

DRAFT Field Protocols for Performing Mid-Atlantic Tidal Fringe Wetland Assessment Version 1.0 10/2/2006

USE OF METHOD

The Mid-Atlantic Tidal Fringe Wetland Assessment (MATFWA) draft field protocols were developed to assess the condition of reference mesohaline and polyhaline tidal fringe wetlands in the Indian, Nanticoke, and York Rivers. Methods described in these protocols are currently being field-tested.

GENERAL GUIDANCE

A. Time and Effort Involved

varies

B. Field Teams

varies

C. Experience and Qualifications Needed

The MATFWA should only be performed by individuals who have completed a training course on how to properly perform this method. Users of this method should have experience and/or education in the identification of wetlands including an understanding of the various stressors that impact different wetland types, native flora of the region, and soil properties.

KEY TO DETERMINING WETLAND SUBCLASS IN THE COASTAL PLAIN

I. Is the wetland influenced by tidal cycles from a Bay or Ocean?

No – go to step III

Yes – go to step II

II. Is the wetland influenced by tidal cycles from the Ocean?

A. Yes – Marine Tidal Fringe subclass

B. No – Estuarine Tidal Fringe subclass

1. Further classify estuarine tidal fringe subclass by salinity

a) Polyhaline – 18 to 30 ppt

b) Mesohaline – 5 to 18 ppt

c) Oligohaline – 0.5 to 5 ppt

d) Freshwater - <0.5 ppt

III. Is the wetland in a valley or stream channel where it gets inundated by overbank flooding from that stream or river in an unaltered condition (i.e. if a stream has been channelized and no longer receives overbank flooding it could still be a riverine wetland in an altered condition)?

A. No – go to step IV

B. Yes – Riverine subclass

IV. Is the wetland in a topographic depression, outside areas that are inundated by overbank flooding, in which water ponds during at least part of the year?

A. No – go to step V

B. Yes – Depressional subclass

V. Is the wetland at the bottom of a topographic slope?

A. No – Flat Subclass

B. Yes – Slope subclass

LOCATING ASSESSMENT AREA

The Assessment Area (AA) is the area within a wetland that will be sampled using the MATFWA. All measurements will be performed in the AA with the exception of landscape measurements that assess conditions surrounding the AA. When subjectively selecting a reference wetland, a center point should be located that is representative of the wetland and the ecological condition (least disturbed to highly degraded) to be sampled.

- A. Mark the center of the AA.
- B. Establish the AA as a 40-m radius circle centered on the point (0.5ha area), with one axis or 80m transect running parallel to the upland/wetland interface and the other axis perpendicular to the first.
- C. **Note:** Attempt to locate the AA entirely within either high or low marsh, and within the dominant vegetation zone (i.e. *Spartina alterniflora*, *Juncus roemerianus*/*Scirpus robustus*) present.

SITE INFORMATION DATASHEET

Site #: Unique number for site (assigned by crew leader)

Site Name: Names are given to each site

Date: Month, day, and year of sampling

Field crew: All members of the field crew that participated in sampling the site

Crew Leader Initials: Initials of the Crew Leader present during sampling

HGM Subclass: (Estuarine Tidal Fringe) record salinity classification according to definitions and key above (i.e. polyhaline, mesohaline)

Reference or Assessment Site: Circle that which applies. Reference sites are subjectively selected because they represent a specific condition such as minimally disturbed or impacted by a specific stressor or represent an ecological variation of a wetland class. Reference sites do not have to be pristine or minimally disturbed.

Watershed: record the watershed in which the site is located (Nanticoke, Indian, York)

Wetland Size: record the size of the wetland in hectares, this information can be found from the NWI or state wetland maps

Photos: The frames that are shot should be marked on the datasheet. If more than two are taken, the range can be given.

