Idaho Waterways Survey

A Standard Operating Procedure for Aquatic Plants & Invasive Species

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The purpose of this document is to be used as a reference of the methods and procedures for all ISDA staff and partnering entities conducting survey activities on Idaho's waterways.

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Outline:

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I. Background

This SOP will describe a comprehensive protocol for conducting waterway surveys for each prescriptive survey category.

As each waterway is unique, those conducting the surveys must tailor the approved methods to each. Each waterway will be ranked based on risk for both aquatic weeds and invasive species. The factors that will determine high versus low risk are the following:

1. Lake, reservoir, or stream size
2. Primary use
3. Estimated annual visits
4. Average drawdown amount of reservoirs
5. Known infestations or proximity to known infestations

Each ranked waterway will need to have surveys conducted from areas of highest risk outward. For example, on a lake from the most used boat ramp out in both directions, or on a stream/river from most used boat ramp or access moving downstream. All survey methods conducted will be either shoreline or watercraft assisted. Safety is always of the utmost importance. Those conducting surveys should ensure safety protocols are followed and safety equipment is in place.

In addition to these, the following items should be standard for all survey activities:

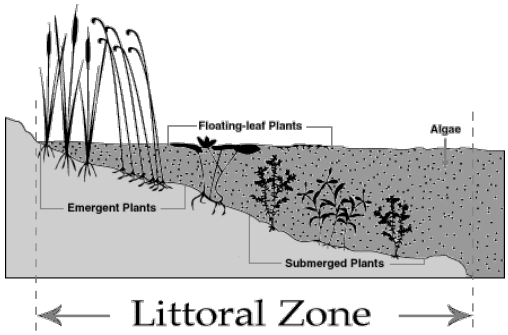
- GPS data unit
- Digital camera
- Photographic reference scale
- Water safety equipment-for watercraft. All ISDA staff must wear Personal Floatation Device (PFD) at all times when using watercraft.
- Depth finding unit or method

II. Aquatic Plants

A. Aquatic Plants Survey Equipment-

The following items listed are for all aquatic plant-sampling methods:

- Weighted aquatic weed rake attached to minimum 14m (~45ft) rope
- Sorting bin/tub
- Waders
- Snorkel or viewing tube
- Wet suit-for under water survey
- Paper towels-sample preservation
- Ziploc bags-sample preservation
- Cooler-sample preservation



B. Aquatic Plant Survey Methods-

There are three different types of aquatic macrophyte (plant) growth and in water. These are emergent plants, floating plants and submerged plants. All plant types inhabit the area of the waterway from shoreline outward where sunlight penetrates all the way to the sediment called the littoral zone. The littoral zone can vary based on water clarity and sediment type, typically 5-10m (16-32ft) in depth. The first step in any aquatic plant survey is to determine this zone. The way to determine this zone is to start at a depth of 5m (~16ft) conduct 1m (~3ft) rake samples. Depending on a presence or absence of macrophyte growth, increase or decrease depth at 1m (~3ft) increments. Repeat this test until either no plants are found (increasing depth) or plants are found (decreasing depth). Typically the more turbid (murky) the waterway appears the littoral zone will be shallower than those with less turbidity. This will give you a generalization of the littoral zone for the waterway.

The two methods used for aquatic plant surveys are visual or 1m (~3ft) rake samples. These methods can be used either as a shoreline or watercraft assisted survey.

1. *Visual*- what you can see. This survey method is a quick method for presence/absence of plants, either topped out or emergent plants. This method, in most cases, cannot provide an accurate density without additional steps or perfect weather and water conditions. Those additional steps being a visual accounting of subsurface macrophyte growth with either a viewing tube or a mask and snorkel. Visual surveys also can be conducted as a subsurface dive operation, but it can be very time consuming. The sampling protocols that will be used in conjunction with the visual method are as follows:
   a. Protocol 1- **Visual survey**-This protocol is used when plants are topped out, emergent, or as a part of a diver survey project. In some cases visual surveys can be performed when water conditions and lighting allows for a clear view to the bottom. Some visual surveys can also be conducted utilizing a view tube. Density will follow same scale as 1m rake sample; **1**-1-25%, **2**-25-75%, and **3**- 75-100%. The scale rating is based on the area visually observed while collecting GPS location. Attributes to be collected are target aquatic plant, density, depth,

waterbody and a sample or picture when necessary. The attributes and their meanings will be discussed in the data recording requirements section.

2. *1m rake sample*- this method uses a modified aquatic weed rake. The rakes head is weighted and attached to a 14m (~45ft) rope. The steps to use this device are as follows:

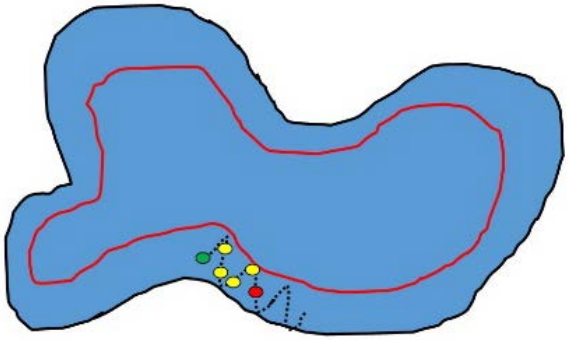
   i. Check depth
   ii. Grasp or attach end of line and coil rope in preperation to throw
   iii. Throw rake and allow time to sink to substrate
   iv. Drag rake along bottom for 1m (~3ft)
   v. Increase speed and angle of pull after 1m (~3ft) to lift rake from bottom. Because the edges of the rake are sharp, be careful and do not drag rake into boat or along shoreline.
   vi. Remove sample from rake into sorting tub/bin

The sampling protocols that will be used in conjunction with the 1m (~3ft) rake sample are as follows:

a. Protocol 2- **Random point survey**- The primary uses of this survey method is for presence/absence during lake wide inventory, or post treatment monitoring. Points are to be taken at random across the littoral zone of the water body. Protocol 2 would target the areas of highest probability of target plant, such as public access areas, then moving outward from there. Should a positive sample be taken advancement to Protocol 3 would be recommended. Attributes to be collected are target aquatic plant, density, depth, number of rakes, waterbody, and a sample or picture when necessary. The attributes and their meanings will be discussed in the data recording requirements section.

b. Protocol 3- **Systematic point survey**- This protocol will be used to better map and monitor target species distribution based on density and plant "patch" size. Sample points will be taken originating from the initial positive point (Protocol 2) every 10m (~32ft) of distance or 5m (~16ft) of depth change, in a zig-zag pattern, within the littoral zone of the waterway. This protocol will help to determine the leading edges of established target pests, which will allow for better management decisions. Attributes to be collected are target aquatic plant, density, depth, number of rakes, waterbody, and a sample or picture when necessary. The attributes and their meanings will be discussed in the data recording requirements section

Systematic point survey, redline indicating edge of littoral zone. Green point start, yellow continue, and red end. Black line is sample of boat route



