throughout the season. 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 crew leader or 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. The point count data can be submitted to the USGS bird database (http://www.mp2-pwrc.usgs.gov/point/Help/Index.cfm).

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.

SAFETY CONSIDERATIONS:
The point count technicians will be working alone and therefore should carry a reliable cell phone or radio, GPS unit, maps, and first aid kit. The crew or section leader 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.

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 or radio, 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.

TARGET SPECIES:
The following list of target species represents the species of greatest conservation concern as chosen by the Steering committee for the Iowa Wildlife Action Plan (Zohrer et al. 2005). 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.

Target breeding bird species:

Common Name	Scientific Name	Habitat
American bittern	*Botaurus lentiginosus*	Wetland
Least bittern	*Ixobrychus exilis*	Wetland
Black-crowned night heron	*Nycticorax nycticorax*	Wetland, wet shrubland
Yellow-crowned night heron	*Nyctanassa violacea*	Wetlands, riparian forest
Trumpeter swan	*Cygnus buccinator*	Wetland
Northern pintail	*Anas acuta*	Wetland, grassland
Canvasback	*Aythya valisineria*	Wetland
Redhead	*Aythya americana*	Wetland
Osprey	*Pandion haliaetus*	Wetland, riparian forest
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

Target breeding bird species continued:

Common Name	Scientific Name	Habitat
Peregrine falcon	*Falco peregrinus*	Riparian forest, deciduous forest
Ruffed grouse	*Bonasa umbellus*	Dense forest, open woodland
Greater prairie chicken	*Tympanuchus cupido*	Grassland
Sharp-tailed grouse	*Tympanuchus phasianellus*	Grassland, shrubland
Northern bobwhite	*Colinus virginianus*	Grassland, shrubland
King rail	*Rallus elegans*	Wetland
Common moorhen	*Gallinula chloropus*	Wetland
Sandhill crane	*Grus canadensis*	Wetland, grassland
Piping plover	*Charadrius melodus*	Wetland
Upland sandpiper	*Bartramia longicauda*	Grassland
American woodcock	*Scolopax minor*	Deciduous forest, open woodland, riparian forest
Wilson's phalarope	*Phalaropus tricolor*	Wetland, grassland
Forster's tern	*Sterna forsteri*	Wetland
Least tern	*Sterna antillarum*	Wetland
Black tern	*Chlidonias niger*	Wetland
Black-billed cuckoo	*Coccyzus erythropthalmus*	Riparian and deciduous forests, open woodland, shrubland
Yellow-billed cuckoo	*Coccyzus americanus*	Deciduous forest, shrubland, open woodland
Barn owl	*Tyto alba*	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
Whip-poor-will	*Caprimulgus vociferus*	Deciduous forest, open woodland
Red-headed woodpecker	*Melanerpes erythrocephalus*	Savanna, open woodland, deciduous forest
Acadian flycatcher	*Empidonax virescens*	Deciduous forest, riparian forest
Willow flycatcher	*Empidonax traillii*	Wet shrubland
Least flycatcher	*Empidonax minimus*	Deciduous forest, open woodland
Brown creeper	*Certhia americana*	Deciduous and riparian forest
Bewick's wren	*Thryomanes bewickii*	Open woodland, shrubland
Sedge wren	*Cistothorus platensis*	Grassland, wetland
Veery	*Catharus fuscescens*	Riparian and deciduous forest
Wood thrush	*Hylocichla mustelina*	Deciduous and riparian forest
Northern mockingbird	*Mimus polyglottos*	Open woodland, savanna, shrubland

Target breeding bird species continued:

Common Name	Scientific Name	Habitat
Loggerhead shrike	*Lanius ludovicianus*	Savanna, shrubland
White-eyed vireo	*Vireo griseus*	Open woodland, shrubland
Bell's vireo	*Vireo bellii*	Shrubland, savanna
Blue-winged warbler	*Vermivora pinus*	Deciduous forest, shrubland
Cerulean warbler	*Dendroica cerulea*	Deciduous forest
Black-and-white warbler	*Mniotilta varia*	Deciduous forest
Prothonotary warbler	*Prothonotaria citrea*	Riparian forest
Worm-eating warbler	*Helmitheros vermivorus*	Deciduous forest
Louisiana waterthrush	*Seiurus motacilla*	Riparian and deciduous forest
Kentucky warbler	*Oporornis formosus*	Deciduous and riparian forest
Hooded warbler	*Wilsonia citrina*	Deciduous forest
Yellow-breasted chat	*Icteria virens*	Open woodland, shrubland
Dickcissel	*Spiza americana*	Grassland
Eastern towhee	*Pipilo erythrophthalmus*	Open woodland, shrubland
Field sparrow	*Spizella pusilla*	Shrubland, grassland
Lark sparrow	*Chondestes grammacus*	Grassland, shrubland, savanna
Grasshopper sparrow	*Ammodramus savannarum*	Grassland
Henslow's sparrow	*Ammodramus henslowii*	Grassland
Bobolink	*Dolichonyx oryzivorus*	Grassland
Eastern meadowlark	*Sturnella magna*	Grassland, savanna

Target migratory bird species:

Common Name	Scientific Name	Habitat
American white pelican	*Pelecanus erythrorhynchos*	Wetland
Yellow rail	*Coturnicops noveboracensis*	Wetland, grassland
Whooping crane	*Grus americana*	Wetland, grassland
American golden-plover	*Pluvialis dominica*	Wetland
American avocet	*Recurvirostra americana*	Wetland
Greater yellowlegs	*Tringa melanoleuca*	Wetland
Lesser yellowlegs	*Tringa flavipes*	Wetland
Solitary sandpiper	*Tringa solitaria*	Wetland
Hudsonian godwit	*Limosa haemastica*	Wetland
Marbled godwit	*Limosa fedoa*	Wetland
Stilt sandpiper	*Micropalama himantopus*	Wetland
Buff-breasted sandpiper	*Tryngites subruficollis*	Wetland, short grassland
Short-billed dowitcher	*Limnodromus griseus*	Wetland
Golden-winged warbler	*Vermivora chrysoptera*	Deciduous forest, open woodland, shrubland

Target migratory bird species continued:

Common Name	Scientific Name	Habitat
Canada warbler	*Wilsonia canadensis*	Deciduous forest
Le Conte's sparrow	*Ammodramus leconteii*	Grassland
Nelson's sharp-tailed sparrow	*Ammodramus nelsonii*	Grassland, wetland
Rusty blackbird	*Euphagus carolinus*	Riparian forest, wetland, wet shrubland

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.

Automated Recordings

Use frog loggers instead of technicians to record bird calls.

Visual Encounter Surveys

Bird species will be recorded while searching for other species. This is an incidental method of data collection and may not be used in analysis, although the species will be included on the species list for the site.

Nest Searching

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

DATA SHEETS:
Data sheets for this protocol are located in Appendix 2.

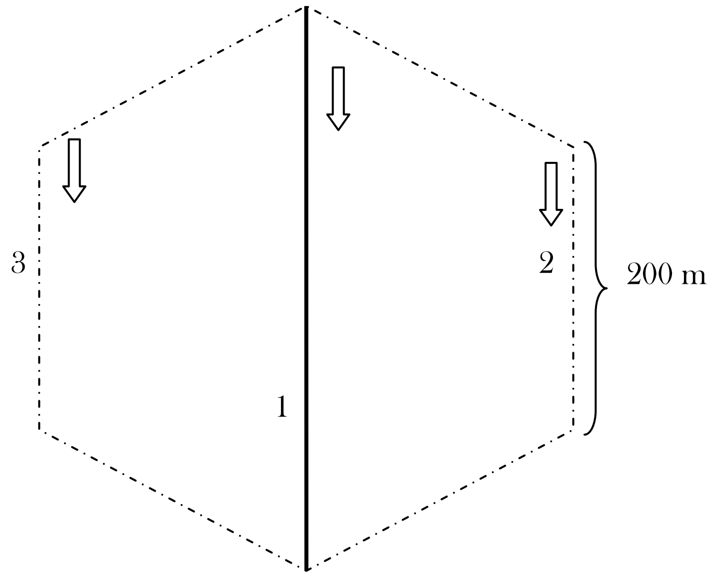
Chapter Twelve
Butterfly Monitoring Protocol

IOWA BUTTERFLY MONITORING:
The primary butterfly survey method 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 transect should be considered the primary transect as it should cross through the designated habitat type. It is critical that the primary transect be surveyed with every visit to the sampling plot by the butterfly crew. Should additional transect distance be necessary then 1 (or 2) other 200-m transects should be walked as well during the same visit. These transects are also at a north-south direction and can be either the east or west side of the primary transect at a spacing of 173.2 m. 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 building 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 primary transect can be divided into habitat sections and is the dividing line of the permanent hexagonal plot. Should a transect cross a 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.