Lat/Long: Latitude and longitude coordinates in digital degrees

Qualitative Condition: rate the condition of the wetland from least disturbed to highly disturbed based on best professional judgment and relative to other sites in the watershed. Use Figure 1 as a guide for determining the appropriate rating. Surrounding landuse may be considered in the Qualitative Condition rating. *Note: sites that are potential reference standard sites should have a qualitative condition score of 1.*

Tiered Aquatic Life Use Conceptual Model: Draft Biological Tiers - 1

(10/22 draft)

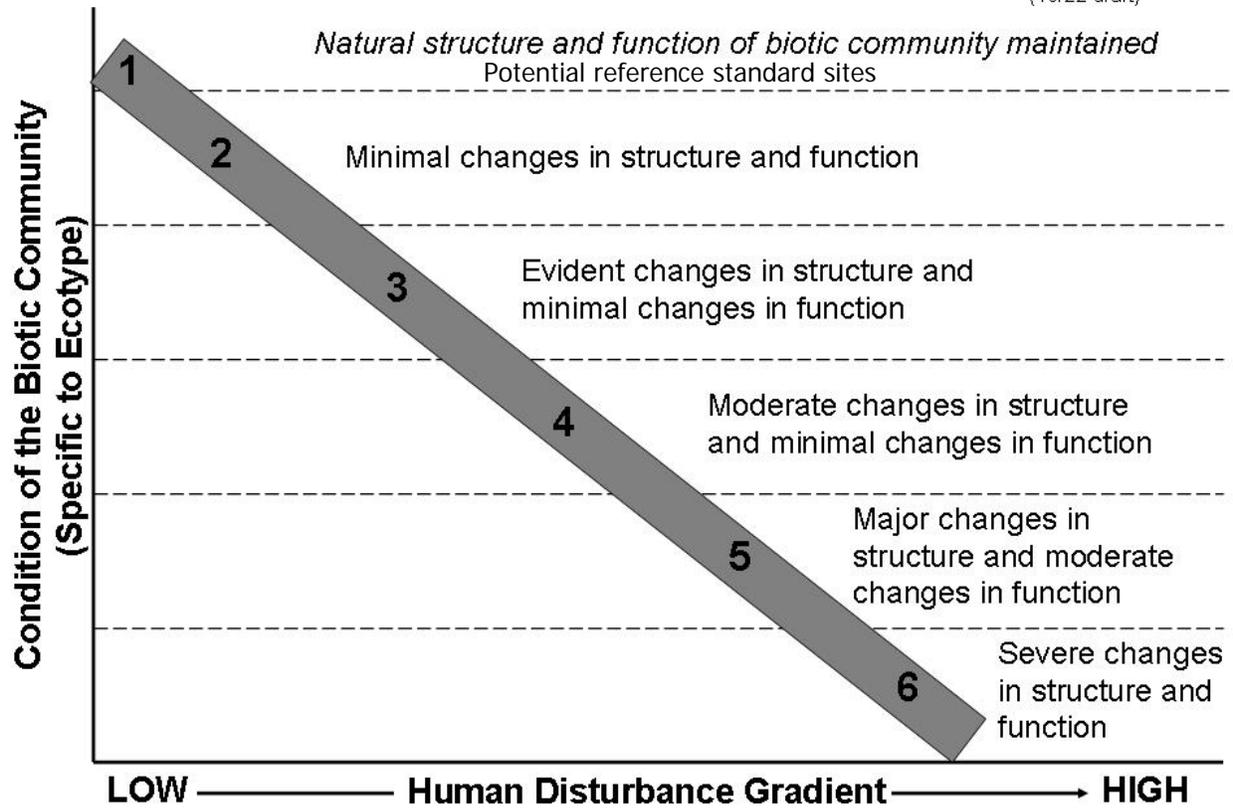


Figure 1. Diagram of narrative criteria for qualitative ranking of condition

I.A. Wetland Vegetation Zones and Features

What to measure: Percent cover of the AA of all vegetation communities and wetland features present in the AA.

How to measure: Walk the assessment area and observe the different vegetation communities and wetland features present. Circle the dominant wetland vegetation zone (low marsh or high marsh) on the space provided on the top of the box.