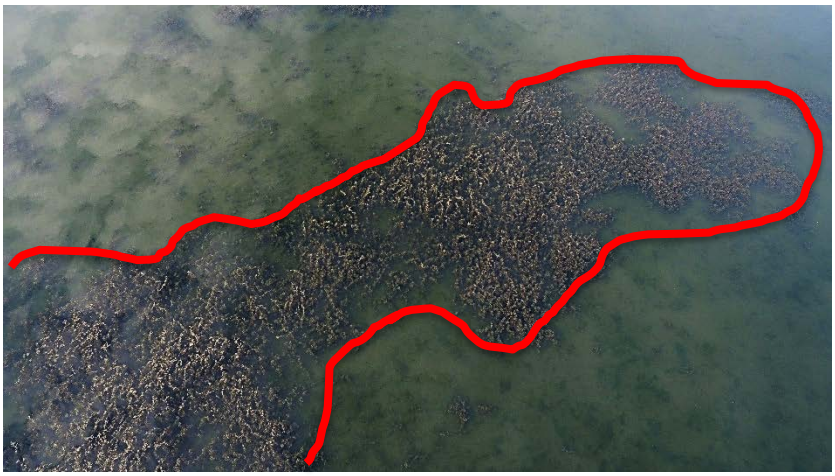


Image of Eurasian watermilfoil patch from 30 ft above the surface of the water

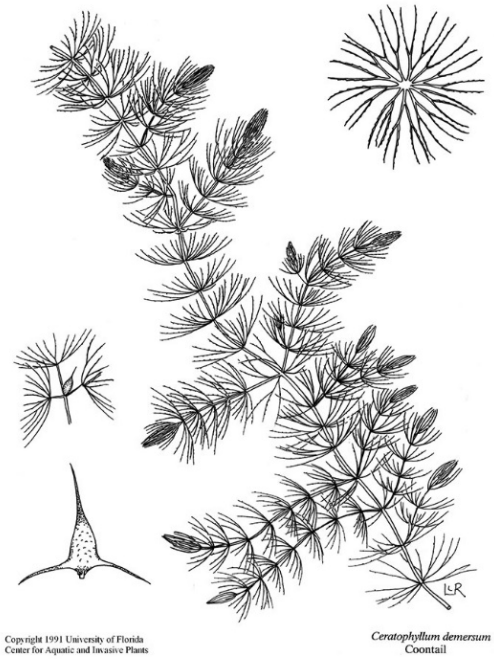
One of the key issues with aquatic plant management is being able to determine the target plants density. For ease of in field determinations, these ratings will use a 0-3 scale. With 0 being no plants found, 1 being 1% to 25%, 2 being 25% to 75%, and 3 being 75% and up. When using the 1m rake sample be careful not to arbitrarily attribute density by assuming sample is completely one species. Use the following figure as a guide. With the understanding that the image is only representing the target pest and not what could be a part of the retrieved sample.

0 – 0% target weed *1- 1%- 25% target weed* *2- 25% - 75% target weed* *3- 75% and up target weed*

C. Identification

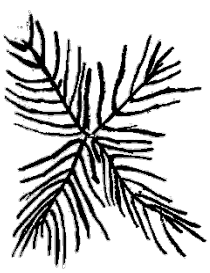
The most important aspect of making management strategy decisions is having an accurate plant identification. The sketches and general characteristics information for the following plant species is for reference only. Note that hybridization, environmental influences, and plant stress can skew the "normal" behavior of the plant in question. In addition, this reference will not list all species that could be seen in Idaho waterways. What will be covered is an example native and invasive species of each type of plant growth that can be found. Those types being submerged, free-floating, and emergent. To ensure the most accurate way to ensure proper identification is both in the base knowledge of plant taxonomy and the methodology used in identification. An additional method, which will be covered in the next section (Sample Preservation), is submitting the findings to an expert for diagnostic review.

1. Submerged Plants - plants that are rooted in the soil with plant parts growing with most all of the plants parts being completely submersed. Submersed plants usually require less light to grow than emergent plants and can be found in the deeper parts of the littoral zone. Examples of Submerged plants would be elodea, coontail, Eurasian watermilfoil, and Hydrilla.



Coontail

Scientific- *Cerstophyllum demersum*
Type- Submerged
Lifecycle- Perennial
Status- Native
Identifying characteristics- whorl of 5 or more fan shaped leaves that divide by fork-like structures and have spines along the midrib that give the plant a rough feel. Coontail is also unique in that it does not have roots, but rather specialized buried stems (DiTomaso, Healy, 2003) which allows this plant to also free float Often times confused as a milfoil. The difference is milfoils will have more of a feather like leaf where Coontail's will be forked. See illustration below.


northern milfoil


Eurasian watermilfoil

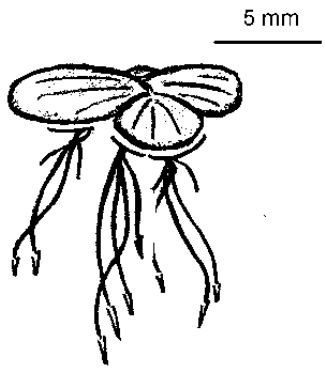

coontail

Hydrilla

Scientific- *Hydrilla verticillata*
Type- Submerged
Lifecycle- Perennial
Status- Idaho Noxious Weed, EDRR
Identifying Characteristics- whorls of 4-8 leaves with serrated leaf edges. Leaf midribs will also have teeth on the underside. Hydrilla produces tubers from the roots and turions along leaf axils. Native elodea can often be mistaken for this species but it does not produce a tuber or turions, and has smooth leaf edges and midribs.

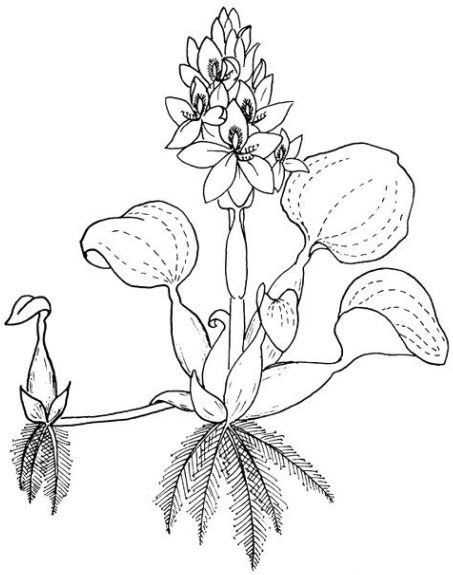
2. Free-Floating Plants- plants are not rooted in the soil and are floating freely at or near the surface of the water. Some plant species can be confused for free-floating plants but they have root structures that are in the soil and are truly emergent species. Common free-floating plants are duckweed, water meal, bladderwort, water hyacinth, and giant salvinia.

Duck weed



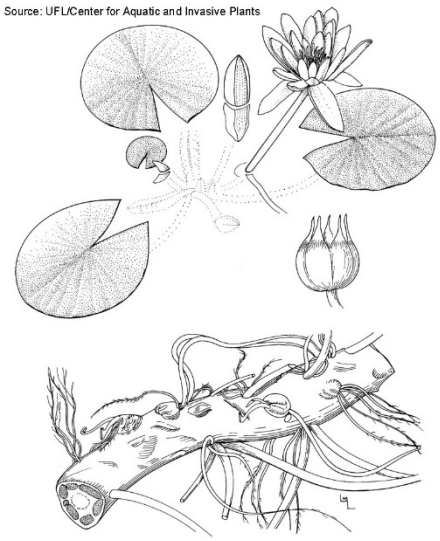
Scientific- *Lemna minor*
Type- Free-floating
Lifecycle- Annual
Status- Native
Identifying Characteristics- free-floating plant is very small and a light green in color. Plant may have up to 3 leaves (fronds) with one hair protruding from each leaf. Duckweed prefers calm waters, undisturbed by current or wave action. There are several different species of duckweed that look similar as well as another native species called watermeal.