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. This information will be used to compile species lists for the property. The purpose of the additional effort is to document butterflies associated with a property in general, not necessarily the habitat it represents. 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).

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 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. 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 to record the amount of time spent, and always stop the time count when capturing or identifying an individual.

The butterfly season will begin June 1 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. 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^o and 35^o C (70-95^{o}F) with winds less than 16 km/hr ($\tilde{}$10 mph). Most surveys should be conducted on sunny weather days. Sites 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, July, and August. No human activity should occur in the survey area during the morning before the surveys are being conducted.

HABITAT & PLANT COMPOSITION DATA COLLECTION:
See Chapters 19 and 20. This information will be recorded under those protocols. No additional habitat information will be recorded as part of the butterfly monitoring protocol.

EQUIPMENT NEEDED:
Compass
Flagging & tall stakes in certain habitats
Stopwatch
Butterfly forceps
Glassine envelops
Pinning kit
Butterfly net

EQUIPMENT continued:
Hand lens
Field guides
Zip-lock baggies
Digital camera with macro lens
Dissecting scope (left in lab or office)
Standard field kit: Clip board, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets, nail polish or spray paint.

STAFF & TRAINING:
Two weeks of training is recommended and should include 1) field guide use and id, 2) trips to University collections to discuss 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.
- 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 will need to have voucher specimens collected.
- Crew member should be rotated such that each site is visited by more than one observer to reduce the effect of observer bias.
- 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 this technique will be used to compute abundance indices when possible. However, given that different species will have differing detection probabilities, rigorous comparisons of abundance indices between species cannot be made.

SAFETY ISSUES & CONSIDERATIONS:
The butterfly transect technicians will be working alone and therefore should carry a reliable cell phone or radio, GPS unit, maps, and first aid kit. The crew or section leader 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 Iowa Wildlife Action Plan (Zohrer et al. 2005). 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.

Target butterfly species:

Common Name	Scientific Name	Habitat
Pepper and salt skipper	*Amblyscirtes hegon*	Edge of woods & grass waterways
Arogos skipper	*Atrytone arogos*	Prairies & grasslands
Dusted skipper	*Atrytonopsis hianna*	Bluestem grasslands & oldfields
Pipevine swallowtail	*Battus philenor*	Forest, open fields, & roadsides
Swamp metalmark	*Calephelis muticum*	Wet meadows & marshes
Common ringlet	*Coenoympha tullia*	Prairie & marsh edge
Wild indigo duskywing	*Erynnis baptisiae*	Roadsides
Sleepy duskywing	*Errynis brizo*	Oak barrens, sand or shale soils
Dreamy duskywing	*Erynnis icelus*	Woodland or edge
Columbine duskywing	*Erynnis lucilius*	Rocky wooded ravines
Olympia white	*Euchlow olympia*	Open woods, river bluffs, poor soils, & grasslands
Baltimore checkerspot	*Euphydryas phaeton*	Wetlands
Two-spotted skipper	*Euphyes bimacula*	Sedge meadows & marshes
Sedge skipper	*Euphyes dion*	Sedge wetlands
Zebra swallowtail	*Eurytides Marcellus*	Woodland along rivers
Silvery blue	*Glaucopsyche lygdamus*	Open fields & woodland openings
Dakota skipper	*Hesperia dacotae*	Prairie
Leonardus skipper	*Hesperia leonardus*	Open grassy areas
Ottoe skipper	*Hesperia ottoe*	Mid- and tall grass, high-quality prairie
Purplish copper	*Lycaena helloides*	Moist or disturbed areas
Powesheik skipperling	*Oarisma powesheik*	High-quality tallgrass prairie
Mulberry wing	*Poanes massasoit*	Wetland fens
Broad-winged skipper	*Poanes viator*	Wetalnd fens
Zabulon skipper	*Poanes zabulon*	Riparian, oldfield, & woodland edges
Byssus skipper	*Problema byssus*	Tallgrass prairie
Acadian hairstreak	*Satyrium acadica*	Raiparian & oldfield
Hickory hairstreak	*Satyrium caryaevorum*	Forest
Edward's hairstreak	*Satyrium edwardsii*	Woodlands, clearings, & areas of poor soil
Striped hairstreak	*Satyrium liparops*	Forest openings and edges, prairie streamsides
Regal flitillary	*Speyeria idalia*	Prairie & open grassland

ADDITIONAL METHODS FOR SPECIAL LOCATIONS:

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).

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).

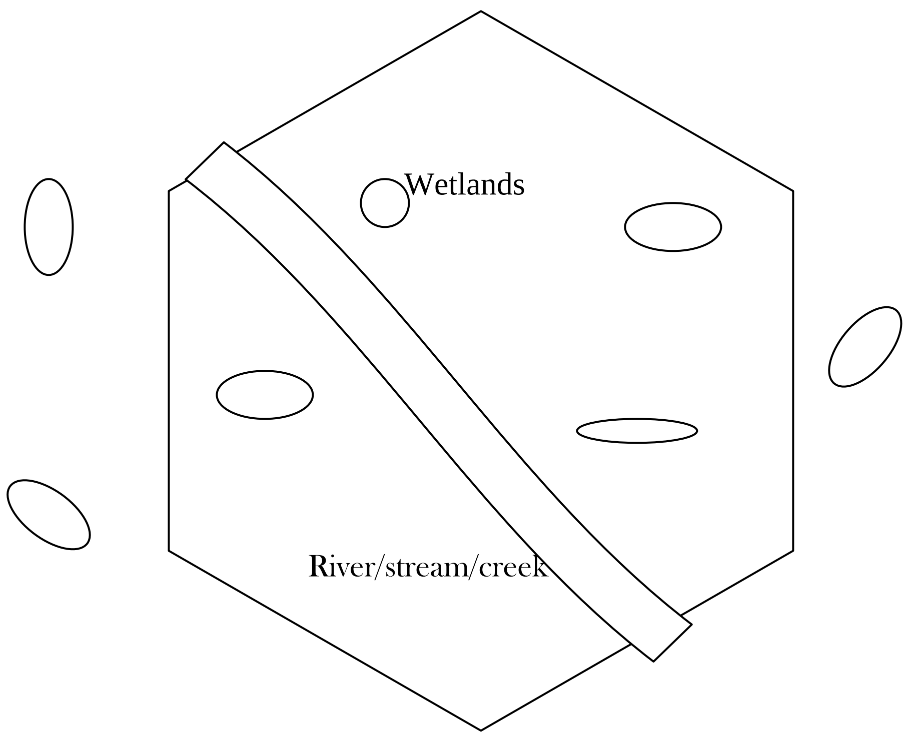
DATA SHEETS:

Data sheets for this protocol are located in Appendix 2.

Chapter Thirteen
Damselfly and Dragonfly Monitoring Protocol

IOWA 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. These exoskeletons can be collected without impacting odonate populations and identified at a later date in the lab. This protocol will search for adult dragonflies and damselflies as well.

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. The entire 26 acre (10 hectare) hexagon will serve as the primary sampling unit. 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.



