Quality Assurance Project Plan (QAPP) for the Assessment of Salt Marsh Elevation Enhancement Projects Using Sediment Placement

EPA WPDG # CD00A01323

EPA QA Tracking # 25156

June 24, 2025

State of Rhode Island
Department of Environmental Management
Office of Water Resources and
Narragansett Bay National Estuarine Research Reserve

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David Gregg, Ph.D., RI Natural History Survey Project Contract Manager, (401) 874-5800

Signature/Date 6/26/2025

A3. Table of Contents

APPENDIX A. Field Methods Manual: US Fish and Wildlife Service (Region 5) Salt Marsh Study (James-Pirri et al. 2002)
APPENDIX B. Properties and Behaviors of Soils On-line Lab Manual, Chapter 3 (Hossain et al. 2021)
APPENDIX C. Soil Quality Test Kit Guide (USDA 2001)

A. PROJECT MANAGEMENT

A3. Distribution List (EPA QA/R-5 A3)

- Signatories (Title Page) (EPA QAPP Standard A1)
    - Michele McCaughey, michele.mccaughey@dem.ri.gov
    - Elise McNally, mcnally.elise@epa.gov
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- EPA Region 1, New England Wetlands Monitoring Coordinator, Beth Alafat, alafat.beth@epa.gov
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- Rhode Island Coastal Resources Management Council (CRMC), Leah Feldman, Policy Analyst, lfeldman@crmc.ri.gov
- Policy Analyst, crmc.ri.gov
- Rhode Island Natural History Survey (RINHS), Wetlands Scientist, Thomas Kutcher, tkkutcher@rinhs,org

A4. Project Organization (EPA QA/R-5 A4) (EPA QAPP Standards A8 and A10)

Henceforth, the Department of Environmental Management (DEM) Office of Water Resources and the Narragansett Bay National Estuarine Research Reserve (NBNERR) will be the lead agencies to co-manage and implement this project. DEM has contracted with the RI Natural History Survey (RINHS) and the RINHS has recruited and hired staff. Qualified and experienced personnel are available to execute the work. The following people will administer and conduct this work as outlined below and depicted in Figure 1.

- Michele McCaughey, Environmental Scientist III, DEM Office of Customer and Technical Assistance (OCTA) - Quality Assurance Manager;
- Elise McNally, EPA Region 1 - Quality Assurance Reviewer;
- Donna Smith-Williams, EPA Region 1 - Wetland Program Development Grant Project Officer and primary point of contact;
- Susan Kiernan, Administrator, DEM Office of Water Resources – DEM Program Manager, Responsible for contract agreement and fiscal grant management, and general program oversight;
- Caitlin Chaffee, Reserve Manager, NBNERR – Responsible for project management and communication;
- Kenneth Raposa, PhD., Research Coordinator, NBNERR – Project Quality Assurance Officer. Experienced salt marsh research ecologist with over 30 peer-reviewed publications, salary-funded externally to this QAPP. Responsible for data review, quality assurance as described

below, report writing, supervision of field staff, and other duties consistent with this QAPP;
- David Gregg, RINHS, Executive Director – Responsible for contract management and supervision of RINHS staff; and
- Thomas Kutcher, Wetland Scientist, RINHS – Responsible for field and office data collection, entry and analysis, QAPP and report writing, and supervision of field staff.

Project Quality Assurance Manager Independence

The DEM Quality Assurance Manager (QAM) is a salaried state employee working independently of the environmental information operations subject to this QAPP. The QAM will not be involved with the program or project management of the Project.