Water hyacinth



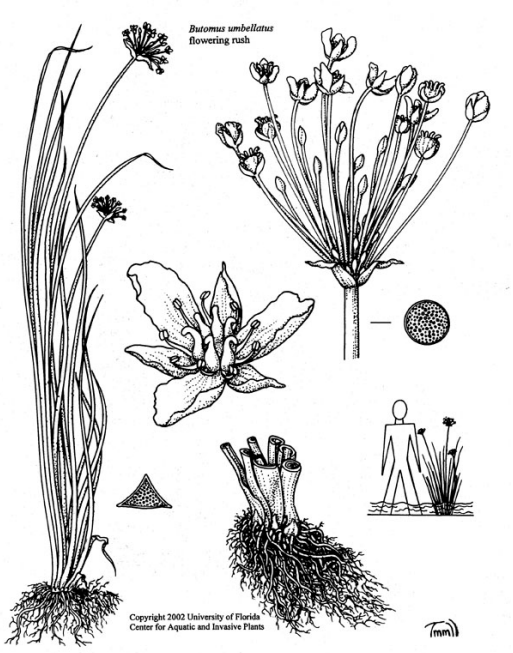
Scientific- *Eichhoria crassipes*
Type- Free-floating
Lifecycle- Perennial
Status-Idaho Noxious Weed, EDRR
Identifying Characteristics- Broad thick glossy oval shaped leaves with long sponge like stalks. Root hairs are feather like and dark in color. The plant can grow up to 1m (~3ft) tall with a very showy purple to lavender flowers. Hyacinth reproduces rapidly through runners and each plant produces several thousand seeds. This plant does best in warm waters and does not tolerate freezing temperatures, but has been found in Idaho in geo thermal waters

3. Emergent Plants- plants that are rooted in soils below the water's surface. With plant parts that are at or above the waters surface. Some species of emergent aquatic plants can have structures that float at the waters surface. Common emergent plants would be Fragrant waterlily, Bulrush, Cattails, Flowering rush, Parrotfeather milfoil, and Phragmites.



Fragrant waterlily

Scientific- *Nympaea odorata*
Type- Emergent
Lifecycle- Perennial
Status- Native
Identifying Characteristics- Plant is rooted to subsurface soils where it grows from branched rhizomes. The long stems that grow terminate in smooth, round floating leaves that may or may not be at the waters surface. The upper surface of the leaf has a waxy coating that is very water repellent. The flowers of this plant also float and are very fragrant. The flower opens and closes with each day, with white petals and yellow stamens.



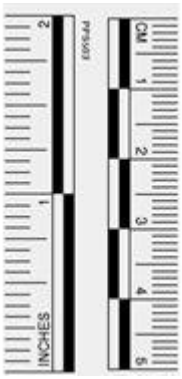
Flowering Rush

Scientific- *Butomus umbellatus*
Type- Emergent or submerged
Lifecycle- Perennial
Status-Idaho Noxious Weed, Containment
Identifying Characteristics- can grow up to 1.5m (~5ft) tall with leaves having a distinct triangle cross-section. Flower stem is round, with flower being very showy and white to pink in color. Flowers are arranged in a umbrella like shape (Umbel) with a large cluster of flowers. Roots are rhizomatous and very brittle aiding in the plants spread. This plants leaves are similar in appearance to native bulrush with the distinct difference being the cross-section as well as the showy flower.

D. Sample Preservation

During all survey activities there will be several times when a sample will be needed. Some of these instances could be for general plant identification or DNA analysis. The methods described here are again for reference purposes only and additional requirements may be necessary depending on the reasoning for sample collection, and the analytics being performed on the sample. The two primary methods that will be used as a part of ISDA surveys are photo documentation and full plant specimen collection.

1. Photo documentation- this is the most common method for general plant identification. The following guidelines should be followed when taking pictures. Multiple pictures will be needed, as not all key characteristics will be visible in a single picture.
    - If possible, retrieve the entire plant. After retrieval rinse plant, including roots, if needed to remove algae or other debris.
    - As aquatic plants appear differently out of water as opposed to in water (especially submerged plants) the sample should be photographed in a tote/bin with water to allow the plant to demonstrate key characteristics. If the plant in question is emergent water will not be needed.
    - For plants that are too large to fit in a sample tote/bin (phragmites), take sample photos with a solid, non-reflective background.
    - In all pictures a photographic reference scale, such as a ruler, coin, or forensic scale, should be used.
    - Pictures of key plant characteristics can be very diagnostic and helpful in the identification process. Some of these include plant cross-sections (whorl structures, stem shape, etc.), flower or flower-like structures, and leaf or leaf-like structures.
    - Please make sure all photos are clear, focused, and have adequate lighting.
    - In addition to photos taken, be sure to record characteristics that cannot be captured in a photo. Such as whether or not the plant has a smell (aromatic) or the texture and feel of the plant in question. Also be sure to note the habitat or growth characteristics observed.
    - All plant materials collected using this method should not be returned to the waterbody. Some could be invasive and most spread by fragmentation.



2. Full Plant Specimen- this method is most commonly used when additional diagnosis is needed, such as diagnostic DNA testing. The testing that will be done on these samples will each have additional requirements based on species and the entity/lab performing the analytics. Specifics on how the sample will need to be taken, when during the life cycle it should be collected, and which plant parts are most needed (DNA work at times does

not require the entire plant). The Following are generalities to be used to collect a full plant specimen.

- Retrieve as much of the target plant as possible, including roots.
- Rinse plant with clean water to remove debris and algae.
- Place plant on damp paper towel and then gently wrap specimen up with additional damp paper towels.
- Place wrapped sample in Ziploc bag and place in a cooler.
- Be sure to label all samples with the date, time, sample location (GPS), waterbody name, species (if known), number of sample (x of x), and your name.
- Refrigerate prior to shipping but do not freeze. Cooler temperatures will help to preserve DNA and slow down the plant degradation processes.

For aquatic plant survey/sampling questions contact:
Jeremey Varley: Jeremey.varley@isda.idaho.gov phone: (208) 332-8667

The general information regarding common practices are as described by the ISDA noxious weeds Section manager.
The aquatic plant information and identification referenced, contains both anecdotal information and information referencing the Aquatic and Riparian Weeds of the West DiTomaso, Healy, 2003

III. Dreissenid Veliger Early Detection Monitoring

A. Sample Location Selection

Samples should be collected using a boat, if possible. Preferably, a minimum of three locations within each waterbody should be sampled with a minimum of three plankton tows for each location. If possible, more than three tows and more than three sites should be sampled to increase the likelihood of collecting veligers, if present. Larger waterbodies, in particular, will require more intensive sampling. Samples from the same area will be composited together. Spread out sample sites to further increase the likelihood of collecting veligers. In lakes and reservoirs, focus sampling near dams, intakes, outflows, inflows, marinas, boat launches, and in areas that are downwind, downstream, in open water and are near-shore. In large streams and rivers, focus sampling in the main stem, downwind/downstream and near shore areas around boat launches, marinas and other structures that can create eddies.

B. Sampling Frequency

Veligers can exhibit spatial and temporal patchiness throughout the water column and high sampling frequency (weekly or bi-weekly) increases the likelihood of collecting veligers if they are present. The optimal time to sample for veligers is during peak spawning events, which vary with adult population density, location and temperature. Optimal temperatures for veliger production are between 16° and 19°C (61° and 66 °F). If possible, collect samples at a minimum of three times during the spawning season beginning when water temperatures are greater than 14°C (57 °F) and continuing until deep water temperatures exceed 24°C (75 °F). Veliger sampling can be performed anytime during the day but preferably not immediately following a storm event.