In the space provided, estimate the percent cover (refer to cover class midpoints below) of the assessment area for all the vegetation communities and wetland features present in the AA. If a vegetation community or wetland feature is present that is not listed, use the other box(es) and write in a description of the community/feature. If a community or feature is not present record a "0".

Where to sample: In the assessment area (AA)

Equipment needed: none

II. Barriers to Landward Migration

What to measure: any structure, feature, etc., preventing the landward migration of the marsh

How to measure: Features can be identified from the Level II shoreline survey maps and validated in the field. In the field, record any features located along the upland/wetland interface between the two endpoints of the transect parallel to the upland/wetland interface. This may require standing at one endpoint and walking a straight line to the upland/wetland interface. Then follow the interface approximately 80m to a point corresponding to the other endpoint of the transect, and record any additional features observed.

Where to sample: along the wetland/upland interface

Equipment needed: none

III. Tidal Restriction

What to measure: any disturbances restricting tidal waters from entering or leaving a site

How to measure: Features can be identified from aerial photographs and validated in the field. Any additional features observed in the field should be noted in the box.

Where to sample: within the AA as well as the surrounding area

Equipment needed: none

IV. Hydrology Alterations

What to measure: This is a qualitative variable and should be discussed as a group at the completion of sampling. Observations should be made on alterations to the hydrology of the marsh within the Assessment Area.

How to measure: Walk the AA and estimate the percent of the AA impacted either by being covered with fill or by being impounded with water due to a road, control structure, or culvert. Also note the presence of any ditches on the marsh. **Fill is defined as any soil, debris, garbage, or excavated material placed in the Assessment Area (this includes ditch spoil).** Check the appropriate line for the condition(s) that is/are present in the marsh.

Where to sample: In the assessment area

Equipment needed: none

V. Sketch of Site

Draw a sketch of the site. Important attributes to include are different vegetation communities, roads in or adjacent to the AA, and other large alterations such as development activities in part of the site. Also include an arrow indicating North.

VEGETATION DATASHEET

Site #: Unique number for site (assigned by crew leader)

Site Name: Names are given to each site

Date: Month, day, and year of sampling should be provided

Vegetation Zone: Record the vegetation zone (low marsh, high marsh)

Observers: All members of the field crew that participated in collecting data on this sheet

A single data sheet will be used to record plant species data in the AA. Herbs will be sampled in 16 1m² subplots located along two perpendicular transects that dissect the AA.

I. Species

What to measure: The percent cover of all species including woody and herbaceous species.

How to measure: Estimate the cover class of each species and unvegetated ground in each 1m² quadrat. Record the midpoint of the appropriate cover class. Cover classes and their midpoints are listed on the datasheet. When recording the percent cover of vegetation use the following guidelines:

- Lay the plot down as to not disturb the vegetation, if the area is very dense (i.e. a *Phragmites* stand) assemble the plot around the vegetation.
- Estimate the cover of all species present within the plot.
- If a plant is rooted outside of the plot but is contributing cover to the understory of the plot then include this species in the cover estimates, i.e. a plant is leaning into the plot from the outside. Make sure that this situation is not caused artificially when laying down the plot.
- If a plant is rooted in the plot but most of the leaves are leaning outside of the plot, record the presence of this species in the plot but with a low cover class (note exceptions to this below for woody shrubs and trees)

Where to measure: 16, (1m x 1m) quadrat plots (1m²). Plots should be placed at 31, 21, 11, and 1m, or at 49, 59, 69, or 79m from the center of the AA along each transect that perpendicularly bisects the AA. The quadrat should be placed starting at the designated distance (i.e., the first plot should be between 49 and 50m, or between 31 and 30m, from the center depending on which direction you are moving along the tape). If the vegetation has been disturbed by walking along the transect, place the plot off to the side of the transect in an undisturbed area by randomly selecting which side of the transect to place the plot.