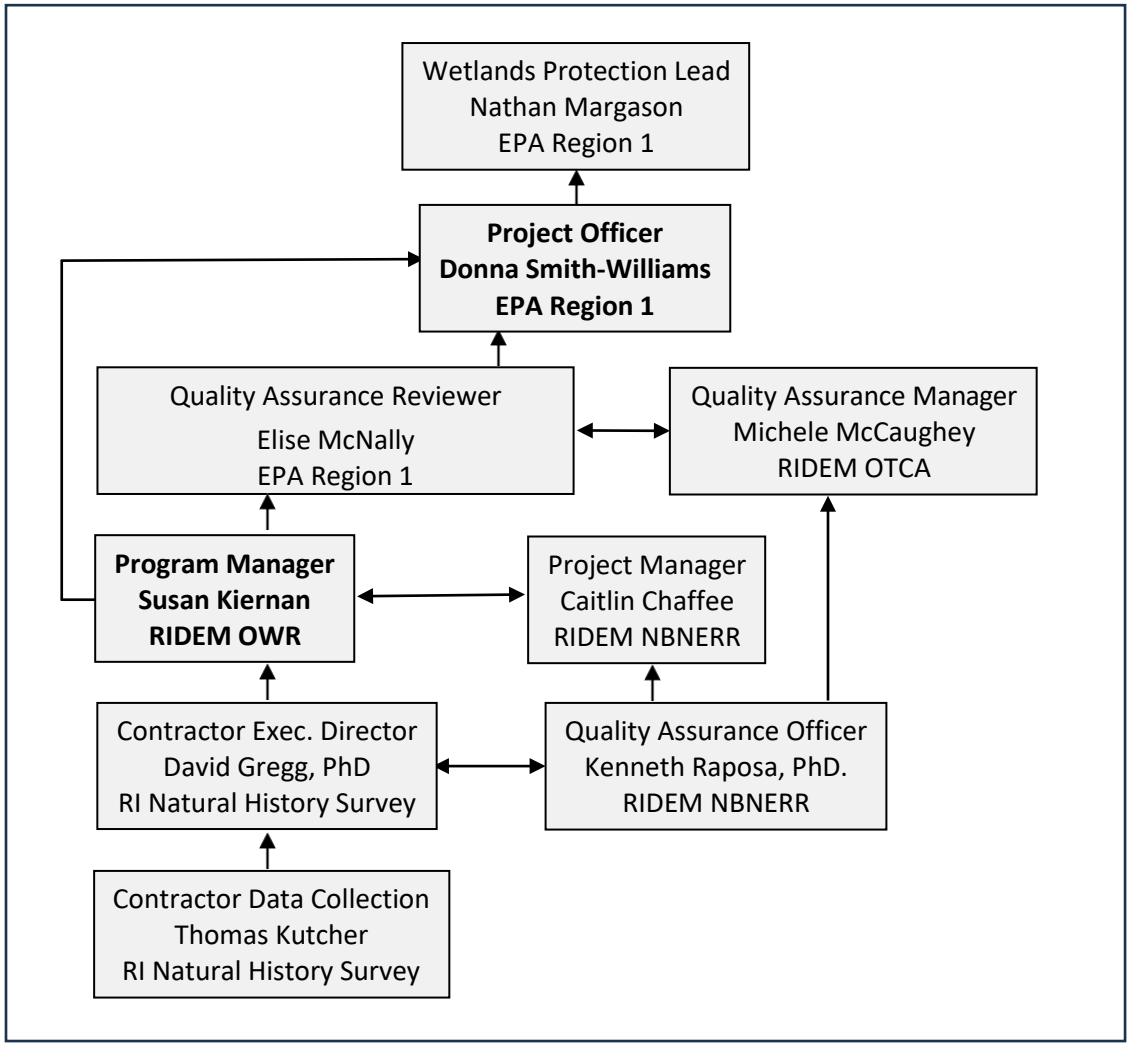

Figure 1. Organizational Chart for Project management and quality assurance.

A5. Problem Definition/Background (EPA QA/R-5 A5) (EPA QAPP Standard A4)

Background

Salt marshes perform a host of functions that are valuable to people and wildlife, including water pollution filtration and uptake, protection from riverine and coastal flooding, provision of critical habitat

for fish and wildlife, and recreation and aesthetics. Recently, efforts have been made to improve marsh function through the beneficial re-use of dredge sediments placed on the marsh surface to enhance elevation and promote rigorous plant growth. Four sediment-placement (hereafter, SP) projects have been conducted in Rhode Island in the past decade and another is in planning. DEM and NBNERR recognize the importance of monitoring and assessment to inform the management, conservation, and enhancement of salt marshes.

This QAPP details the quality assurance procedures for a project (hereafter, the Project) that involves the development of a multi-metric sediment-placement index (hereafter, SPI) that can be used to assess the outcomes of SP enhancements and, importantly, investigate the mechanisms affecting variability in the favorability of SP outcomes, so that managers can use informed adaptive management to improve outcomes in future SP interventions.

A6. Project/Task Description and Schedule (EPA QA/R-5 A6) (EPA QAPP Standard A5)

The Project will assess the utility of several metrics to indicate environmental factors that can characterize SP outcomes. SP enhancement projects typically aim to promote the re-establishment and vigor of salt marsh vegetation and the functions that rely on healthy marsh vegetation, such as support of salt-marsh-dependent fauna. Revegetation with native salt-marsh species is the keystone indicator of SP success and can alone be used to assess *how* successfully an SP project is developing; but other factors are needed to evaluate *why* an SP-treated marsh is or is not rigorously revegetating. We will use existing and new data to develop metrics based on existing and new monitoring parameters (Table 1). The metrics will be based on ranges and thresholds of monitoring parameters that either support or do not support the propagation and recruitment of native salt marsh vegetation, as determined through research of peer-reviewed literature and through empirical findings of the data collected by this Project. Metrics will be assigned coefficients based on those ranges/thresholds and their suitability for supporting vegetation regrowth. The SPI is expected be flexible to utilize data available for a given project, thus, it is expected that metrics scores will be averaged to provide an overall SPI index, if desired.