C. Sampling Equipment

- Plankton net (simple, conical plankton tow net, 63 or 64 µm pore size. *The mesh size is critical*)
- Line for deploying the net (30 m on spool or about 100 feet)
- Sample bottles (polyethylene material, 500 mL volume, leak-proof screw lid)
- Decontamination materials: 2 large buckets (>5 gal), bleach, white vinegar and two spray bottles, one containing white vinegar and one with tap water
- Preservative (absolute ethanol (ETOH). Do NOT use denatured ethanol or isopropyl alcohol (rubbing alcohol)
- Rubber gloves and eye protection (for handling ETOH)
- Data unit or field data sheets (waterproof paper), labels, waterproof marker and pencils
- Global Positioning System (GPS) unit
- Tweezers or small spatula (*recommended*)
- Boat (*recommended*)
- Temperature probe (*recommended*)
- Sealable plastic bags (e.g. Ziploc)
- Plastic garbage bags (large enough to hold 4 sample bottles)
- Cooler with ice packs or cubed / crushed ice

D. Plankton Sample Collection

Vertical and/ or oblique plankton tows are recommended if a watercraft is available. Veligers can be found throughout the water column, ranging from near the surface, to depths greater than 400 ft. If no watercraft is available, horizontal tows from shore can be collected.

Do not keep plankton tows that contain large amounts of sediment. **Large amounts of sediment interfere with sample analysis, bind up preservative, and may damage sampling equipment.** If your sample contains large amounts of sediment, dump the contents of net back into the lake, thoroughly rinse net and cod-end piece in lake, and then repeat the tow. Some sediment (i.e. suspended solids) may be captured in plankton tows, especially in turbid systems. In small amounts, some inorganic debris is acceptable.

<u>Vertical Plankton Tow</u>

1. Secure the cod-end piece and check that the line is securely attached to plankton net. Secure the other end of the line to the boat. Sometimes, vertical tows are easier to do in a two-person team, one with the sample cup and one handling the plankton tow net.

2. Record water temperature at depth. If water temperature is within the range for mussel spawning (14 - 24 ^{o}C), proceed with sample collection. Record depth and temperature of water at 1-meter intervals down to the maximum sample depth, or at the lake bottom.

3. Lower the net 30 m (100 ft) below water surface, or as deep as possible to 1 m above the sediment and record.

4. Keep net at this depth for 30 seconds then manually retrieve using a hand-over-hand technique at a rate of 0.5 m/s (1.5 ft/ s). Slow and steady retrieval is the key to collecting a good plankton tow.

5. Flush the collected sample into the cod end by raising the net so the cod piece is at the water surface. Rinse organisms down into the cod by lowering the net back into the water. Keep the opening above the water's surface, then quickly pull net straight up (this action will move collected plankton into the cod-end piece). Repeat this procedure several times to ensure that all the organisms inside the net are concentrated in the cod end.

6. A spray bottle filled with deionized or distilled water is to be used to wash down the net. Spray the outside of the net starting at the mouth to concentrate veligers into the cod end. Do not use tap water, since residual chlorine may destroy veliger tissue and DNA necessary for sample analysis.

7. Carefully unscrew the cod-end piece without spilling collected water and plankton. Condense the sample as much as possible before pouring into sample bottle. Condense the sample by swirling the cod-end piece. You may need to use tweezers or a spatula to gently clear the mesh netting in the cod-end piece to allow the water to filter through. Rinse the cod-end piece with a spray bottle several times with minimal volume of water and put rinses into the same sample bottle. If multiple sample bottles are needed for one

sample tow label the bottles “1 of 2”, “2 of 2”, etc. to ensure proper tracking of the samples.

8. Add additional plankton tow samples from the same area until the sample bottle is ¾ full.

9. Mark the sample level on the bottle with permanent ink (Draw a line).
10. While wearing the proper personal protective equipment (PPE), add ethanol (see Sample Preservation procedures on page 4).

11. Inspect existing docks, pilings, and along shoreline for mussels and other invasive aquatic species. If possible, wade upstream and downstream from the boat launch area. Pick up and inspect the rocks and plants in the area to look for invasive plants, snails, clams, crayfish and mussels. If you encounter an invasive organism, or something unfamiliar, collect a sample and submit it to ISDA. Record species encountered under “Notes” on the data unit or on the sample data collection form.

Figure 1: Seven vertical plankton tows collected and compiled into one sample bottle for analysis.

Horizontal Plankton Tow

Attach a weight (1-2 kg or 2-4 lbs) to the line immediately in front of the net opening to keep net below the water surface.

1. Secure the cod-end piece and check that the line is securely attached to plankton net.

2. Hold the ring of net, (metal loop that holds the net mouth open) using thumb and forefinger. Make large loops of the line and hold loosely with the same hand holding the net.

3. Firmly hold the other end of the line with free hand.

4. Throw the net using a sidearm-style, opening your hand upon release to allow line to feed out with the net.
5. Allow net to sink into waterbody. A weighted cod-end piece will aid in pulling the net into the water. If an air bubble gets trapped in the net, retrieve the net and start again.

6. Manually retrieve net using a hand-over-hand technique at a rate of 0.5 m/ s (1.5 ft/s). Keep the net off the sediment to avoid snagging and collecting debris.

7. Follow steps # 5 through # 11 used for vertical plankton tows.

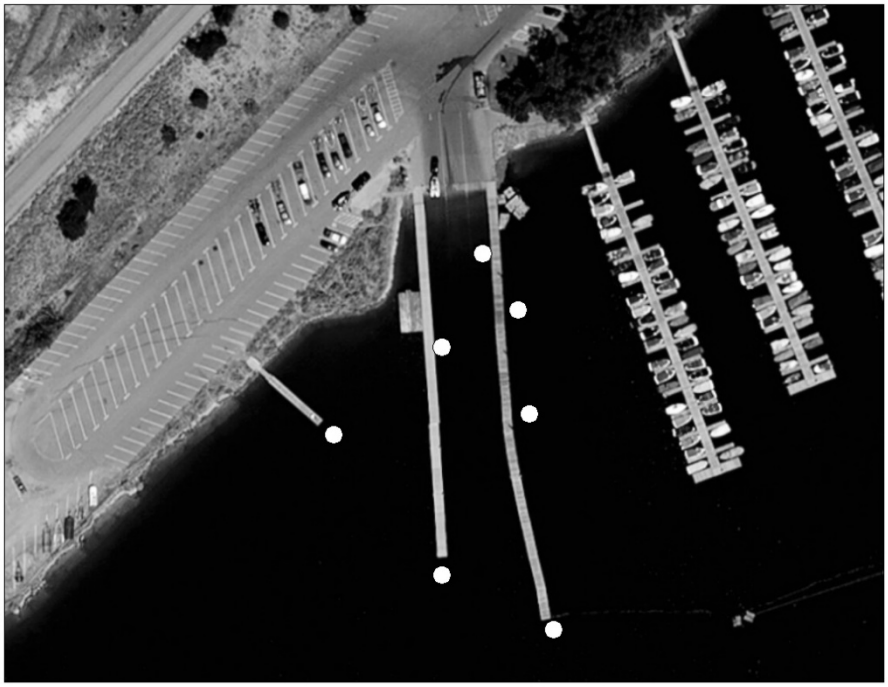


Figure 2: Six plankton tows, horizontal and vertical, collected and compiled in one sample bottle for analysis. A second shoreline sampling location should be in the adjacent marina.

E. Labeling and Associated Parameters

Record the following information on both the sample bottle label and on the data unit / field datasheet. Use an indelible ink permanent marker for bottle label and a pencil for datasheet. Be careful because permanent marker ink will smear when in contact with ethanol.

- Date of collection
- Waterbody name
- Site location. Be as descriptive as possible
  - If more than one sample at that location record (1 of 2, 2 of 2)
- GPS coordinates (Decimal Degrees)
- Water depth (meter)
- Number and length of tows

- Name of collector

Upload the data from the data unit daily. Be sure to check the information to ensure that all the information is accurate and complete.