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 on the property boundaries unless the adjacent landowner has granted permission for use of the adjacent property.

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 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 December 6, 2006) (http://www.iowaodes.com) has time-of-year activity calendars for adult odonates in addition to records of odonates by county. This information should be used as a basis for 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. 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, an abundance class or index should be assigned, following the classification of Chovanec and Waringer (2001). These categories include 1=single individual, 2=rare (for Iowa this would be 2-5 individuals), 3=frequent (6-10 individuals), 4=abundant (11-20 individuals), and 5=extremely abundant (>21 individuals). Remember that this ranking 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.

In addition to the adult abundance indices, 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 VES 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.

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.

HABITAT & PLANT COMPOSITION DATA COLLECTION:
See chapter 20 for information on aquatic habitat and chapter 19 for terrestrial habitat and plant composition measurements. As the same areas will be searched for amphibians, fish, and/or mussels, no additional habitat data is expected to be collected under the dragonfly & damselfly protocol. However, dragonfly and damselfly technicians should coordinate with other crews to ensure that all needed habitat data is collected.

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
Dissecting scope (leave in lab)
Standard field kit: Clip board, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets, nail polish or spray paint.

STAFF & TRAINING:
Two weeks of training is recommended and should include 1) field guide use and id, 2) trips to University museums to discuss 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.
- Crew member should be rotated such that each site is visited by more than one observer to reduce the effect of observer bias.
- 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 trapping day, each observer should review data sheets to ensure all information present. At the end of the week, the field crew leader should review the collected data sheets as well.

DATA ANALYSIS:
The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of area occupied using program PRESENCE or MARK. The data collected with this technique will be used to compute abundance indices

when possible. However, given that different species will have differing detection probabilities, comparisons of abundance indices between species should be interpreted with caution.

SAFETY ISSUES & CONSIDERATIONS:
The odonate technicians may be working alone and therefore should carry a reliable cell phone or radio, GPS unit, maps, and first aid kit. The crew or section leader 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 Iowa Wildlife Action Plan (Zohrer et al. 2005). Distribution maps for these species can be found at www.iowaodes.com. Appendix 1 contains a list of additional, more common, dragonfly and damselfly species which may be encountered during the monitoring efforts.

Target dragonflies & damselflies species:

Common Name	Scientific Name	Habitat
Spangled skimmer	*Libellula cyanea*	Artificial ponds, lakes
Slaty skimmer	*Libellula incesta*	Old river oxbow
Rusty snaketail	*Ophiogomphus rupinsulensis*	Sandy, rocky creeks
Sioux snaketail	*Ophiogomphus smithi*	Sand-bottomed creeks
Mocha emerald	*Somatochlora linearis*	Wooded edges
Brimstone clubtail	*Stylurus intricatus*	Sandy streams
Blue-faced meadowhawk	*Sympetrum ambiguum*	Temporary pools, oxbows
Carolina saddlebags	*Tramea Carolina*	Marsh
Emma's dancer	*Argia emma*	Small streams
Spotted spreadwing	*Lestes congener*	Edge of pools, marsh
Elegant spreadwing	*Lestes inaequalis*	Ponds
Sweetflag spreadwing	*Lestes forcipatus*	Marsh, pond edge
Sulpher-tipped clubtail	*Gomphus militaris*	Artificial ponds, lakes
Rapids clubtail	*Gomphus quadricolor*	Rocky creeks
Canada darner	*Aeshna canadensis*	Marsh, pond edge
Variable darner	*Aeshna interrupta*	Lakes, ponds, streams
Blue-eyed darner	*Aeshna multicolor*	Small lakes, ponds
Green striped darner	*Aeshna verticalis*	Marshes, pond edges
Four-spotted skipper	*Libellula quadrimaculata*	Marshes, wooded ponds
Royal river cruiser	*Macromia taeniolata*	Lakes, rivers
Cyrano darner	*Nasiaeschno pentacantha*	Shaded creeks, lakes, oxbows
Smoky shadowdragon	*Neurocordulia molesta*	Large rivers
Stygian shadowdragon	*Neurocordulia yamaskanensis*	Mississippi River
Paiute dancer	*Argia Alberta*	Small streams, road ditches
Prairie bluet	*Coenagrion angulatum*	Lakes, ponds
Boreal bluet	*Enallagma boreale*	Marsh
Alkali bluet	*Enallagma clausum*	Pond edges without vegetation
Vesper bluet	*Enallagma vesperum*	Deep lakes, ponds

ADDITIONAL METHODS FOR SPECIAL LOCATIONS:

It may be necessary to collect a voucher specimen of adult odonates for later identification or proof of identification. Before doing this, be sure that you have written permission from the DNR. 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 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).

DATA SHEETS:

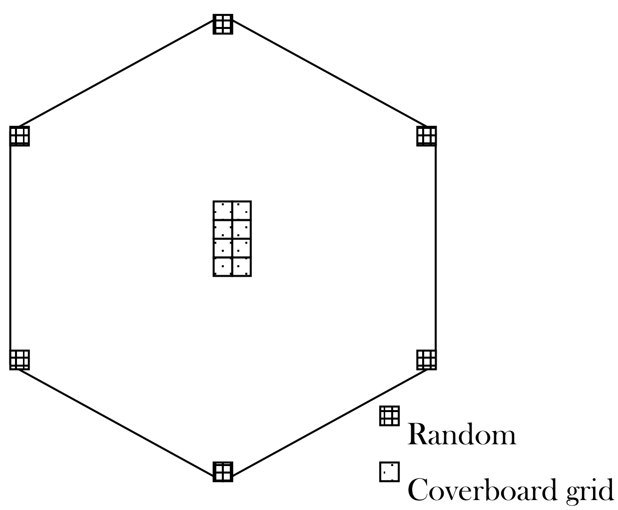
Data sheets for this protocol are located in Appendix 2.

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.

IOWA SNAIL MONITORING:
While the 9 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 illustrated below. Each block is 2 x 2 inches (5 x 5 cm) in size. These individual coverboards are important in two respects. The first is in monitoring additional habitats associated with a sampling area, provided that the hexagonal points fall in habitats other than that used for the site classification. The second reason for using the additional coverboards is for the assessment of spatial aggregation (Henry et al. 2003). Should snail populations be declining, it is conceivable that this could manifest in an increased aggregation of the remaining individuals in suitable habitat (Henry et al. 2003).

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

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 on the next page. 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.

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.

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
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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

HABITAT & PLANT COMPOSITION 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. See chapter 19 for information on terrestrial habitat and plant composition measurements and chapter 20 for habitat data collection in aquatic areas. As the same areas will be searched for birds and mammals, no additional habitat data is expected to be collected under the terrestrial snail protocol. However, terrestrial snail technicians should coordinate with other crews to ensure that all needed habitat data is collected.

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, 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 id, 2) trips to University museums to discuss defining species characteristics, 3) field practice with an experienced observer, and 4) proficiency testing. Crews will also need training on habitat data collection.

DATA QUALITY & MANAGEMENT:
Snail species may be 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 will 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

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:

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 Pleistocence 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.

SUGGESTED FIELD GUIDES:
Leonard, AB. 1959. Handbook of Gastropods in Kansas. The State Printing Plant. Topeka, Kansas.