The SPI is intended to act as a set of metrics that can be interpreted individually or in aggregate (through simple averaging) to assess which parameters have been made suitable for biological recovery (mainly vegetation, but also avian, nekton) by SP restoration activities. It is expected that some metrics may be correlated with others in the SPI, as the physical and biological properties of salt marshes oftentimes interact. To address the potential for collinearity of metrics to affect SPI interpretation, each metric will be assessed against vegetation recovery separately, rather than by using multiple regression (which can be confounded by collinearity). Aggregating the metrics through averaging will also reduce the influence of collinearity on the SPI. Additionally, pairs of metrics will be analyzed using correlation and other analysis, as needed, and any metrics determined to be redundant, unnecessary, or misleading will be further assessed for their utility and efficiency in the index.

Existing and new data based on existing parameters and methods will use EPA-approved quality assurance procedures detailed in an earlier approved QAPP titled *Quality Assurance Project Plan (QAPP) for Nine Salt Marsh Monitoring and Assessment Methods**,** applicable July 2024 through July 2029* (EPA QA Tracking # QA24157) (DEM 2024) (hereafter the "Methods QAPP"), available at: https://dem.ri.gov/sites/g/files/xkgbur861/files/2025-06/ninemethod-saltmarsh-qapp-24157.pdf. Procedures for data collected using new methods developed specifically for this Project are detailed below.

Table 1. Sampling parameters to be piloted for the SP-Assessment Index (SPI) with references to the methods, procedures, and relevant quality assurance project plans.

#	Sampling Parameter	Method Source	QAPP Reference
1	MarshRAM rapid assessment	Kutcher 2022	Methods QAPP 2024
2	Vegetation composition and plant height	James-Pirri et al. 2002	Methods QAPP 2024
3	Avian species composition and abundance	www.tidalmarshbirds.org	Methods QAPP 2024
4	Nekton species composition and abundance	James-Pirri et al. 2002	Methods QAPP 2024
5	Marsh surface elevation and accretion	Callaway et al. 2013	Methods QAPP 2024
6	Soil shear strength	Turner 2011	Methods QAPP 2024
7	Height and duration of tidal water	James-Pirri et al. 2002	Methods QAPP 2024
8	Soil porewater salinity	James-Pirri et al. 2002	This QAPP
9	Soil Texture	Wentworth 1922	This QAPP
10	Soil Infiltration Rate	USDA 2001	This QAPP

Metrics for the SPI will be developed and applied at four recovering SP marshes located in Rhode Island (Table 2). The data collection will occur during the growing season (June through October). Method development will entail researching literature to determine thresholds for each parameter (Table 1) that would indicate poor to excellent conditions for vegetation propagation and survival (i.e., recovery), applying those thresholds to each metric as coefficients, and testing the metrics on the SP marshes by applying them and analyzing the results to determine whether categories are indicative of vegetation recovery.

Table 2. Information about four recovering SP-enhanced salt marshes in Rhode Island being assessed using the SPI in this Project. USFWS (United States Fish and Wildlife Service); RICRMC (Rhode Island Coastal Resources Management Council).

SP Marsh	Latitude	Longitude	Lead Agency	SP Area (Ha)	First Year of SP
Sachuest Point	41.4849	-71.2452	USFWS	4.8	2016
Narrow River	41.4552	-71.4496	USFWS	14	2019
Ninigret	41.3606	-71.6416	RICRMC	15	2017
Quonochontaug	41.3350	-71.7210	RICRMC	25	2019

A7. Quality Objectives and Criteria for Measurement Data (EPA QA/R-5 A7)

Measurement data will be collected with the intent of characterizing parameters for analysis against data collected at an earlier time or against sets of data from other treatments (such as comparing salt marshes to each other). The quality objective is that samples accurately represent actual conditions adequately to run statistically-valid analyses elucidating trends or categories that can inform management (*representativeness*). The methods are measured as described in sections below. Quality criteria are that the collection and handling of data ensure *precision* and minimize user *bias* to produce consistent and reliable results. Qualified field personnel will conduct these methods as designed and reported in peer-reviewed and published literature. Through communications with field personnel and review of the data, the Project Quality Assurance Officer will verify that data were collected properly.

A7.1 Precision, Bias, and Representativeness

Precision, *bias*, and *representativeness* are integrated in the sampling designs of each of the sampling methods of the Project. Precision, bias, and representativeness for sampling methods 1 through 6 (Table 1) are covered in the Methods QAPP (RIDEM 2024). Precision, bias, and representativeness for methods for height and duration of tidal water (7), soil porewater (8), and soil texture (9) are addressed below:

7. Height and duration of tidal water

   Water levels are recorded using Onset® Hobo® self-contained water-level loggers according to James-Pirri et al. (2002; Appendix A) and manufacturer's directions. Water levels will be monitored at eight groundwater wells per marsh, each located equidistantly along a linear transect running from the water's edge to the upland interface. Each transect (one per marsh by eight wells per transect) will be located in an area selected by a team of qualified salt-marsh ecologists to be *representative* of marsh-wide outcomes of SP enhancement in terms of vegetation recovery and elevation. *Precision* is dependent upon factory calibration of the water-level loggers, which is reported by the manufacturer to be ±0.05% typical and ±0.1% maximum, and careful measuring by the field staff. Only qualified field staff will deploy the loggers and take field measurements.