Equipment needed: 1m x 1m herb plot quadrat, two 100m measuring tapes

Unidentified Species:

Species unable to be identified in the field should be given an Unknown name and number (Ex: Unknown spp. # 1, Unknown *Carex* # 2) and a sample should be collected

for pressing if there are multiple plants of the same species at the site. DO NOT collect a specimen where there is only one plant to avoid any impact to rare and threatened species. If this is the case take detailed notes about the plant or attempt to key it out in the field. Unknown species should be pressed so as to include the whole plant (flower and roots), and should be labeled with: Site name, Site #, Plot #, habitat that plant was growing (i.e. on a tidal bank, in standing water, in salt panne), Name of Collector, and Date (Figure 3). The unknown name and number should also be placed on the datasheet along with an “x” in the column labeled “C” for collected. After the unknown plant is pressed, it should be entered into the unknown database; and then once it is identified, the unknown database should be updated with the scientific name. When updating the datasheets with the scientific name, the unknown name and number should be erased and replaced with the scientific name, and an “x” should be placed in the column labeled “I” for identified.

Figure 3. Example of a plant label to include with a collected specimen

UK Juncus #1
Salt Meadow 103
Subplot #2
Growing on tidal bank
7-4-03 Jacobs

II. Invasive Species

What to measure: Invasive species in Assessment Area

How to measure: Walk the Assessment Area and note the presence of invasive species that are listed on the Delaware, Maryland, and Virginia National Heritage Program (NHP) lists. Record the midpoint for the percent cover that each species is covering for the subplots within the AA, the percent cover of the species in the vegetation zone that the vegetation plot is in, and for the entire Assessment Area. **Note:** if any invasive species are seen outside the AA but not within the wetland make a note of the species and the intensity of their occurrence in the comments section.

Where to measure: In entire Assessment Area.

Equipment needed: NHP invasive species lists for each state (see Appendix B at end of protocols)

PORE WATER AND SOILS

Site #: Unique number for site (assigned by crew leader)

Site Name: Names are given to each site

Date: Month, day and year of sampling should be provided

Vegetation Zone: Record the vegetation community where each soil sample is collected

Observers: All members of the field crew that participated in collecting data on this sheet

I. Pore Water

What to measure: pore water pH, Dissolved Oxygen (mg/L), Conductivity (μ s), Temperature (Celsius), and salinity (ppt).

How to measure: Calibrate the YSI meter for dissolved oxygen before taking measurements at each site using the following steps:

Protocol for calibrating the YSI meter- Dissolved Oxygen

*Should be calibrated each time YSI is turned off and on.

1. Turn on instrument and press “mode” button until dissolved oxygen is displayed in mg/l. Wait for the DO and temperature readings to stabilize (usually takes 15 minutes).
2. Once stabilized, use two fingers to press and release both the UP ARROW and DOWN arrow buttons at the same time.
3. The LCD will prompt you to enter the local altitude in hundreds of feet. In Delaware enter “1”. Use the arrow to increase or decrease the altitude. Press Enter once the correct altitude is displayed.
4. The YSI should then display “CAL” in the lower left, the calibration value in the right, and the current DO reading on the main screen. Make sure current DO reading is stable before you continue.
5. Press Enter. The display should read SAVE and then return to normal operation mode.

Protocol for changing DO membrane

* The membrane cap should be changed once a month.

1. Unscrew and remove sensor probe guard.
2. Unscrew and remove the old membrane cap. Make sure the tip is a shiny gold. If it is not, it will have to be cleaned (See instruction manual)
3. Rinse with distilled water.
4. Prepare electrolyte solution according to the directions on the KCL solution bottle.
5. Hold membrane cap and fill at least $\frac{1}{2}$ way with electrolyte solution.
6. Screw membrane cap onto the probe moderately tight. A small amount of electrolyte should overflow. Hold the membrane cap with a chem.-wipe while changing so oils and dirt do not touch the membrane.
7. Once new cap is on, check for air bubbles.