F. Sample Preservation

Absolute ethanol (ETOH) is used for sample preservation. Before using ETOH, read and understand the Chemical Safety Data Sheet (SDS) document that is available in all ETOH storage areas. Always wear chemical safety goggles and rubber gloves when handling. Only open the container in a well-ventilated area. Keep away from sources of ignition. No smoking when handling ETOH.

Preserve samples using 25% ETOH immediately after collection (see procedure below) to ensure sample integrity. Following the addition of ETOH, place sample immediately on ice. Do NOT use denatured or rubbing alcohol.

1. Make sure sample bottle is ¾ or less full of sample. If needed, pour some sample into another bottle. Tighten cap and thoroughly mix sample prior to pouring into another sample bottle.
2. Mark the level of sample on the outside of sample bottle using a permanent marker.
3. Put on chemical safety goggles and rubber gloves.
4. Using a stock solution of 95% to 100% ETOH, add one part ETOH to three parts sample to achieve a final concentration of approximately 25% ETOH. A ruler or measuring tape may be placed alongside the sample bottle to help estimate the rations.
5. Tightly secure sample bottle lid.
6. Immediately place samples on ice in a cooler.

Samples preserved using a final solution of 25% ETOH must be stored on ice and kept refrigerated prior to and following analysis. Microbial activities will continue in 25% ETOH and degrade samples. Place sample bottles in a cooler filled with ice in field immediately after collection, then transfer to a refrigerator as soon as possible. Ship samples FedEx Priority Overnight to Aquaticus LLC Lab in Chiefland FL (see address below) the same day or the day following collection. Please avoid weekend deliveries. It is strongly recommended that samples and ice packs are placed in an airtight container that is then placed inside the cooler. This will help maintain low temperatures during transit.

G. FedEx Mailing Procedure

1. Place sample bottles in sealable plastic bags and then place bottles inside a plastic garbage bag. Sample bottles from the same waterbody may share sealable and garbage bags. Samples from separate waterbodies should be bagged separately.
2. Place several frozen blue ice packs into each garbage bag containing plankton samples.
3. Place garbage bags into a cooler. Be sure there are no less than 4 blue ice packs in the cooler.
4. Secure the cooler lid and tape it closed.
5. Take cooler to the nearest FedEx location (http://fedex.com/Dropoff/start)
6. Send the cooler to the following address:

Aquaticus LLC (formerly Western Biological Services LLC*)
12251 NW 85th Ave

Chiefland FL 32626
Attn: Steve Wells

7. Billing Reference: ISDA Invasive Species. (PCA 42214)
8. Send samples Priority Overnight (Avoid Weekend Deliveries).

H. Equipment Decontamination

Field equipment must be decontaminated at the site to prevent transfer of organisms within and between systems.

Decontamination procedures are as follows:

1. Be sure all sampling equipment is thoroughly rinsed following sample collection.
2. Soak equipment in 5% bleach solution for 30 minutes.
   - To make 5% bleach solution use 7oz (a little less than 1 cup) of household bleach to 1 gal (16 cups/ 128 oz) of water.
3. Thoroughly rinse equipment with fresh-water.
4. Soak equipment in 5% acetic acid solution (white vinegar) for 24 hours.
   - To make 5% vinegar solution use 7oz (a little less than 1 cup) of vinegar to 1 gal (16 cups/ 128 oz) of water.
5. Thoroughly rinse equipment with fresh-water and allow to dry.

Clean, drain and dry all equipment and watercraft following use. If possible, avoid launching watercraft into more than one waterbody per day to allow thorough dry time. If possible, decontaminate watercraft using hot water power wash (≥140^{o}F on boat surface).

For sampling questions contact:
Nic Zurfluh: Nicholas.zurfluh@isda.idaho.gov phone 208-631-9574.

The information in this protocol is based on the zebra and quagga mussel early detection monitoring protocol from Portland State University Center for Lakes and Reservoirs.

IV. Dreissenid Adult Early Detection Monitoring

A. Sample Location Selection

- Submerged hard surfaces including docks, pilings, channel markers, floating bathrooms, buoys, bridge abutments, seawalls, rocks, and logs.
- Shoreline areas including gravel, sand, mud, cobble and woody debris, especially in downwind, downstream, or other positions where shells and debris are concentrated.
- The bottoms and sides of submerged objects especially in protected and shaded areas such as nooks, crannies and junctions of two different surfaces.
- Areas where the water currents and/or wind patterns are likely to concentrate the planktonic larvae, as well as dead adult shells, e.g., near dam or outflow, particular bays, eddies, etc.
- High-use boater areas and points of entry, e.g., near marinas and launches.
- Main-stem, open water and near-shore areas.
- Lake bottom to exposed shoreline areas in well mixed water-bodies, e.g., reservoirs along Snake River.
- Thermocline to surface in stratified lakes and reservoirs.

B. Sample Collection Techniques

Substrate inspection (Hand pat-down)

1. Locate suitable existing submerged surfaces to inspect. Accessible surfaces (i.e., within arm's reach) that are good candidates for visual and tactile inspections, include the undersides and sides of dock floats, floating bathrooms, buoys and mooring chains, and the underside and sides of rocks found in shoreline areas.
2. Carefully pat surface with the palm of your hand and fingers. Do not run your hand along surfaces because of sharp objects. Note that periphyton may obscure attached bivalves and other specimens.
3. Remove hard protruding objects for visual inspection.
4. Record activities in data unit.
5. Retain suspect specimens in sample container or Ziploc bag. Label sample (see part D). Place in cooler on ice.

Surface scraper

1. Locate suitable submerged structures to inspect. Surface scrapers work well on vertical concrete walls, bridge abutments and cutwaters, channel markers, pilings, underwater booms, and breakwaters.
2. Carefully position boat near structure to sample (e.g., channel marker) and maintain position either using the motor or using water current and wind to position boat against structure.
3. When using the surface scraper, lower it into the water as deep as the pole will allow. Using both hands on the pole, bring the metal rim of the mesh box in contact with the substrate surface and quickly pull up, keeping the metal rim in contact with the surface to be sampled. The sessile communities collected in the mesh are visually inspected for the presence of bivalves while in the field.

4. Repeat step #3 at multiple locations per structure in order to sample a representative portion. Discard debris.
5. Record activities in data unit.
6. Retain suspect specimens in sample container or Ziploc bag. Label sample (see part D). Place in cooler on ice.

WARNING: Be careful not to pin arms between the boat and structure (i.e. pinch points).

Benthic ponar grab

1. Deploy the ponar dredge in areas of gravel, small cobble, sand and mud. Engage the spring-pin into dredge and carefully lower the dredge keeping tension on the rope. Lower dredge until it settles in or on bottom. Dredge pin will disengage and you can feel the dredge deploy.
2. Quickly retrieve dredge and dump contents into plastic sorting bin.
3. Pour liquid, debris, and sediment out of bin through sieve.
4. Inspect remaining contents for bivalves and snails. Discard debris.
3. Record activities in data unit.
4. Retain suspect specimens in sample container or Ziploc bag. Label sample (see part D). Place in cooler on ice.

Drawdown shoreline walk

1. Walk in a zig-zag pattern parallel to shoreline in wade-able depths near boat launches and other areas that contain shells, cobble, gravel, and sand.
2. Stop every other step to pull out loose rocks, cobble and woody debris and/or aquatic plants to visually inspect for mussels and snails.
3. Visually inspect for bivalve shells partly buried in sand as well as dead shells on top of sediment.
4. Record activities in data unit.
5. Retain suspect specimens in sample container or Ziploc bag. Label sample (see part D). Place in cooler on ice.