8. Porewater Salinity

   Precision, *bias*, and *representativeness* for the vegetation sampling methods are addressed in James-Pirri et al. (2002; Appendix A), *Soil Salinity*, pp. 15-17; where sampling techniques and data handling are designed specifically to ensure adequate *precision* and minimize user *bias* for analysis. Several prior studies have shown that following these methods results in adequately precise, unbiased, and representative data for conducting meaningful change analysis (e.g., Roman et al. 2002, Raposa et al. 2017). Representativeness will be assured through the distribution of sampling stations, where one salinity sample will be collected at each groundwater-well station spatially distributed as described above in Method 7, *height and duration of tidal water*.

9. Soil Texture

   Precision, *bias*, and *representativeness* for soil texture analysis will be assured through sample-collection design and standardization of analysis procedures as described following. *Representativeness* will be assured through the distribution of sampling stations, where one soil sample will be collected at each groundwater-well station as described above in Method 7, *height and duration of tidal water*; and further assured through standardized sampling to 15cm using a standard soil auger to collect the samples. *Precision* and *bias* will be addressed through following the procedures detailed in the *Properties and Behaviors of Soils On-line Lab Manual*, Chapter 3 (Hossain et al. 2021; Appendix B), except as noted, following. Soil sieves consistent with Wentworth (1922) classification methods, as indicated in Figure 1, will be used to increase relevance for vegetation support. Soils will first be run through a #5 standard sieve to remove particles larger than 4mm and plant roots, which will not be considered in the soil analysis. Each sample will then be standardized to a 100g for further sieving and analysis. Sieve mesh sizes used will be #10 (2mm), #18 (1mm), #35 (0.5mm), #60 (0.25mm), #120 (0.13mm), and 230 (0.06mm) (Wentworth 1922). Silt and clay fractions (<0.06mm) will not be further analyzed. Organic content will be assumed to be negligible (zero), as an organic fraction has had little time to develop in the SP sediments.

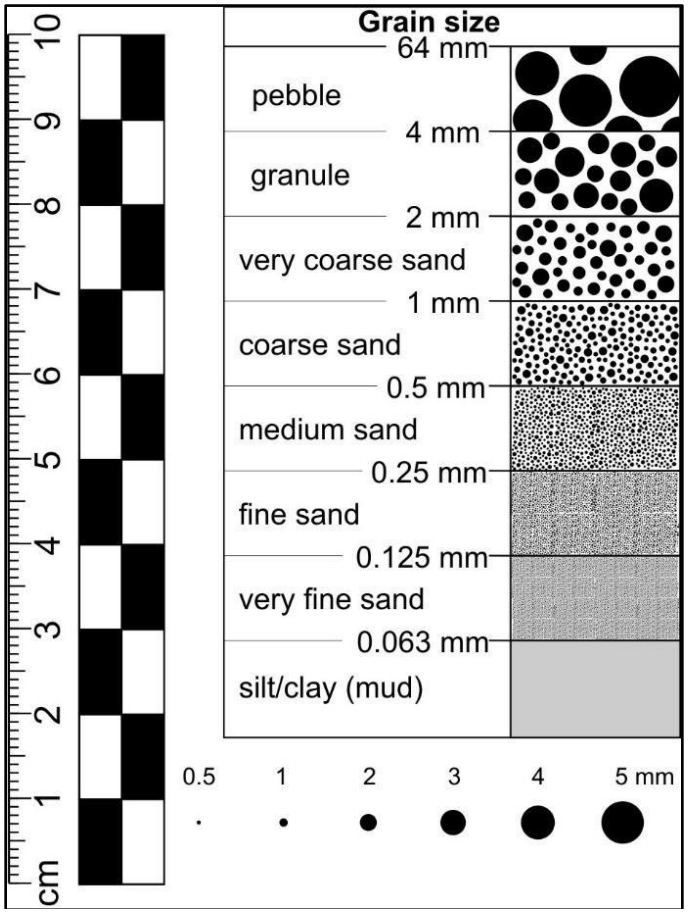

Figure 1. Soil grain sizes from Wentworth (1922) to be analyzed in this Project.