8. Screw the sensor probe guard back on.

Calibrate the pH pen each morning before taking measurements as follows:

Protocol for calibrating the pH meter

1. Press ON/OFF button
2. Dip electrode ½ to 1” into chosen buffer (4 or 7)
3. Press CAL button the enter Calibration mode. “CA” will flash on the screen.
4. After at least 30 seconds press the HOLD/CON button to confirm calibration. The display will show “CO” and then switch to the buffer value reading.
5. Rinse electrode

Field Measurements: Use the following procedures to locate the macroinvertebrates/soil/pore water sampling location. Obtain four random numbers from a random numbers (1 to 39) table. Starting from center in each quadrant, measure the specified distance from center (at a 45° angle) to the point to be sampled. The 45° angle insures that the area to be sampled has most likely not been trampled during vegetation sampling. Follow the surface macroinvertebrate, soil bearing capacity, and plant fragments sampling procedures. After removing the soil core (see below), allow the hole to fill with water. Using the YSI, record the dissolved oxygen (mg/L), specific conductivity (mS), temperature (°C), and salinity (ppt) readings. Turn the pH pen on and allow it to sit for ~30 seconds. Record the reading on the datasheet.

Note: specific conductivity should be recorded from the screen with mS units and a flashing °C at the bottom of the screen

Where to measure: within each quadrant of the AA

Equipment needed: YSI meter that can measure temperature, specific conductivity, salinity, and dissolved oxygen; pH pen and pH calibration buffer solutions

II. Soil Bearing Capacity

What to measure: soil bearing capacity

How to measure: After performing the surface macroinvertebrate sampling procedure, the vegetation inside the 1/10m ring should all be removed. Calibrate the soil penetrometer by placing the tip on a hard surface and pressing the Start key. Without placing any extra pressure on the penetrometer, hold down the Review key while pressing and releasing the Start key. After a few beeps, you may then release the Review key. Once the unit stops beeping, a reading (usually 10 or 15psi) should appear on the screen. The penetrometer is now calibrated. Place the tip of the penetrometer on bare substrate avoiding any remaining vegetation. Press the start key and VERY slowly press the penetrometer into the soil. Continue pushing the penetrometer until the bottom of the control box almost reaches the magnetic ring. Then slowly and gently pull the penetrometer out of the soil. The unit will beep once as you remove it from the soil. Once removed from the soil, press the Review key and record the measurements (psi) from 0 to 18 inches on the datasheet. If the marsh is very hummocky, obtain readings for both the hummock and hollow areas. If the marsh is not hummocky, obtain readings within the 1/10m ring and in any bare panne or pool areas within the quadrant. Repeat

these measurements in each quadrant. (modified from Carlisle and Wigand, 2004; M. Stolt, personal communication)

Where to measure: within Assessment Area

Equipment needed: penetrometer, spade or shovel

III. Plant Fragments Quantification

What to measure: root fiber volume from the root zone

How to measure: The plant fragments measurements will be taken in the same area as the pore water and soil compaction measurements. After the surface macroinvertebrate and soil compaction measurements are recorded, remove the metal 1/10m ring from the substrate. In the bare area where the vegetation has been clipped, use a shovel to dig a section of the marsh soil interface. From the soil core, use the sheers to cut out a 5cm x 5cm x 2cm sample just below the plants in the top zone of the substrate (at a depth of 2-4cm). Cut the sample into smaller pieces and place the pieces into a wide mouth bottle with at least 250mL of water. Shake very well. Dump the bottle contents onto a 2mm sieve. Gently push the materials against the sieve to remove excess water, leaving the fibrous, partially-decomposed plant matter and other plant fragments on the sieve. Collect the remaining fibrous material in the sieve and place it into the tube of a 60cc syringe. Push the plunger down on the syringe to further squeeze out excess water. Push the syringe plunger with pressure to remove most of the water. When there is significant resistance, stop. Record the final root fiber volume from the syringe. Repeat the same procedure at a depth of approximately 25-27cm. If you are unable to cut a sample from 25-27cm, record the depth of the deeper sample next to the measurements on the datasheet. Repeat in each quadrant within the AA (adapted from Carlisle and Wigand, 2004; M. Stolt, personal communication). Record any comments and the vegetation community next to the measurements on the datasheet.