Plant shake-down

1. Collect macrophytes with thatch rake or by hand and place into 5-gallon white-colored bucket with fresh water.
2. Vigorously shake the macrophytes in bucket and water to detach invertebrates.
3. Visually inspect contents for crayfish, bivalves and snails on plants when placing plants into bucket, and again when removing plants and sorting for macrophyte collection.
4. Allow bucket and water to sit in sunlight while sorting plants.
5. Visually inspect the sides of bucket for small attached snails.
6. Pour liquid and debris out of bucket through sieve.
7. Inspect remaining contents for bivalves and snails. Discard debris.
8. Record activities in data unit.
9. Retain suspect specimens in sample container or Ziploc bag. Label sample (see part D). Place in cooler on ice.

Place all sampler containers and bags collected from the same water body into a large plastic trash bag and tie off.

C. Sampling Equipment

- Stainless steel ponar dredge, 25 lb.
- Line for deploying the dredge (30 m on spool or about 100 feet)
- Solid bottom sieve pan
- 2 ½ / 64 in. round sieve
- 4 ½ / 64 in. round sieve
- Surface scraper(s)
- Thatch rake
- Digital camera
- Sorting bin (white color)
- 5-gallon bucket (white color)
- Sample bottles (polyethylene material, 500 mL volume, leak-proof screw lid)
- Decontamination materials: 2 large buckets (>5 gal), bleach, white vinegar and two spray bottles, one containing white vinegar and one with tap water
- Preservative (absolute ethanol (ETOH). Do NOT use denatured ethanol or isopropyl alcohol (rubbing alcohol)
- Rubber gloves and eye protection (for handling ETOH)
- Data unit or field data sheets (waterproof paper), labels, waterproof marker and pencils
- Global Positioning System (GPS) unit
- Tweezers or small spatula (*recommended*)
- Boat (*recommended*)
- Sealable plastic bags (e.g. Ziploc)
- Plastic garbage bags (large enough to hold 4 sample bottles)
- Cooler with ice packs or cubed / crushed ice

D. **Labeling**

Record the following information on both the sample bottle label and on the data unit / field datasheet. Use a permanent ink marker for bottle label and a pencil for datasheet. Be careful because permanent marker ink will smear when in contact with ethanol.

- Date of collection
- Waterbody name
- Site location. Be as descriptive as possible
- GPS coordinates (Decimal Degrees)
- Water depth (meter)
- Name of collector

Upload the data from the data unit daily. Be sure to check the information to ensure that all the information is accurate and complete.

E. **Equipment Decontamination**

Field equipment must be decontaminated at the site to prevent transfer of organisms within and between systems.

Decontamination procedures are as follows:

1. Be sure all sampling equipment is thoroughly rinsed following sample collection.
2. Soak equipment in 5% bleach solution for 30 minutes.
   - To make 5% bleach solution use 7oz (a little less than 1 cup) of household bleach to 1 gal (16 cups/ 128 oz) of water.
3. Thoroughly rinse equipment with fresh-water.
4. Soak equipment in 5% acetic acid solution (white vinegar) for 24 hours.
   - To make 5% vinegar solution use 7oz (a little less than 1 cup) of vinegar to 1 gal (16 cups/ 128 oz) of water.
5. Thoroughly rinse equipment with fresh-water and allow to dry.

Clean, drain and dry all equipment and watercraft following use. If possible, avoid launching watercraft into more than one waterbody per day to allow thorough dry time. If possible, decontaminate watercraft using hot water power wash (≥140^{o}F on boat surface).

For sampling questions contact:
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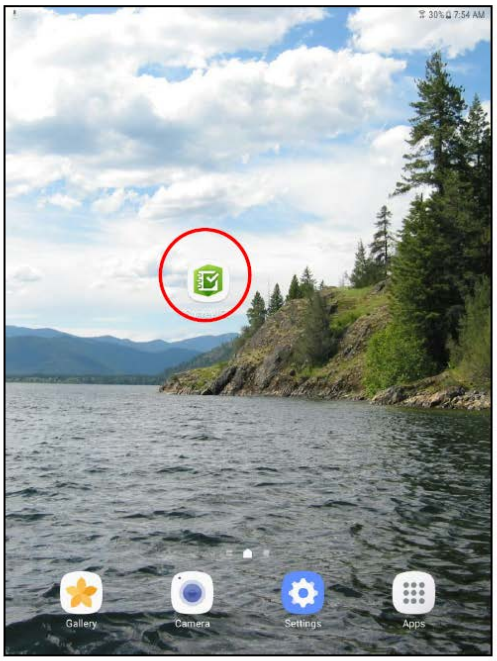
V. Data Recording

Aquatic Survey Program

...uploading your data just got a whole lot easier!



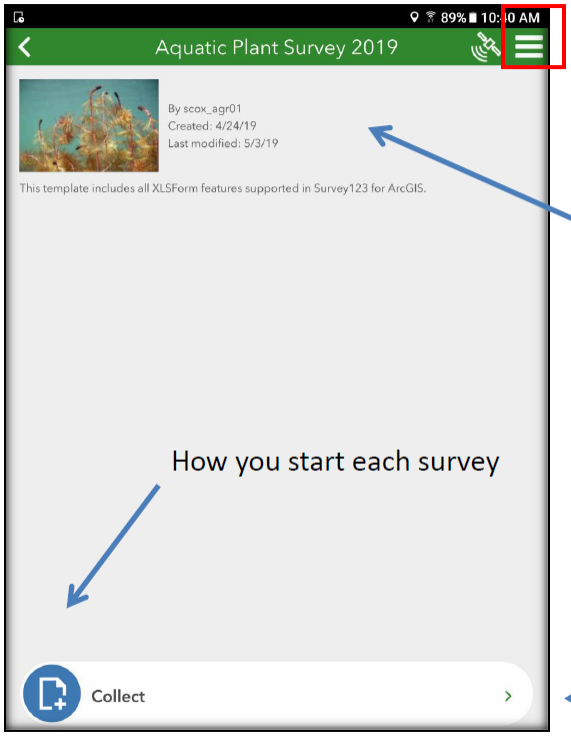
This is the new Inspection Program

Link to the Settings

Link to the Apps on the Unit

Link to the Main Page

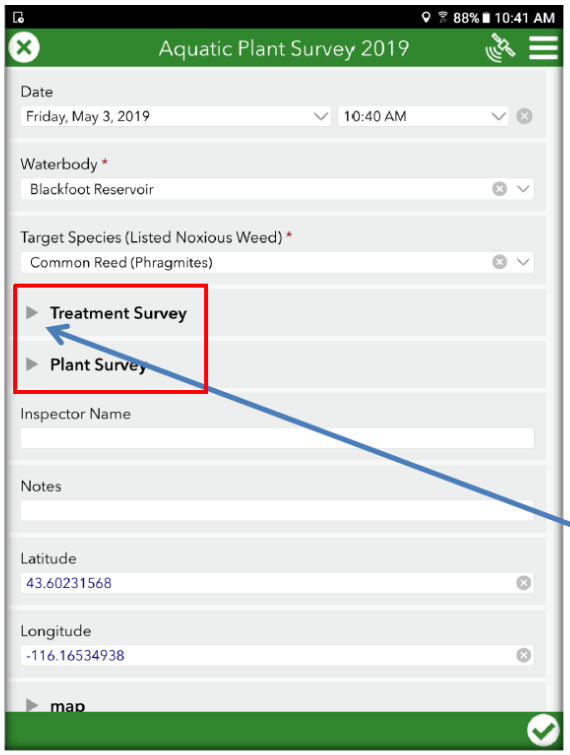
Back Button



Menu to delete the entire survey form. Don't delete the survey form unless you are advised.