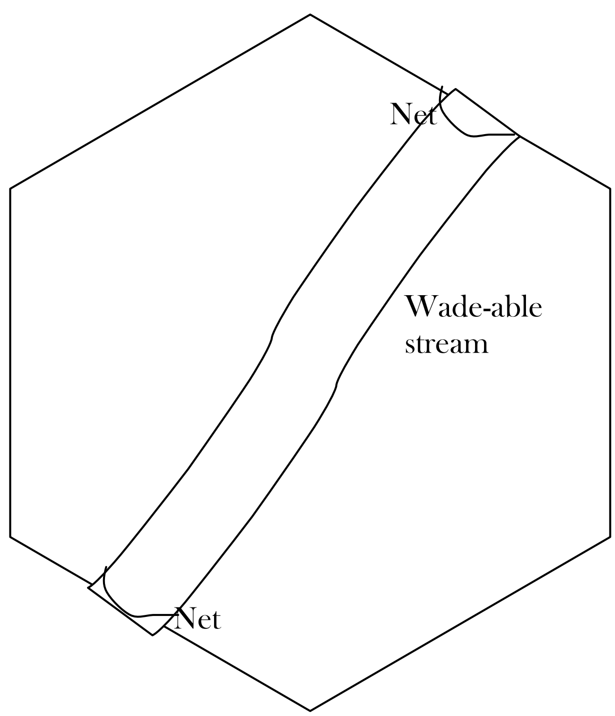
DATA SHEETS:
Data sheets for this protocol are located in Appendix 2.

Chapter Fifteen
Fish Monitoring
Wadeable Streams & Rivers

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

IOWA 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.

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 may 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 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 IDNR 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 IDNR 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 $\leq$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, trails, 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 samples.
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 a 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 IDNR 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 INDR (2001). Appendix 4 is copied verbatim from the INDR (2001) sampling protocol (pages 6-14) please reference this for the macroinvertebrate sampling.

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 Appendix 5.

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

HABITAT AND PLANT COMPOSITION DATA COLLECTION:
It is expected that the Aquatic Habitat Monitoring Protocol (Chapter 20) will acquire all necessary habitat data. That chapter includes information on collecting data on the habitats stratified into a wetland classification (i.e. river, stream, creek, impoundment, lake, etc.), as well as wetlands which occur on sites classified as terrestrial habitats. As the same areas will be searched for multiple species, no additional habitat data is expected to be collected under the fish in wadeable streams protocol. However, fisheries technicians should coordinate with other crews to ensure that all needed habitat data is collected.

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, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets.
Field guides
Rubber 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:
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, 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 IDNR (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 hot water and bleach.

TARGET SPECIES:
The following list of fish species represents the 67 species of greatest conservation need as chosen by the Steering committee for the Iowa Wildlife Action Plan (Zohrer et al. 2005) 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.

Target species:

Common Name	Scientific Name	Habitat
Chestnut lamprey	*Ichthyomyzon castaneus*	Mississippi and Chariton rivers
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
Bowfin	*Amia calva*	Mississippi River
Longnose gar	*Lepisosteus osseus*	Mississippi and Missouri Rivers & larger tributaries
American eel	*Anguilla rostrata*	Mississippi and Missouri Rivers & larger tributaries
Skipjack herring	*Alosa chrysochloris*	Mississippi and Missouri Rivers
Mooneye	*Hiodon tergisus*	Larger interior rivers statewide
Goldeye	*Hiodon alosoides*	Missouri River & large streams in W, S, and SE
Brook trout	*Salvelinus fontinalis*	NE corner
Grass pickerel	*Esox americanus*	Missouri River & tributaries
Central mudminnow	*Umbra limi*	N 1/3
Largescale stoneroller	*Campostoma oligolepsis*	NE 2/3
Western silvery minnow	*Hybognathus agryritis*	Missouri drainage
Mississippi silvery minnow	*Hybognathus nuchalis*	Mississippi drainage
Plains minnow	*Hybognathus placitus*	Missouri drainage
Speckled chub	*Macrhybopsis aestivalis*	Large interior rivers statewide
Flathead chub	*Platygobio gracillis*	Missouri drainage
Sicklefin chub	*Macrybopsis meeki*	Missouri River
Silver chub	*Macrybopsis storeriana*	Larger interior rivers statewide
Gravel chub	*Erimytax x-punctatus*	Central & NE
Pallid shiner	*Hybopsis amnis*	Upper Mississippi River
Pugnose minnow	*Opsopoeodus emiliae*	Mississippi River
Pugnose shiner	*Notropis anogenus*	West Lake Okojobi
River shiner	*Notropis blennius*	Mississippi and Missouri Rivers & larger tributaries
Ghost shiner	*Notropis buchanani*	Mississippi River
Blacknose shiner	*Notropis heterolepis*	NW
Spottail shiner	*Notropis hudsonius*	Natural lakes, Mississippi River
Ozark minnow	*Notropis nubilus*	NE ¼
Weed shiner	*Notropis texanus*	Cedar & Mississippi Rivers
Topeka shiner	*Notropis Topeka*	W ¾
Channel mimic shiner	*Notropis volucellus*	Upper Mississippi River

Target species continued:

Common Name	Scientific Name	Habitat
Longnose dace	*Rhinichthys cataractae*	NE corner
Pearl dace	*Margariscus margarita*	Worth county
Blue sucker	*Cycleptus elongates*	MS and MO Rivers & larger tributaries
Black buffalo	*Ictiobus niger*	Mississippi River & large tributaries
Black redhorse	*Moxostoma duquesnei*	Turkey & upper Iowa river drainages
Golden redhorse	*Moxostoma erythrurum*	Small & medium streams statewide
River redhorse	*Moxostoma carinatum*	Upper pools of Mississippi
Greater redhorse	*Moxostoma valenciennesi*	Upper Mississippi River
Spotted sucker	*Minytrema melanops*	Mississippi River
Brown bullhead	*Ameiurus nebulosus*	N 1/3
Slender madtom	*Noturus exilis*	Mississippi River tributaries
Tadpole madtom	*Noturus gyrinus*	Statewide
Freckled madtom	*Noturus gyrinus*	Mississippi River & large tributaries
Pirate perch	*Aphredoderus sayanus*	Mississippi River & large tributaries
Trout perch	*Percopsis omiscomycus*	NW ¼; Upper Mississippi River, Grand & Chariton Rivers
Burbot	*Lota lota*	MO River, MS River & tributaries
Banded killifish	*Fundulus diaphanous*	Natural lakes in NW; Missouri River
Blackstripe topminnow	*Fundulus notatus*	E 1/3
Mottled sulpin	*Cottus bairdi*	Lower Bear Creek
Slimy sculpin	*Cottus cognatus*	NE corner
Warmouth	*Lepomis gulosus*	S ½; Mississippi River
Pumpkinseed	*Lepomis gibbosus*	Mississippi River & natural lakes
Slenderhead darter	*Percina phoxocephala*	Mississippi drainage
Blackside darter	*Percina maculate*	Mississippi River
River darter	*Percina shumardi*	Mississippi River
Northern logperch	*Percina caprodes*	Mississippi drainage, Clear Lake
Crystal darter	*Crystallaria asprella*	Mississippi & Turkey Rivers
Western sand darter	*Annicrypta clara*	Mississippi River
Banded darter	*Etheostoma zonale*	NE ¼
Mud darter	*Etheostoma asprigene*	Mississippi River & tributaries
Orangethroat darter	*Etheostoma spectabile*	SE ¼
Least darter	*Etheostoma microperca*	Maquiketa, tributary to Otter Creek

ADDITIONAL METHODS FOR SPECIAL LOCATIONS:

<u>Minnow Traps</u>

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.

DATA SHEETS:

Data sheets for this protocol are located in Appendix 2.

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.

IOWA 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 Wadeable 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.

<u>Electrofishing</u>

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 Appendix 5.

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.

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.

HABITAT AND PLANT COMPOSITION 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.

See Chapter 20 for information on aquatic habitat measurements. As the same areas will be searched for amphibians, fish, dragonflies & damselflies, and/or mussels, no additional habitat data is expected to be collected under the fish in lakes protocol. However, fisheries technicians should coordinate with other crews to ensure that all needed habitat data is collected.

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

STAFF & TRAINING:
Two weeks of training (beginning on August 15) is recommended and should include 1) field guide use and id, 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 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, 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:

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

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

SAFETY 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 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 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.

TARGET SPECIES:
The following list of fish species represents the 67 species of greatest conservation need as chosen by the Steering committee for the Iowa Wildlife Action Plan (Zohrer et al. 2005). These animals are those that may be potentially encountered along an aquatic environment. 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 additional, more common, species which may also be encountered during the monitoring efforts.