Where to measure: within Assessment Area.

Equipment needed: clippers, shovel, sheers, carpenter's ruler, wide mouth bottle, water, 2mm sieve, 60 cc syringe

MACROBENTHOS

Site #: Unique number for site (assigned by crew leader)

Site Name: Names are given to each site

Date: Month, day and year of sampling should be provided

Vegetation Zone: Record the vegetation community and wetland zone sampled

Observers: All members of the field crew that participated in collecting data on this sheet

I. Macrobenthos

What to measure: macroinvertebrates

How to measure: Obtain four random numbers from a random numbers (1 to 39) table. Starting from the AA center, in each quadrant measure the specified distance from center (at a 45° angle) to the point to be sampled. The 45° angle insures that the area to be sampled has most likely not been trampled during vegetation sampling. Place the 1/10m metal ring on the substrate at this point (try to avoid knocking snails off the vegetation when placing the ring). Count all macroinvertebrates on the vegetation within the ring by cutting the stalks and carefully examining them for snails. Once all the vegetation is removed, count any snails, crabs, or mussels on the substrate surface. Record the number of each species found within the ring, as well as any fiddler crab burrows evident on the substrate surface. Also record the dominant vegetation within the ring. In two of the quadrants, obtain a substrate sample as follows. After performing the plant fragments measurements, cut a 5x5x18cm deep sample from the soil core. Place the substrate sample in a Zip-Lock bag labeled with site number, sampling location (quadrant number), and date of sampling. Finally, along a tidal creek or ditch draining the AA, place the 1/10m metal ring on the bank. Identify and count the number of macroinvertebrates on the vegetation and substrate surface as detailed above. Then use a shovel and the sheers to collect a 5 x 5 x 18cm sediment sample. Place the sediment in a Zip-Lock bag labeled with site number, sampling location (bank sample), and sampling date. Each of the three macroinvertebrate substrate samples should be washed in the sieve bucket before transport back to the lab for sorting. Dump the sample into the sieve bucket. Submerge the base of the bucket in water and agitate to remove as much small material as possible. Carefully collect the remaining sample and return it to its respective labeled Zip-Lock bag. Store samples in the cooler for transport back to the office. Then place the samples in the refrigerator until sorting.

Where to measure: vegetation and substrate surface at all four quadrant sampling locations; from soil core at two quadrant sampling locations; at surface and from soil core at one bank location

Equipment needed: 1/10m metal ring, shovel, sheers, carpenter's ruler, zip-lock bags, sharpie marker, sieve bucket

MAN-MADE STRUCTURES (Delaware Sites ONLY)

Site #: Unique number for site

Site Name: Names are given to each site

Date: Month, day and year of sampling should be provided

Vegetation Zone: Record the vegetation zone where the structure exists

Observers: All members of the field crew that participated in collecting data on this sheet

I. Man-made structures in wetlands

What to measure: presence of structures in study wetlands and vegetation effects of the structures

How to measure: Record the presence and description of any walkway, duck blind, pier, dock, boat ramp, rip-rap, etc. within the study wetland. Record the type of construction materials (salt treated wood, fiberglass grates, aluminum floats, fill, etc.) for each structure. Measure the extent of the impacted area (m²) (length, width, compass orientation for walkways). For walkways, assess the health (height) of wetland vegetation under the structure compared to adjacent vegetation.