Name of the project you are using.

Choose "Collect" to start survey.



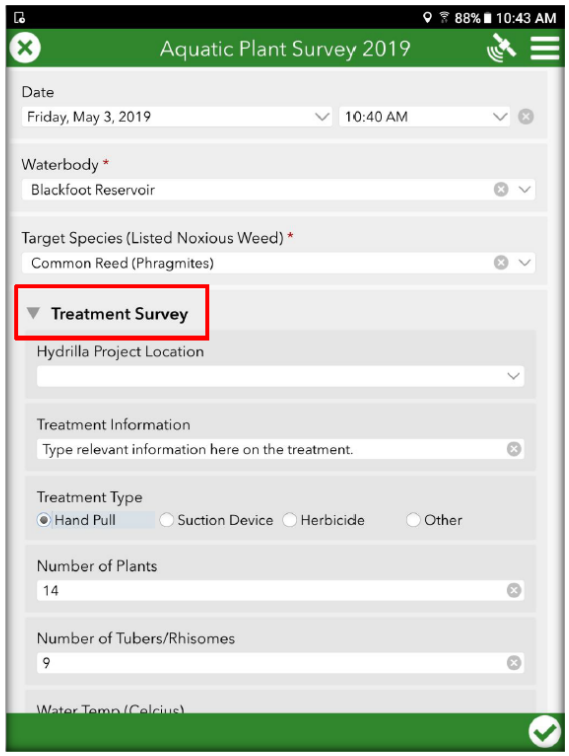
Date and Time

Waterbody where the activity is taking place.

Target Plant

Is a treatment occurring, or is it a general plant survey?

Determine which you are doing and then click on the sideways triangle to open the questions.



Treatment Survey option...

For Hydrilla only there is a project location question. If you are doing Hydrilla work please choose from the pick-list.

Relevant information

What type of Treatment?

Number of plants removed/treated?

Number of tubers/rhisomes removed/treated?

Enter in the water temperature and soil temperature if possible for the survey. Remember to use Celcius.

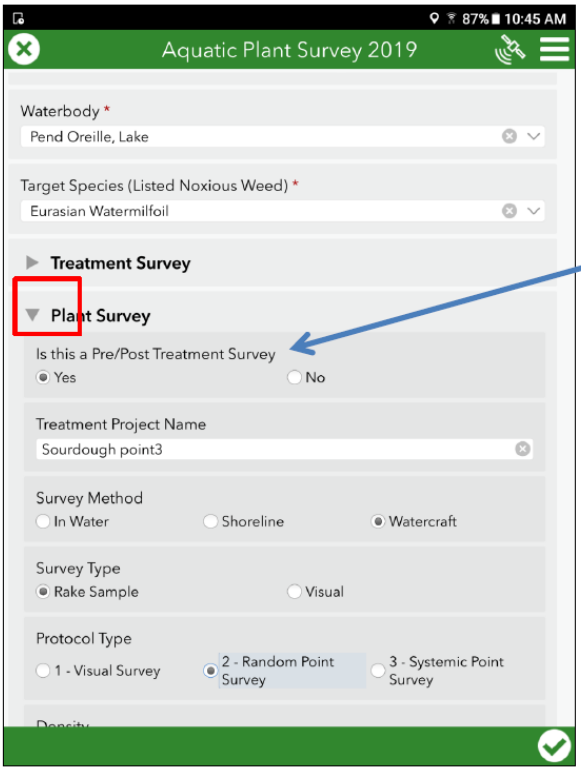
Did you take pictures?

If you need to add pictures click on the sideways triangle to open the ability.

You have the ability to enter up to three (3) pictures.

The camera icon will take a new picture, and the folder icon will allow you to search on the device for an image you already took.

Plant Survey option…



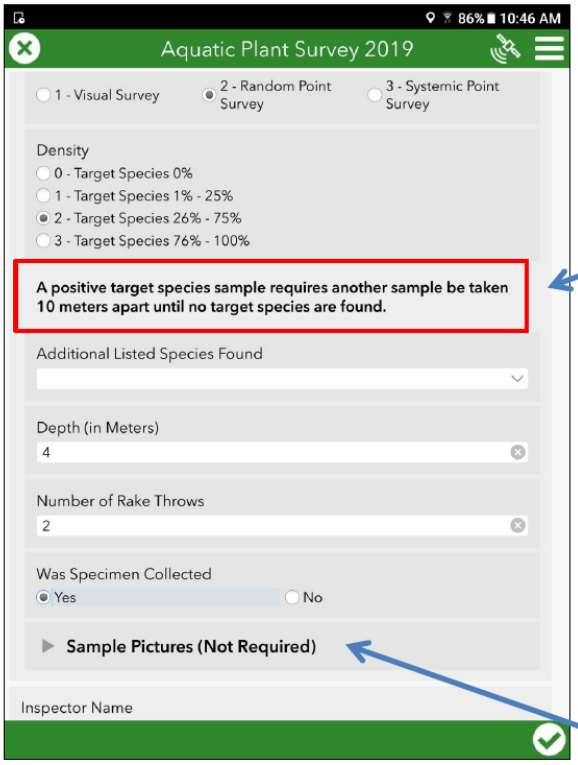
Is the plant survey part of an approved treatment?

If yes please indicate the treatment project name.

Which Survey method did you perform?

Which Survey type did you perform?

Which protocol type did you perform?



Density of the **target** plant ONLY

No plants found will be a "0"

If you enter a "1", "2", or "3" a message will pop-up reminding you that you will need to take another sample 10 meters away to help determine where the population ends.

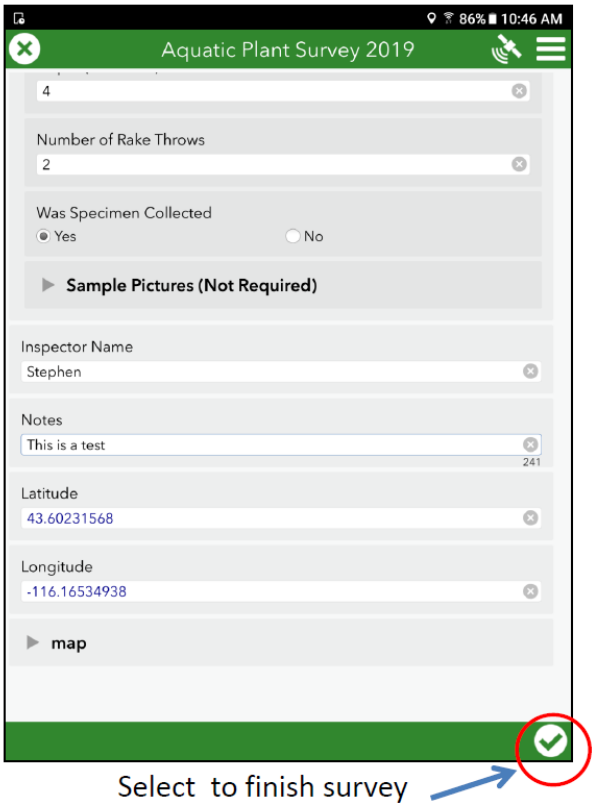
Must be listed species in Idaho

Depth of the plant survey

Number of rake throws

Was a specimen collected?

You have the ability to enter up to three (3) pictures.