Target species:

Common Name	Scientific Name	Habitat
Chestnut lamprey	*Ichthyomyzon castaneus*	Mississippi and Chariton rivers
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
Bowfin	*Amia calva*	Mississippi River
Longnose gar	*Lepisosteus osseus*	Mississippi and Missouri Rivers & larger tributaries
American eel	*Anguilla rostrata*	Mississippi and Missouri Rivers & larger tributaries
Skipjack herring	*Alosa chrysochloris*	Mississippi and Missouri Rivers
Mooneye	*Hiodon tergisus*	Larger interior rivers statewide
Goldeye	*Hiodon alosoides*	Missouri River & large streams in W, S, and SE
Brook trout	*Salvelinus fontinalis*	NE corner
Grass pickerel	*Esox americanus*	Missouri River & tributaries
Central mudminnow	*Umbra limi*	N 1/3
Largescale stoneroller	*Campostoma oligolepsis*	NE 2/3
Western silvery minnow	*Hybognathus agryritis*	Missouri drainage
Mississippi silvery minnow	*Hybognathus nuchalis*	Mississippi drainage
Plains minnow	*Hybognathus placitus*	Missouri drainage
Speckled chub	*Macrhybopsis aestivalis*	Large interior rivers statewide
Flathead chub	*Platygobio gracillis*	Missouri drainage
Sicklefin chub	*Macrybopsis meeki*	Missouri River
Silver chub	*Macrybopsis storeriana*	Larger interior rivers statewide

Target species continued:

Common Name	Scientific Name	Habitat
Gravel chub	*Erimytax x-punctatus*	Central & NE
Pallid shiner	*Hybopsis amnis*	Upper Mississippi River
Pugnose minnow	*Opsopoeodus emiliae*	Mississippi River
Pugnose shiner	*Notropis anogenus*	West Lake Okojobi
River shiner	*Notropis blennius*	Mississippi and Missouri Rivers & larger tributaries
Ghost shiner	*Notropis buchanani*	Mississippi River
Blacknose shiner	*Notropis heterolepis*	NW
Spottail shiner	*Notropis hudsonicus*	Natural lakes, Mississippi River
Ozark minnow	*Notropis nubilus*	NE ¼
Weed shiner	*Notropis texanus*	Cedar & Mississippi Rivers
Topeka shiner	*Notropis Topeka*	W ¾
Channel mimic shiner	*Notropis volucellus*	Upper Mississippi River
Longnose dace	*Rhinichthys cataractae*	NE corner
Pearl dace	*Margariscus margarita*	Worth county
Blue sucker	*Cycleptus elongates*	Mississippi and Missouri Rivers & larger tributaries
Black buffalo	*Ictiobus niger*	Mississippi River & large tributaries
Black redhorse	*Moxostoma duquesnei*	Turkey & upper Iowa river drainages
Golden redhorse	*Moxostoma erythrurum*	Small & medium streams statewide
River redhorse	*Moxostoma carinatum*	Upper pools of Mississippi
Greater redhorse	*Moxostoma valenciennesi*	Upper Mississippi River
Spotted sucker	*Minytrema melanops*	Mississippi River
Brown bullhead	*Ameiurus nebulosus*	N 1/3
Slender madtom	*Noturus exilis*	Mississippi River tributaries
Tadpole madtom	*Noturus gyrinus*	Statewide
Freckled madtom	*Noturus gyrinus*	Mississippi River & large tributaries
Pirate perch	*Aphredoderus sayanus*	Mississippi River & large tributaries
Trout perch	*Percopsis omiscomycus*	NW ¼; Upper Mississippi River, Grand & Chariton Rivers
Burbot	*Lota lota*	Missouri River, Mississippi River & tributaries
Banded killifish	*Fundulus diaphanous*	Natural lakes in NW; Missouri River
Blackstripe topminnow	*Fundulus notatus*	E 1/3
Mottled sulpin	*Cottus bairdi*	Lower Bear Creek
Slimy sculpin	*Cottus cognatus*	NE corner
Warmouth	*Lepomis gulosus*	S ½; Mississippi River
Pumpkinseed	*Lepomis gibbosus*	Mississippi River & natural lakes
Slenderhead darter	*Percina phoxocephala*	Mississippi drainage
Blackside darter	*Percina maculate*	Mississippi River
River darter	*Percina shumardi*	Mississippi River
Northern logperch	*Percina caprodes*	Mississippi drainage, Clear Lake

Target species continued:

Common Name	Scientific Name	Habitat
Crystal darter	*Crystallaria asprella*	Mississippi & Turkey Rivers
Western sand darter	*Annicrypta clara*	Mississippi River
Banded darter	*Etheostoma zonale*	NE ¼
Mud darter	*Etheostoma asprigene*	Mississippi River & tributaries
Orangethroat darter	*Etheostoma spectabile*	SE ¼
Least darter	*Etheostoma microperca*	Maquiketa, tributary to Otter Creek

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 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.

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 or species.

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.

DATA SHEETS:

Data sheets for this protocol are located in Appendix 2.

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 (Gutreuter et al. 1995), and the Great River Ecosystems Field Operations Manual, Environmental Monitoring and Assessment Program (Angradi et al. DRAFT 2005). The reader should refer to both of the above documents for more in depth information.

IOWA 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.

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 LRTM 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 a fisheries staff person. 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 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 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.

Seining

Seining may be the most efficient method to sample for small fish species (such as *Etheostoma* and *Notropis* species). This involves pulling a seine through the water. The seine should be of 1/8" or 3/16" mesh size, and have floats attached at the top and weights attached at the bottom. For most wade-able streams and rivers in Iowa a haul seine should be sufficient. If not performed correctly, fish could escape from under the net.

Along the shoreline parallel to the one side of the hexagon, 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 25 or 50 meters. The 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 for a limited amount of time until processing. As close to 200 m as possible should be covered with this method (Quist et al. 2003).

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 the appendix. 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.

In some situations it may be necessary to collect tissue for age-growth calculations. This will most likely 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 envelops on which the following information is recorded: site name, sampling gear used, date of sampling, species, length, weight, and any comments. At the end of each day scale envelops should be spread out to allow to dry completely. This is especially important for spines which can go rancid quickly if not allowed to dry.

<u>Water Sample Collection</u>

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.

<u>Benthic Macroinvertebrate Sampling</u>

This data is qualitative and semi-quantitative, providing a list of macroinvertebrate species as well as an abundance index to the taxon seen. These techniques will not allow density or biomass estimates to be made. 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, the modified-Hess sampler, the Surber sampler, or the 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 IDNR 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 INDR (2001). Appendix 4 is copied verbatim from the INDR (2001) sampling protocol (pages 6-14). Please reference this for the macroinvertebrate sampling.

HABITAT AND PLANT COMPOSITION DATA COLLECTION:

It is expected that the Aquatic Habitat Monitoring Protocol (Chapter 20) will acquire all necessary data for fish. In depth details concerning the aquatic data acquisition can be found in Chapter 20. 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) in the sampling plot which were surveyed would also need to have aquatic habitat characteristics measured. All field crews need to coordinate with each other to ensure that all needed habitat data is collected at each site.

EQUIPMENT NEEDED:

Water collection jars
Dip nets
Trawling equipment

EQUIPMENT continued:
Electroshocking boat and associated equipment
Inflatable life preserver
Plastic calipers or measuring board
Standard field kit: Clip board, 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
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:
Two weeks of training is recommended and should include 1) field guide use and id, 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 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. 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 area occupied and detection probabilities using program PRESENCE or program MARK. For additional information on the PAO techniques, see Chapter 5 (Data Analysis).

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

1. Species composition

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

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

SAFETY 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 or radio, 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 hot water and bleach.