10. Soil Infiltration Rate

Precision, *bias*, and *representativeness* for soil texture analysis will be assured through sampling design and standardization of analysis procedures as described, following. *Representativeness* will be assured through the distribution of sampling stations, where one soil-infiltration sample will be conducted at each groundwater-well station as described above in Method 7, *height and duration of tidal water*; and further assured through standardized sampling following USDA (2001). *Precision* and *bias* will be addressed through following the standardized procedures detailed USDA (2001; Appendix C), except: saltwater sourced on-site will be used in the test to mimic tidal-water infiltration; soils will be primed with 2cm of saltwater prior to running the two infiltration tests to improve *precision*; and tests will be run from a starting water depth of 5cm and ending at 2cm or 20 minutes, whichever comes first, to increase *precision* over NRCS methods.

A7.2 Completeness, Comparability, and Sensitivity

All data collection will pursue *complete* application of each method. Data that are deemed to be incomplete and thus unusable or unsuitable for analysis by the Project Quality Assurance Officer will be discarded. Prior applications of each of the methods covered in this QAPP indicate adequate *sensitivity*, and suitable *comparability* with prior data or with data across sampling treatments (such as for *height and duration of tidal water* and *salinity*, Raposa et al. 2022, etc.). It is expected that following the methods described in Sec. B1-2-4 will generate data with similar comparability and sensitivity as the

data generated using these same methods in prior studies.

A8. Special Training Requirements/Certification (EPA QA/R-5 A8)

Only trained and experienced scientists will conduct the monitoring and assessment methods covered in this QAPP. NBNERR and RINHS monitoring and assessment partners have been instrumental in developing and testing most of the methods detailed in this QAPP, and trained, qualified personnel will conduct the work. Kutcher (Subawardee, data collection) has nearly two decades of experience conducting the assessment methods covered in this QAPP and will, as needed, provide training to any untrained technicians collecting data. Kenneth Raposa, PhD, has been the Research Coordinator for the Narragansett Bay National Estuarine Research Reserve for over 20 years and has published numerous studies using *height and duration of tidal water* and *salinity* sampling methods covered under this QAPP. Kutcher and Raposa both have extensive scientific field and laboratory experience adequate for following procedures for soil sampling and analysis as detailed in cited sources (Wentworth 1922, Hossain 2021, USDA 2001). Training of technicians for any Project method will be conducted by demonstration of the method in the field, and observation, by the trainers, of the trainee consistently performing the method to the standards detailed in this QAPP. An email will be submitted to the QAO documenting the training and the performance of the trainee for each method the trainee will conduct.

A9. Documents and Records (EPA QA/R-5 A9)

The format for all data reporting packages will be consistent with the requirements and procedures used for data validation and data assessment described below. This QAPP will be reviewed regularly by the Project Manager and Quality Assurance Officer, to confirm that it remains current and accurate, and is effective at meeting the Project and data quality objectives.

A9.1 QA Project Plan Distribution

This QAPP will be distributed to all appropriate persons within DEM OCTA, Office of Water Resources, NBNERR, CRMC, EPA Region 1, and the RINHS as identified in section A3 of this QAPP. It will also be posted on the DEM web page for Environmental / Quality Assurance Project Plans @ https://dem.ri.gov/data-maps/data.php#quapps and may be posted on the NBNERR web page @ http://www.nbnerr.org.

A9.2 Field Documentation and Records

Field data will be hand-recorded by completely filling out paper field forms specific to each method, including the date of the field observations and the identity or the person making the observations; waterproof paper will be used in rainy weather. Salinity and soil field data will be housed at the RINHS office until analysis and reporting are complete, whereas water-level field data will be housed at NBNERR until analysis and reporting are complete. All field data forms will then be transferred to NBNERR to be held as detailed below. Daily activities (e.g., location, mileage, assessments) of field staff will be documented in diaries, timesheets, and logs as required by EPA grant conditions on relevant OMB circulars cited therein and held by the RINHS.

A9.3 Laboratory Documentation and Records

Field data will be entered into an electronic spreadsheet (Microsoft Excel) at RINHS or NBNERR, where they will be housed on a hard drive and the “cloud” (Google Drive or similar) until analysis and reporting are complete (see timeline), at which time they will be transferred to NBNERR along with any secondary and derived data. The Project will implement proper document control procedures consistent with DEM's Quality Management Plan (revised Nov. 22, 2022). The NBNERR Project Quality

Assurance Officer will have ultimate responsibility for any and all changes to records and documents after submittal to NBNERR and shall be responsible for their retention and storage. The RINHS will copy all final Excel spreadsheet files and GIS shapefiles collected under this QAPP to the DEM Office of Water Resources Program Manager or assigned representative for permanent retention under DEM Record Series 6.13.4.