Select to finish survey

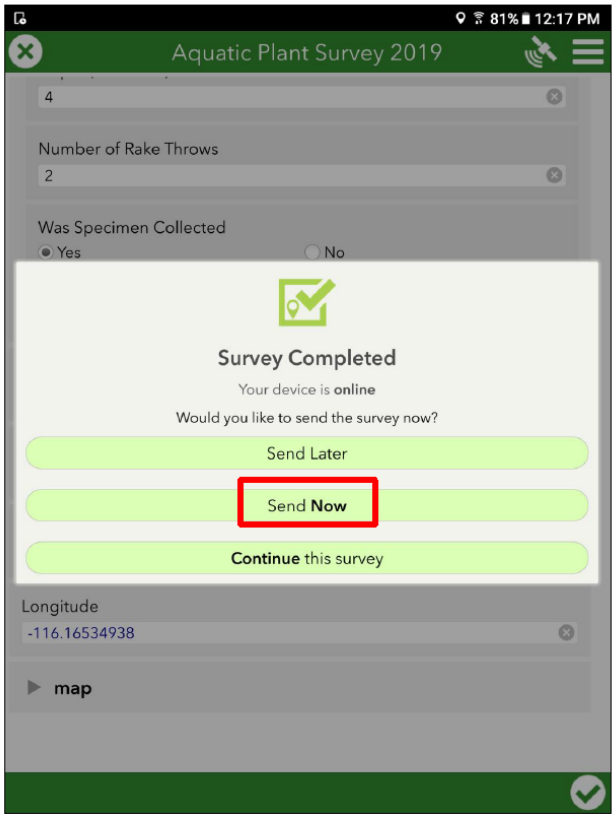
The GPS coordinates are generated automatically for you.

If the coordinates are not generating for you then you may not have the GPS turned on your device.

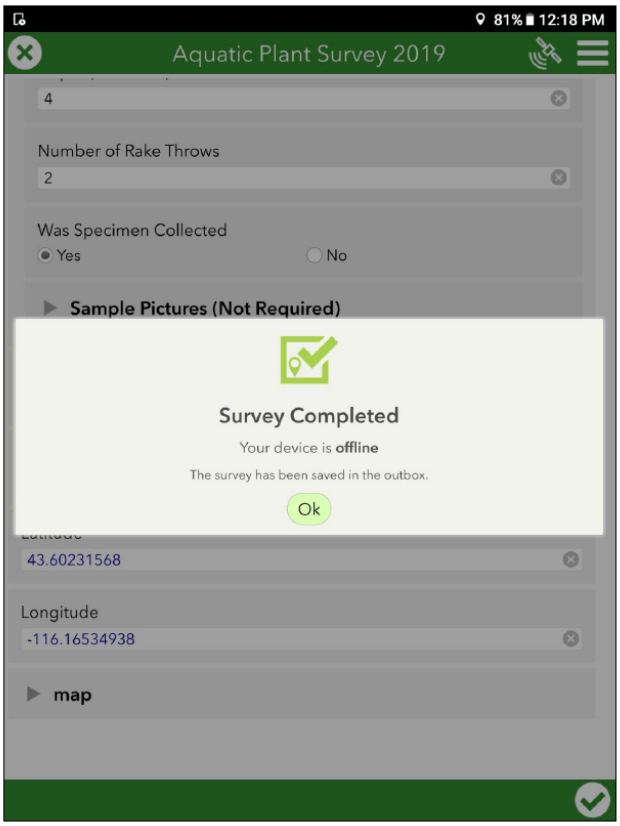
Depending on the device you may need to start the weed survey over to allow it to recognize the GPS you just turned on.

If you are in a narrow canyon and GPS will not register please document in the notes as much information as you can allowing ISDA to "assign" a location at a later date.

The "map" is closed automatically. Tap on the triangle symbol to activate the map if you choose.



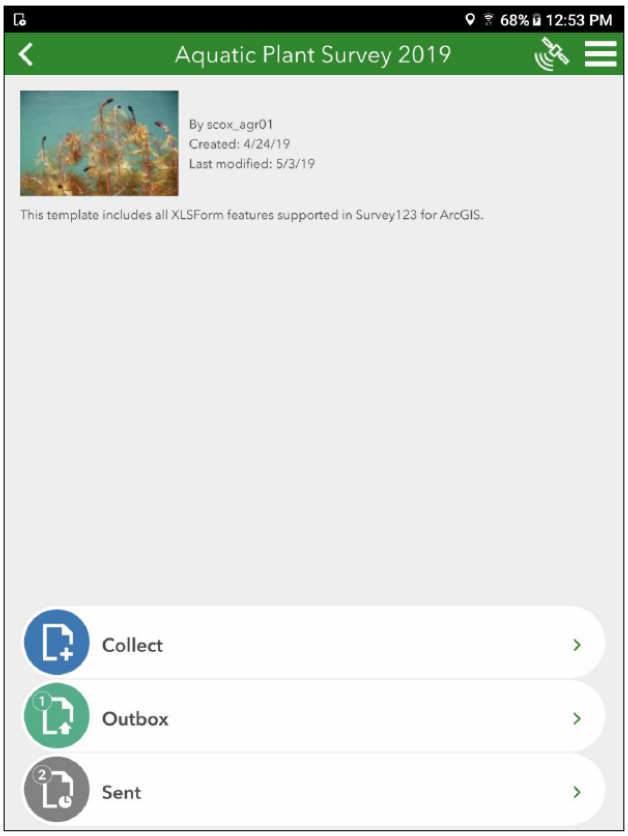
If you are connected to the internet you have the option to upload the inspection form immediately…please do it.



If you are working at an area with a weak internet signal (MiFI) or you don't have access to the internet at all you will be working with the unit "offline".

The information will be saved in the outbox to be uploaded at a later time.

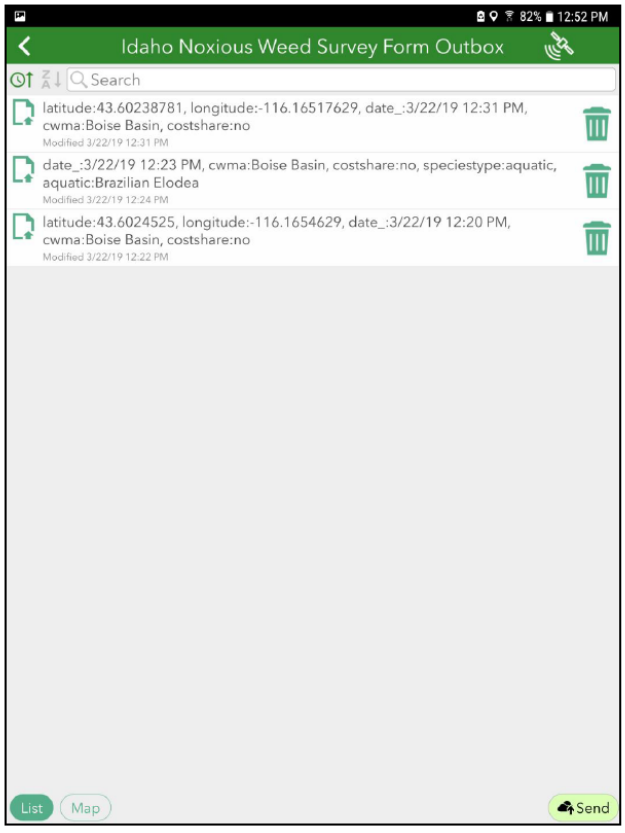
Working Off-Line



Most likely you will be working in an area without an internet connection.

If that is true you will need to access your data to upload it.

Access the plant surveys you have performed and need to upload from the “Outbox” tab.



Make sure you are connected to the internet or you will not have the “Send” button on the bottom right.

Simply select the “Send” button and all the records will be uploaded at the same time.