TARGET SPECIES:
The following list of fish species represents the 67 species of greatest conservation need as chosen by the Steering committee for the Iowa Wildlife Action Plan (Zohrer et al. 2005). These animals are those that may be potentially encountered along an aquatic environment. 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 additional, more common, species which may also be encountered during the monitoring efforts.

Target species:

Common Name	Scientific Name	Habitat
Chestnut lamprey	*Ichthyomyzon castaneus*	Mississippi and Chariton rivers
Silver lamprey	*Ichthyomyzon unicuspis*	Mississippi River
American brook lamprey	*Lampetra appendix*	Northeast ¼
Lake sturgeon	*Acipenser fulvescens*	Mississippi River
Pallid sturgeon	*Scaphirhynchus albus*	Missouri River
Shovelnose sturgeon	*Scaphirhynchus platorynchus*	Mississippi and Missouri Rivers

Target species continued:

Common Name	Scientific Name	Habitat
Paddlefish	*Polydon spathula*	Mississippi, Missouri, Des Moines, Iowa, Cedar, and Skunk rivers
Bowfin	*Amia calva*	Mississippi River
Longnose gar	*Lepisosteus osseus*	Mississippi and Missouri Rivers & larger tributaries
American eel	*Anguilla rostrata*	Mississippi and Missouri Rivers & larger tributaries
Skipjack herring	*Alosa chrysochloris*	Mississippi and Missouri Rivers
Mooneye	*Hiodon tergisus*	Larger interior rivers statewide
Goldeye	*Hiodon alosoides*	Missouri River & large streams in W, S, and SE
Brook trout	*Salvelinus fontinalis*	NE corner
Grass pickerel	*Esox americanus*	Missouri River & tributaries
Central mudminnow	*Umbra limi*	N 1/3
Largescale stoneroller	*Campostoma oligolepsis*	NE 2/3
Western silvery minnow	*Hybognathus agryritis*	Missouri drainage
Mississippi silvery minnow	*Hybognathus nuchalis*	Mississippi drainage
Plains minnow	*Hybognathus placitus*	Missouri drainage
Speckled chub	*Macrhybopsis aestivalis*	Large interior rivers statewide
Flathead chub	*Platygobio gracillis*	Missouri drainage
Sicklefin chub	*Macrybopsis meeki*	Missouri River
Silver chub	*Macrybopsis storeriana*	Larger interior rivers statewide
Gravel chub	*Erimytax x-punctatus*	Central & NE
Pallid shiner	*Hybopsis amnis*	Upper Mississippi River
Pugnose minnow	*Opsopoeodus emiliae*	Mississippi River
Pugnose shiner	*Notropis anogenus*	West Lake Okojobi
River shiner	*Notropis blennius*	Mississippi and Missouri Rivers & larger tributaries
Ghost shiner	*Notropis buchanani*	Mississippi River
Blacknose shiner	*Notropis heterolepis*	NW
Spottail shiner	*Notropis hudsonius*	Natural lakes, Mississippi River
Ozark minnow	*Notropis nubilus*	NE ¼
Weed shiner	*Notropis texanus*	Cedar & Mississippi Rivers
Topeka shiner	*Notropis Topeka*	W ¾
Channel mimic shiner	*Notropis volucellus*	Upper Mississippi River
Longnose dace	*Rhinichthys cataractae*	NE corner
Pearl dace	*Margariscus margarita*	Worth county
Blue sucker	*Cycleptus elongates*	Mississippi and Missouri Rivers & larger tributaries
Black buffalo	*Ictiobus niger*	Mississippi River & large tributaries
Black redhorse	*Moxostoma duquesnei*	Turkey & upper Iowa river drainages
Golden redhorse	*Moxostoma erythrurum*	Small & medium streams statewide
River redhorse	*Moxostoma carinatum*	Upper pools of Mississippi

Target species continued:

Common Name	Scientific Name	Habitat
Greater redhorse	*Moxostoma valenciennesi*	Upper Mississippi River
Spotted sucker	*Minytrema melanops*	Mississippi River
Brown bullhead	*Ameiurus nebulosus*	N 1/3
Slender madtom	*Noturus exilis*	Mississippi River tributaries
Tadpole madtom	*Noturus gyrinus*	Statewide
Freckled madtom	*Noturus gyrinus*	Mississippi River & large tributaries
Pirate perch	*Aphredoderus sayanus*	Mississippi River & large tributaries
Trout perch	*Percopsis omiscomycus*	NW ¼; Upper Mississippi River, Grand & Chariton Rivers
Burbot	*Lota lota*	Missouri River, Mississippi River & tributaries
Banded killifish	*Fundulus diaphanous*	Natural lakes in NW; Missouri River
Blackstripe topminnow	*Fundulus notatus*	E 1/3
Mottled sulpin	*Cottus bairdi*	Lower Bear Creek
Slimy sculpin	*Cottus cognatus*	NE corner
Warmouth	*Lepomis gulosus*	S ½; Mississippi River
Pumpkinseed	*Lepomis gibbosus*	Mississippi River & natural lakes
Slenderhead darter	*Percina phoxocephala*	Mississippi drainage
Blackside darter	*Percina maculate*	Mississippi River
River darter	*Percina shumardi*	Mississippi River
Northern logperch	*Percina caprodes*	Mississippi drainage, Clear Lake
Crystal darter	*Crystallaria asprella*	Mississippi & Turkey Rivers
Western sand darter	*Annicrypta clara*	Mississippi River
Banded darter	*Etheostoma zonale*	NE ¼
Mud darter	*Etheostoma asprigene*	Mississippi River & tributaries
Orangethroat darter	*Etheostoma spectabile*	SE ¼
Least darter	*Etheostoma microperca*	Maquiketa, tributary to Otter Creek

ADDITIONAL METHODS FOR SPECIAL LOCATIONS:

Minnow Traps

Minnow traps may be an effective way to find additional small fish. These are used as part of the amphibian protocol to capture tadpoles. Minnow traps should be deployed in water at least deep enough to cover the entrance to the trap opening but with an empty plastic bottle inside in order to keep part of the trap above the water line to allow non-gilled captures to breathe. Traps should be checked daily and left in the water for 3 to 5 days.

Fyke Nets

Fyke nets are passive and catch fish by entrapment. Fyke nets tend to be selective for cover seeking, mobile species (Neilson 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. Fyke nets are usually used in shoreline habitats where the water is about 4 feet (1.22 m) deep at the frame. Sampling sites should be georeferenced and mapped to ensure the same areas are returned to with each visit.

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

Night Time Electrofishing

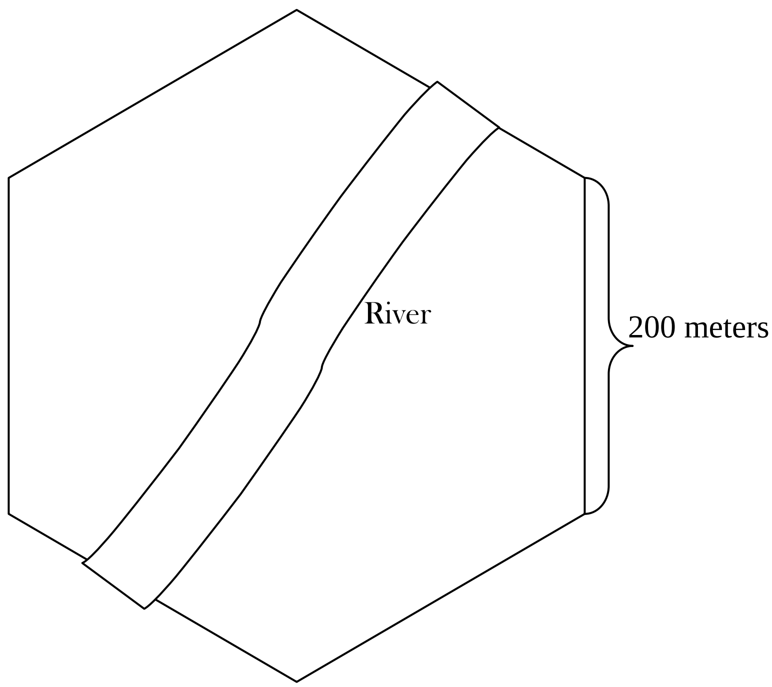
Can be used at the discretion of a biologist and will only be done with supervision from the fisheries biologist for that area. This may increase the species list for the area but can be extremely dangerous. The same technique as daytime electrofishing is followed, only after dark.