The DEM Program Manager and the NBNERR Project Quality Assurance Officer shall retain the final approved QAPP and all updated versions and will be responsible for any distribution of the current version. The DEM Program Manager shall retain copies of all contract- and grant-management documents, and the NBNERR Project Quality Assurance Officer shall retain all draft and final reports, memoranda, and technical correspondence between NBNERR and all project personnel. The RINHS will copy all draft and final reports and memoranda produced under the QAPP to the DEM Office of Water Resources Program Manager or assigned representative for permanent retention under DEM Record Series 6.13.4

Records and documents that will be produced in conjunction with this QAPP may include:

- Completed field forms and site maps
- Excel spreadsheet files for data storage and analysis
- GIS shapefiles of assessment or study areas
- This QAPP
- Draft project reports and appendices
- Final project reports and appendices

Storage of project information

Files, paper and electronic records, and other media such as incidental photographs will be maintained by the NBNERR for a minimum of three (3) years after the completion of this work and delivery of RINHS products to NBNERR. After three years, some records may be moved to the NBNERR Records Archives for storage in accordance with relevant NBNERR record retention policy. All field data forms will be retained by NBNERR permanently. The DEM Office of Water Resources shall also retain records of project deliverables and grant management associated with projects funded by EPA to the Office of Water, pursuant to its records retention schedules. As it is anticipated that wetland assessment will continue indefinitely, the time frames stated are the minimum and likely will be exceeded as the information will be needed for the ongoing program.

Backup of electronic files

Electronic files will be maintained on the NBNERR network server, as well as periodically backed up locally by the NBNERR Project Quality Assurance Officer on CD’s, demountable hard drives, solid state digital storage devices, or the internet "cloud".

A9.4 Bi-annual and/or Final Reports

The draft and revised final reports will be provided to DEM and NBNERR by RINHS. Results of data collected will be documented and reported as follows:

- a detailed outline of methods employed;
- data analyses and demonstrations as listed in section 1.6 and detailed below;
- a site map depicting assessment unit locations; and
- tables and figures as necessary to illustrate the work, analyses, and results.

The NBNERR and project participants and partners will provide written comments on a draft report for any given project. A final report, which will be completed by RINHS and/or other project participants, will incorporate responses to revisions based on the NBNERR or other comments.

B. DATA GENERATION AND ACQUISITION

B1-2-4 Sampling Design, Methods, and Analysis (EPA QA/R-5 B1, B2, and B4) (EPA QAPP Standards B1 and B2)

Sampling design, methods, and analysis for sampling methods 1 through 6 (Table 1) are covered in the existing Methods QAPP (DEM 2024), whereas methods 7 through 10 are detailed below. Target sampling intensities for each method in the SPI will be based upon ongoing monitoring methods already in place, and on maintaining or improving the accuracy or precision of those methods. The expected sampling intensity for each parameter is outlined in Table 3.

Table 3. Sampling intensity for each parameter that may be included in the SPI.

#	Sampling Parameter	# Stations per Marsh	# Surveys per Year
1	MarshRAM rapid assessment	1	1
2	Vegetation composition and plant height	20	1
3	Avian species composition and abundance	2-3	2
4	Nekton species composition and abundance	20	2
5	Marsh surface elevation and accretion	2-4	1
6	Soil shear strength	8	1
7	Height and duration of tidal water	8	One 28-day deployment
8	Soil porewater salinity	8	1 or more
9	Soil Texture	8	1
10	Soil Infiltration Rate	8	1

7. Height and Duration of Tidal Water Methods

   Tidal height and duration will be monitored according to James-Pirri et al. (2002). Water levels will be recorded using Onset® Hobo® self-contained water-level loggers according to manufacturer's directions (available at: https://www.onsetcomp.com/sites/default/files/resources-documents/12315-J%20U20%20Manual.pdf). Eight units will be deployed in each marsh (as detailed in Sec. A7.1.7) during the peak of the growing season for four continuous weeks. The loggers will be programmed to collect one reading every 15 minutes, and lowered into perforated and capped 2" PVC pipe following methods referenced in James-Pirri et al. (2002; Appendix A). Depth of logger in relation to marsh surface will be measured to the nearest mm. Recorded depths will be uploaded to spreadsheet and analytical software directly from the logger at monthly intervals. The depths recorded on the logger will be adjusted to levels relative to the marsh surface, using subtraction, to determine flooding frequencies and water depth.

8. Porewater Salinity Methods

   Porewater salinity will be measured following the methods of James-Pirri et al. (2002; Appendix A). The method consists of using a thin stainless-steel tube and a syringe to extract soil water from the marsh soil and filtering the extracted porewater through a coffee filter and onto the lens of a hand-held refractometer, which is then read and the data recorded.

9. Soil Texture Methods

   A 5-cm diameter by 15-cm deep soil sample will be collected adjacent to each groundwater well (see Sec A7.1.7) using a Dutch auger. The sample will be placed into a zip-lock bag and labeled with the site location, date, time, and well number. Samples will be refrigerated until they can be processed and analyzed. Processing entails drying the samples at 105 degrees C for 20 hours, homogenizing the soils by stirring and shaking, reducing the sample to 100g for further processing and analysis, breaking up soil aggregates with a wooden pestle, running them through a series of soil sieves as detailed in Hossain et al. (2021), weighing the soils retained by each sieve using a professional-grade scientific digital scale accurate to 0.1g, and determining the soil fractions by dividing the retained soils by the total sample size for each sieve for each sample.