Where to measure: around structures

Equipment needed: 50m tapes, compass

Appendix A Equipment List

General Equipment:

Clipboards
Pencils
Post-it-Notes (scrap paper)
Duct Tape
Binoculars
Bug Spray
Sunscreen
First Aid Kit
Flagging
Water cooler
Hip Waders
Site map(s)
Field Protocols
GPS
Compass

Vegetation:

1m² PVC quadrat
2- 100m measuring tapes
NHP invasive species lists

Pore Water:

Compass
50m measuring tape
Random numbers table (1-39)
YSI meter (Dissolved Oxygen, Temperature,
Specific Conductivity, Salinity)
pH meter
pH meter solution
Deionized water in squirt bottle

Soil:

soil penetrometer
Shovel
Carpenter's ruler
Sheers
500mL wide mouthed bottle
bottle of water
2mm sieve
60cc syringe

Macroinvertebrates:

1/10m metal ring
shovel
sheers
carpenter's ruler
Zip-Lock bags
Sharpie marker
sieve bucket

Appendix B. State Invasive Species Lists

Delaware

Acer platanoides
Acer pseudoplatanus
Ailanthus altissima
Akebia quinata
Alliaria petiolata
Ampelopsis brevipedunculata
Arthraxon hispidus
Berberis thunbergii
Bidens polylepis
Cabomba caroliniana
Carex kobomugi
Celastrus orbiculata
Cirsium arvense
Clematis terniflora
Commelina communis
Conium maculatum
Convolvulus arvensis
Duchesnea indica
Egeria densa
Eichhornia crassipes
Elaeagnus umbellata
Eragrostis curvula
Euonymus alatus
Festuca pratensis
Galanthus nivalis
Hedera helix
Hemerocallis fulva
Humulus japonicus
Humulus lupulus
Hydrilla verticillata
Iris pseudacorus
Leuconjum aestivum
Ligustrum vulgare
Lonicera japonica
Lonicera maackii
Lonicera morrowii
Lonicera tatarica
Ludwigia peploides Raven spp. *Glabrescens*
Lysimachia nummularia
Lythrum salicaria
Microstegium vimineum
Murdannia keisak
Myriophyllum aquaticum
Myriophyllum spicatum
Packysandra terminalis
Phalaris arundinacea
Phragmites australis
Pinus thunbergiana
Pistia stratiotes

Poa trivialis
Polygonum cespitosum
Polygonum cuspidatum
Polygonum perfoliatum
Pseudosasa japonica
Pueraria montana
Ranunculus ficaria
Rhodotypos scandens
Rosa multiflora
Rubus idaeus ssp. strigosus
Rubus phoenicolasius
Sorghum halepense
Trapa natans
Urtica dioica
Vinca minor
Wisteria floribunda

Maryland

Acer platanoides
Ailanthus altissima
Ajuga reptans
Alliaria petiolata
Allium vineale
Ampelopsis brevipedunculata
Artemisia vulgaris
Arthraxon hispidus
Bidens polylepis
Bromus sterillis
Carduus acanthoides
Carduus natans
Catalpa spp.
Caulerpa taxifolia
Celastrus orbiculatus
Centaurea maculosa
Cirsium arvense
Cirsium vulgare
Clematis terniflora
Coronilla varia
Dioscorea oppositifolia
Dipsacus sylvestris
Duchesnea indica
Eichhornia azurea crassipes
Elodea densa
Euonymus fortunei
Festuca elatior
Glechoma herderacea
Hedera helix
Heracleum mantegazzianum
Hermerocallis fulva
Humulus japonicus
Hydrilla verticillata
Iris pseudacorus

Laminum amplexicaule
Lamium purpureum
Lespedeza cuneata
Liriope spicata
Lonicera japonica
Lysimachia nummularia
Lythrum salicaria
Microstegium vimineum (Eulalia viminea)
Miscanthus sinensis
Morus albus
Myiophyllum brasiliense
Myiophyllum spictatum
Myosoton aquaticum (Stellaria aquatica)
Ornithogalum nutans
Ornithogalum umbellatum
Paulownia tomentosa
Perilla frutescens
Phalaris arundinacea
Pharagmites australis
Picea glauca
Pinellia ternata
Polygonum cuspidatum
Polygonum perfoliatum
Polygonum sachalinense
Populus alba
Potamogeton crispus
Prunus avium
Pueraria loboata
Ranunculus ficaria
Salvinia molesta
Solanum dulcamara
Sorghum bicolor
Sorghum halepense
Trapa natans
Vinca minor
Wisteria floribunda