DATA SHEETS:

Data sheets for this protocol are located in Appendix 2.

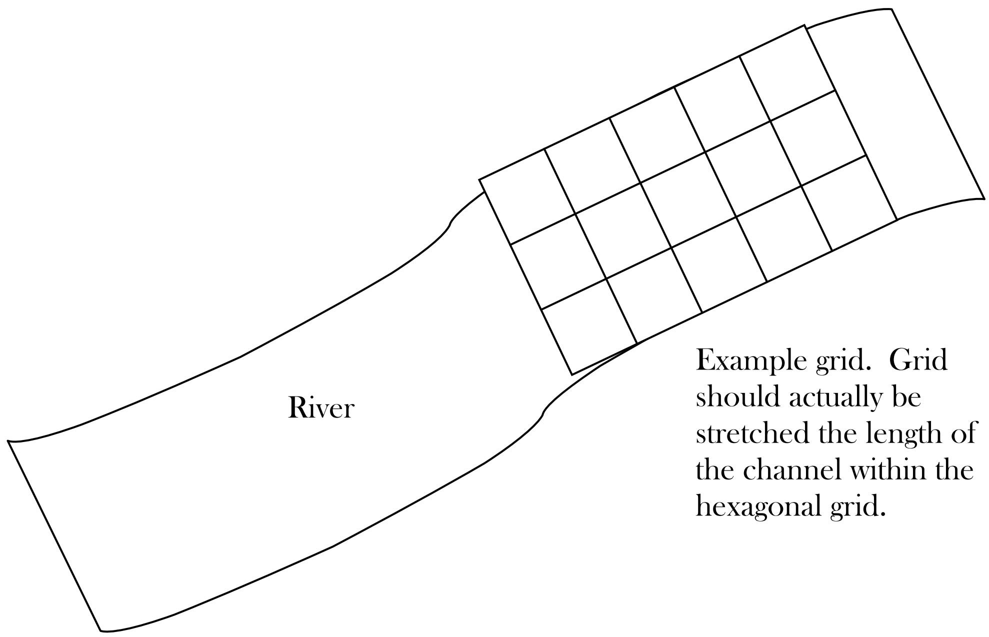
Chapter Eighteen
Mussel Monitoring Protocol

IOWA 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 direction. The other direction should be perpendicular to the stream bank such that it crosses the channel. It may be best to establish this grid by fully extending a surveyors tape along the stream side as a reference guide. GPS the location of the starting end of the grid, along with the starting points of the longitudinal transects. This grid should be established in both deep waters requiring scuba diving (>3 feet deep) and wade able waters < 3 feet deep. The only difference will be that it will be more difficult to establish the grid spacing in a major river (like the Mississippi or Missouri). The grid may have to be placed based on GPS in these systems. Established quadrats will be 1 m^2.



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.

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 <u>Z</u>) randomly choose 4, 12, and 25 as starting locations, then the following table would illustrate the quadrats each would search given the above length and width measurements.

↓ *Riverwidth* → *Riverlength*

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

↓ *Riverwidth* → *Riverlength*

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

↓ *Riverwidth* → *Riverlength*

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

↓ *Riverwidth* → *Riverlength*

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

Each technician should randomly choose a starting quadrat within the first 5 m from the starting downstream location. The technician may not always move in a straight line, depending upon how straight the channel bed is. As another example, a stretch that is *an average* of 4 m wide and 50 m in length, for 25 quadrats and 3 technicians, (and assuming the 3 technicians (*X*, **Y**, and Z) again randomly choose *2*, **12**, and 15 as starting locations, would result in every 24^{th} 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 yellow 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. If necessary, use a sieve to sort through the substrate to search for mussels.

In water over 3 feet deep, it will 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.

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.

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 >20 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.

HABITAT AND PLANT SPECIES COMPOSITION DATA COLLECTION:

It is expected that the Aquatic Habitat Monitoring Protocol (Chapter 20) will acquire all necessary data in the area searched for mussels. Any additional water body (i.e. creeks, streams, ponds, etc) in the sampling plot which was surveyed for mussels would also need to have aquatic habitat characteristics measured. These measurements should be collected as outlined in Chapter 20.

Included in these measurements are data that can be determined from GIS coverages in the lab prior to field work (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. These parameters will still need to be ground-truthed in the field.

EQUIPMENT LIST:
Plastic calipers
Chest waders
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
SCUBA equipment for water > 3 feet deep and certified divers
Standard field kit: Clip board, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & write-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. Two weeks of training (beginning in mid-July) is recommended and should include 1) field guide use and id, 2) trips to University museums to discuss defining species characteristics, 3) field practice with an experienced observer, 4) proficiency testing, and 5) habitat data collection.

SCUBA divers will 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. It may be that the IDNR will need to contract these surveys to the USGS or a consulting firm (e.g. Ecological Specialist Consultants from Missouri).

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 should 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.

Once the first year of data collection is finished, the 1 m^2 size of the quadrat should be reconsidered. It may be that this size should be increased or decreased depending upon the density of mussels found in Iowa waters.

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 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 hot water and bleach (Miller and Payne 1998).

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
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
Fingernail clams	*Musculium sp.* *Pisidium sp.* *Sphaerium sp.*	

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.

DATA SHEETS:
Data sheets for this protocol are located in Appendix 2.

Chapter Nineteen
Terrestrial Plant Species and Habitat Classification Monitoring Protocol

The principal motivation for collecting information on habitat and vegetative characteristics is to monitor potential habitat changes over time. 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.

IOWA HABITAT CLASSIFICATION AND MONITORING:
Within each of the permanent hexagonal sampling sites, 4 plots will 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 will 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.

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

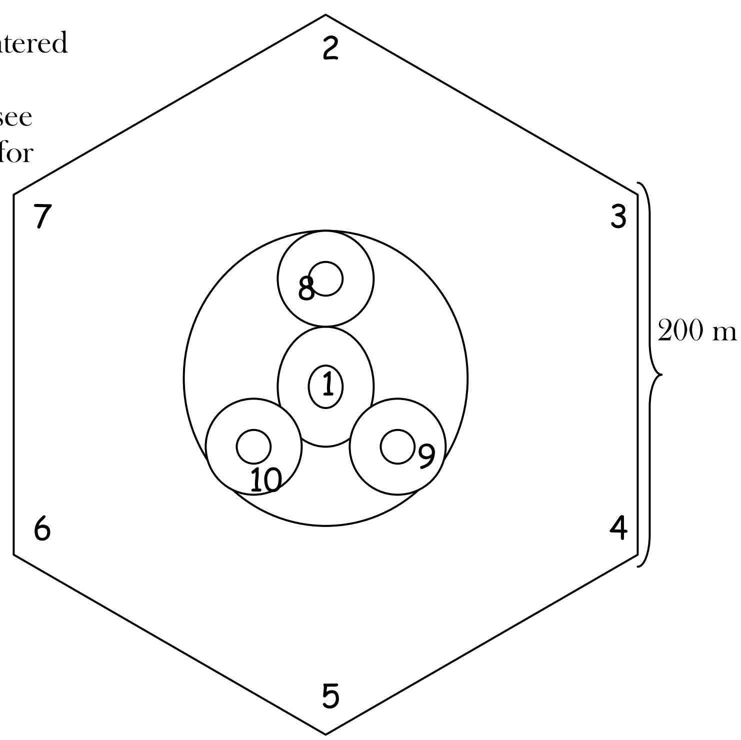
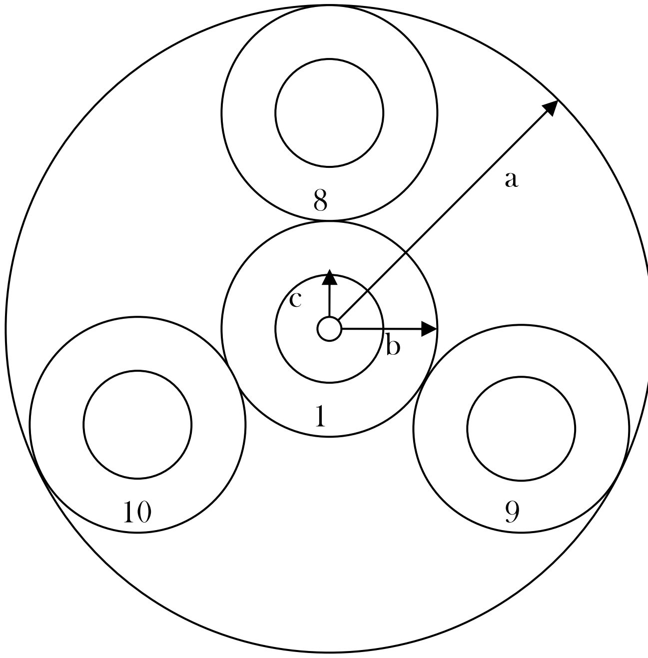