10. Soil Infiltration Methods

    Soil infiltration rate testing will largely follow USDA (2001). A 6-inch diameter by 15-cm deep steel ring will be used for soil infiltration testing. A measuring tape will be fixed permanently to the ring to ensure precision. The ring will be pounded into the soil to a depth of 2.5 cm. On-site salt water will be poured into the ring over a diffuser (to minimize soil disturbance) to a depth of 2.5 cm and allowed to fully infiltrate to prime the soil. A timed infiltration test will follow, in which the ring will be filled with 5cm of water and timed until the level has reached 2cm in depth or until 20 minutes have passed, in which case the final depth of water will be recorded at the 20-minute mark. Infiltration rate will then be calculated as the change in water level (cm) over time elapsed. The timed infiltration test will then immediately be repeated, and the infiltration rates for the two tests will be averaged to produce a final infiltration rate. If the infiltration rates between the two tests differ by more than 10%, a third test will be run, and the average of the two tests in closest agreement will be used as a final infiltration rate.

B3. Sample Handling and Custody (EPA QA/R-5 B3) (EPA QAPP Standard B3)

Soil samples will be collected and stored in zip-lock bags labeled to identify the soils as described in Sec B1-2-4.9. Samples will be transported, by the sampler (Kutcher), directly to RINHS to be refrigerated until they can be processed and analyzed. Because the soil samples will be transported directly to the storage facility after collection, and the organic fraction is expected to be negligible and therefore will not be measured, the samples will be transported at ambient temperature. Heat-resistant labels for each sample will be inserted into each sample's drying container prior to drying, and additionally, the layout of the samples in the drying oven will be documented in case a label is lost. Samples will thereafter be processed and analyzed one sample at a time to avoid labeling mistakes. No other methods for the Project require physical sample collection.

B5. Quality Control Requirements (EPA QA/R-5 B5)

RINHS and NBNERR will work together to ensure that all sampling and assessment activities are conducted within the criteria set for the project, specifically as described in the above sections.

B6. Instrument/Equipment Testing, Inspection, and Maintenance (EPA QA/R-5 B6)

All equipment will be inspected for proper functionality prior to each use.

B7. Instrument/Equipment Calibration and Frequency (EPA QA/R-5 B7)

7. Height and Duration of Tidal Water Methods

Onset® Hobo® self-contained water-level loggers are calibrated by the manufacturer and no user calibration is possible.

8. Porewater Salinity

The refractometer will be calibrated with distilled water prior to each field day and adjusted to a precision within one part per thousand (ppt). If the refractometer cannot meet that precision, it will be replaced with a properly-functioning unit. For any field reading outside of expected salinities (0 to 50 ppt.), an additional reading using a second refractometer will be taken, and the readings will be accepted and averaged if they are within 3 ppt. of each other. Otherwise, both units will be recalibrated in the field and the readings will be taken again. If the readings are still not within 3 ppt. of each other, they will be discarded.

9. Soil Texture

The scientific scale will be calibrated using the zero function before each measurement, and calibrated using a known mass (100g) before each analysis day.

10. Soil Infiltration Rate

No equipment will require calibration.

B8. Inspection/Acceptance Requirements for Supplies and Consumables (EPA QA/R-5 B8) (EPA QAPP Standard B6)

Not applicable. No critical consumables will be used.

B9. Data Acquisition Requirements (Non-Direct Measurements) (EPA QA/R-5 B9)

Geospatial data from the Rhode Island Geographic Information System (RIGIS, available: www.rigis.org) may be used for generating site maps and locating monitoring stations, as detailed in the above sections of this document. RIGIS data are thoroughly quality assured geospatial data, meet FGDC mapping standards, have standardized metadata, and are widely used by State, Federal, and local scientists conducting geospatial analysis in the State of RI.

B10. Data Management (EPA QA/R-5 B10)

Field data will be collected and stored in a metal file cabinet in a locked office in the RINHS or at the NBNERR. All data will be transposed to electronic format in the form of Excel spreadsheet files. The Excel files will be coded by date and corrections to the file will be coded by the revision or correction date followed by the suffix *correction*. Duplicate versions of the datasets will be coded specifically for analysis and kept in a separate folder. Analysis versions will also be coded by date with each use. Baseline and analysis data files will be stored in the RINHS laboratory at East Farm, URI, Kingston, RI on a hard drive and on the internet "cloud" (e.g., Google Drive). Any GIS data will be stored in file folders as shapefiles, which will be housed on two separate hard drives within the RINHS.

Field and electronic data will be quality checked for errors by qualified personnel following data collection and following data upload into Excel. Any corrections will be handled as noted above. The Wetland Scientist will be responsible for data management until the data are transferred to NBNERR at

the end of the analysis and reporting period, at which time the Project Quality Assurance Officer will be responsible for the data.