Virginia

Acer platanoides
Agropyron repens
Agrostis tenuis
Ailanthus altissima
Ajuga reptans
Akebia quinata
Albizia julibrissin
Alliaria petiolata
Allium vineale
Alternanthera philoxeroides
Ampelopsis brevipedunculata
Arostis gigantea
Arrhenatherum elatius
Artemisia vulgaris

Arthraxon hispidus
Arundo donax
Berberis thunbergii
Carduus nutans
Carex kobomugi
Cassia obtusifolia
Celastrum orbiculata
Centaurea biebersteinii
Centaurea jacea
Cirsium arvense
Cirsium vulgare
Commelina communis
Conium maculatum
Conyon varia
Convolvulus arvensis
Dactylis glomerata
Dioscorea oppositifolia
Dipsacus laciniatus
Dipsacus sylvestris
Egeria densa
Elaeagnus angustifolia
Elaeagnus pungens
Elaeagnus umbellata
Eragrostis curvula
Euonymus alata
Euonymus fortunei
Euphorbia esula
Festuca elatior (F. pratensis)
Foeniculum vulgare
Glechoma hederacea
Hedera helix
Holcus lanatus
Humulus japonicus
Hydrilla verticillata
Imperata cylindrica
Ipomoea coccinea
Ipomoea hederacea
Ipomoea purpurea
Iris pseudacorus
Lapsana communis
Lespedeza bicolor
Lespedeza cuneata
Ligustrum obtusifolium
Ligustrum sinense
Lonicera fragrantissima
Lonicera japonica
Lonicera maackii
Lonicera morrowii
Lonicera standishii
Lonicera tatarica
Lonicera x bella
Lotus corniculatus

Lysimachia nummularia
Lythrum salicaria
Melia azedarach
Melilotus alba
Melilotus officinalis
Microstegium vimineum
Miscanthus sinensis
Morus alba
Murdannia keisak
Myriophyllum aquaticum
Myriophyllum spicatum
Pastinaca sativa
Paulownia tomentosa
Perilla frutescens
Phleum pratense
Phragmites australis
Phyllostachys aurea
Poa comressa
Poa trivialis
Polygonum cespitosum
Polygonum cuspidatum
Polygonum perfoliatum
Populus alba
Pueraria montana
Ranunculus ficaria
Rosa multiflora
Rubus phoenicolasius
Rumex acetosella
Rumex crispus
Setaria faberi
Sorghum halepense
Spiraea japonica
Stellaria media
Trapa natans
Ulmus pumila
Veronica hederifolia
Viburnum dilatatum
Vinca minor (V. major)
Wisteria floribunda
Wisteria sinensis
Xanthium strumarium

Glossary

Assessment Area (AA) – area within the mapped wetland that is sampled using the Mid-Atlantic Tidal Fringe Wetland Assessment.

Ditch - A man-made, open drainage-way in or into which excess surface water or groundwater drained from land, stormwater runoff, or floodwaters flow either continuously or intermittently; grid ditching for mosquito control

Fill – any soil, debris, garbage, or excavated material including dredge spoil.

Invasive Species – a non-native or introduced species that has developed a tremendous capacity for reproduction and distribution throughout its new home and that also has a negative impact on environmental, economic, or public welfare priorities. Appendix B. is a list of invasive species found in Delaware.

Mapped Wetland – the wetland that has been chosen for the study.

Pannes – a bare, exposed or water-filled depression in the marsh that often exhibit elevated salinity levels.

Pools – a depression within the marsh that may hold water at low tide.

Reference Site - a site that was selected based on a qualitative ranking of low, medium, or high.

Spoil pile – pile of earth and rock that was excavated or dredged.

Tidal creek – a small, tidally influenced tributary connecting to the mainstem of the river.