Diagram of nested subplots centered around the center of the hexagonal sampling plot.

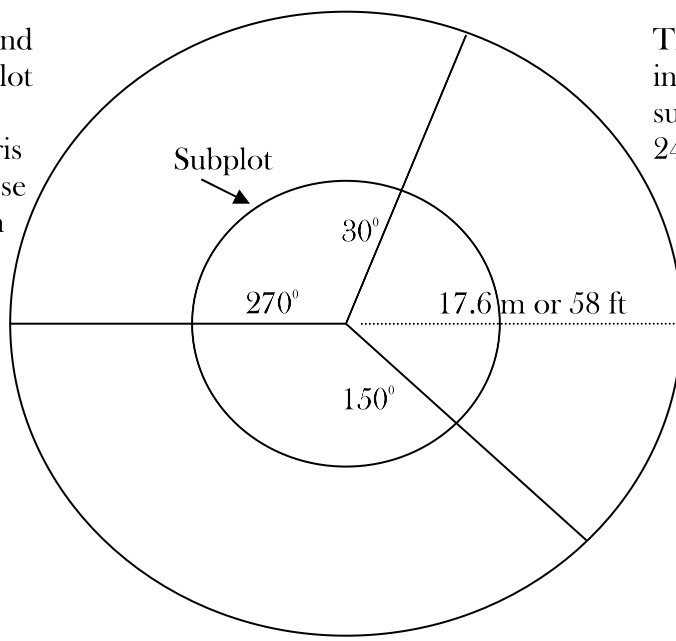
Distances:
a = 56.4 m
b = 17.6 m
c = 7.3 m



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

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).

Q1, Q2, & Q3 are located 4.572 m (or 15 ft) from the center of each subplot at the angles indicated. Q1, Q2, & Q3 are all 1 m^2 quadrats.

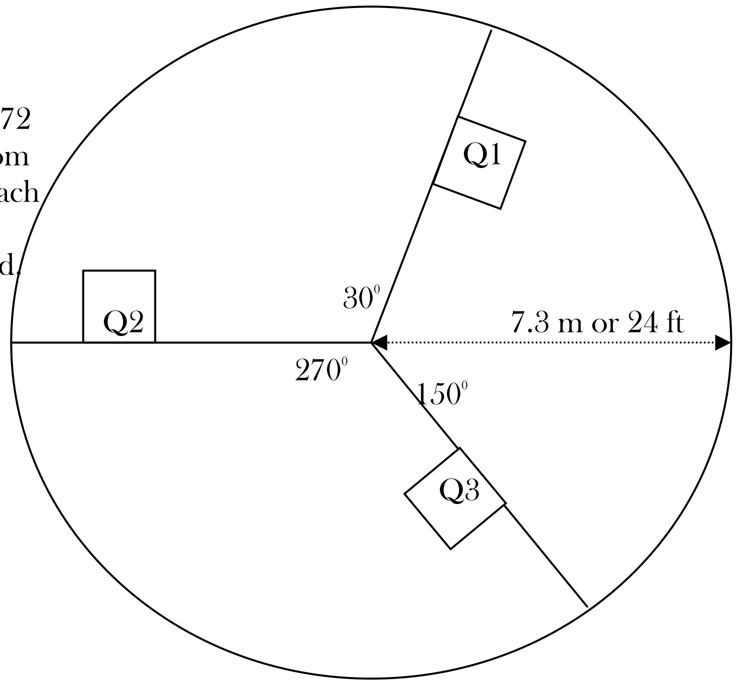


Diagram of the interior of any one of the subplots associated with the larger plant composition monitoring area.

SURVEY METHODS:
This section of the monitoring plan is more important for characterizing the habitat available to species noted in the permanent sampling sites than it is for comparing the area to other places within the US. Therefore, this protocol could be changed significantly from the FS MSIM protocol.

<u>Plot Layout</u>

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 120°, 240°, and 360°, respectively. Within each of the 10 subplots, 3 - 1 m^2 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 10 subplots, 1 woody debris transect (extending 17.6 m or 58 ft) should be established in 1 of the 3 pre-determined aspects (30°, 150°, 270°). Each transect is first marked in a straight line (using a hip chain 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 surveys should be random.

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). 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 3 habitat visits per site.

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

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%

Ground Cover Percent

An estimate of ground cover, using 5 classes: litter, vegetation, rock, soil/sand, and water, should be made such that the final percentage equals 100 for this plot. This plot is a 1 m^2 plot near the middle of the interior subplot. Due to trampling problems associated with centering this plot around the bird point count pole, this plot should be positioned approximately 2 meters toward the center point (point 1). The plot at the center of the hexagon should be positioned 2 m in any direction from this pole.

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°, 150°, or 270° 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 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%. 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 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

Vertical Vegetation

The woody debris transects also serve as point intercept lines for estimating vertical vegetation density. Beginning at 2m from the center point, all vegetation that touches the transect at exactly 2m is recorded (this is a vertical measurement only, the vegetation has to be touching at one point). Plant species and height are recorded. From this data, relative frequency of plant species and vertical density of vegetation can be estimated (Manley et al. 2005). The same procedure is repeated at 7, 12, and 17 meters from the center point.

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.

Tree and Snag Measurement

Tree species, diameter at breast height (cm), and height to nearest m are recorded for all trees and snags $\geq$ 28 cm (11 in) in diameter. Decay class is also estimated for snags following the same categories as used for the interior subplot snag decay classes.

EQUIPMENT LIST:

Plant press, cardboard, and newspaper to collect unknowns (this could be left in truck, but plants should be 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
Standard field kit: Clip board, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets.
Dissecting scope at lab or office

STAFF & TRAINING:

Two weeks of 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:
None.

DATA SHEETS:
Data sheets for this protocol are located in Appendix 2.

Chapter Twenty
Aquatic Habitat Classification Protocol

The principal motivations for collecting information on habitat and vegetative characteristics is to both monitor potential large-scale habitat changes over time and 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.

IOWA 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. The program coordinator has the responsibility of ensuring that duplicate efforts are avoided by the respective sampling teams (i.e. that the same information is not being collected by multiple people). However, 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.

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, artificial lakes, natural 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 topographic maps 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.

<u>Substrate Measurements</u>

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.

<u>Water Depths</u>

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).

<u>Woody Debris</u>

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
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, returning to each site for at least 2 visits might reduce the variation associated with timing, if the visits are close in time and under similar weather conditions. Errors associated with the technician (diligence, species ID, etc.) can be mitigated by having different observers do repeat visits.

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.

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DATA SHEETS:
Data sheets for this protocol are located in Appendix 2.

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