C. ASSESSMENT AND OVERSIGHT

C1. Assessments/Oversight and Response Actions (EPA QA/R-5 C1) (EPA QAPP Standard C1)
Project oversight will be provided through regular correspondence between the NBNERR Project Quality Assurance Officer and RINHS no less than once per month. Correspondence will be in the forms of email and telephone correspondence, review meetings, memoranda, and the exchange of key data and documents. Assessment-oversight will involve review of all aspects of the Project and its progress. Technical advisors, who may include academic experts, state scientists, EPA scientists, and other expert stakeholders may also be consulted throughout the Project. NBNERR and RINHS will respond to input as necessary to ensure the efficient use of Project resources in evaluating the effectiveness of salt marsh assessment tools to meet state reporting requirements and SMMAS objectives (Raposa et al. 2016).

The Project Quality Assurance Officer has the authority to issue a stop work order if something is not going right and to document corrective actions that need to be taken. For example, if, upon quality assurance investigations (always done prior to any analysis), the Project QA Officer or Wetland Scientist finds that the data from any instrument (specifically, water-level logger, refractometer, or scale) does not follow prior trends, or otherwise appears erroneous, indicating malfunction or loss of calibration, The QA Officer will discard any erroneous data and immediately take the instrument out of service to be recalibrated by the manufacturer or replaced with a new instrument.

C2. Reports to Management (EPA QA/R-5 C2) (EPA QAPP Standard C2)
Brief bi-annual memoranda and final reports will be submitted by the RINHS to the Project Quality Assurance Officer, the Program Manager or assigned representative, and the Project Manager at the following project milestones: (1) the completion of field work and prior to the initiation of data analysis, (2) the completion of data analysis, (3) the draft Report, (4) the Final Report.
Memoranda may be appended to or incorporated into the draft and final reports. The EPA Project Officer will receive a copy of the final report.

D. DATA REVIEW AND USABILITY

D1. Data Review, Verification, and Validation Requirements (EPA QA/R-5 D1) (EPA QAPP Standard D1)
The validity and utility of data collected in the field is dependent upon (1) the qualifications of the field personnel, (2) the validity of the methodology, and (3) the appropriate analysis and interpretation of the data, which have been addressed in prior sections of this QAPP. Following the methods described in this QAPP and its appendices should assure the quality of the data. Only trained scientists will be able to assess the quality and validity of the data from these methods. The staff analyzing the data are trained to detect errors or inconsistencies in the data through identifying outliers and other unexpected or erroneous behaviors of the data. Data errors and inconsistencies will be highlighted in a copy of the data spreadsheet renamed with the suffix *ERROR* and sent to the Quality Assurance Officer via state email. Data quality will be further assured through the review of data reports submitted to technical peer reviewers from partner academic, state, or federal agencies. The Quality Assurance Officer will ensure that appropriate external peer reviewers review the data outcomes before data reports are finalized.

D2. Verification and Validation Methods (EPA QA/R-5 D2) (EPA QAPP Standard D1)
The data quality will be verified by the Wetlands Scientist and other field partners by reviewing field datasheets and electronic data as described above. The Quality Assurance Officer will be made aware of data inconsistencies and errors as described in Section D1, and will have the power to require data to be discarded or re-collected, as possible, if it does not meet the requirements detailed in Sec. A7.

7. Water-level Loggers
Water-level data will be uploaded directly from the loggers to a computer using the internal logger software. The Quality Assurance Officer will produce and review data trends for inconsistencies and outliers. Any data points flagged as inconsistent against expected or typical outcomes will be reviewed and, as necessary, corrected in the electronic dataset. Any inconsistencies that cannot be corrected through this review process will be discarded prior to analysis, and this action will be documented in data reports.

8-9. Porewater, soil-texture, and soil infiltration rate
Measurement data collected will be recorded by qualified personnel. The data will be aggregated and inspected on field and laboratory datasheets and following upload to electronic spreadsheet software, where the data will be analyzed for validity using statistical and spreadsheet software to review the data for inconsistencies and outliers. Any inconsistencies or discrepancies in outcomes among users or platforms will be investigated and rectified. Data that cannot be rectified as error-free will be discarded and re-analyzed or re-collected, as relevant and possible, and this action will be documented in data reports.

D3. Reconciliation with User Requirements (EPA QA/R-5 D3)
The goals of the Project include testing the applicability of wetland monitoring and assessment methods across a range of conditions, and demonstrating the utility of the methods and resulting data in assessing SP-enhancement outcomes. Because of the complex nature of the data, it is expected that the interpretation and application of the data will be primarily limited to the wetland professionals who requested the work (the users). As such, the data will be collected specifically for the direct use of the intended users. However, it is anticipated and intended that the data will also be used and interpreted for generating reports, graphics, and other outreach materials aimed at secondary and tertiary consumers of the information, such as for salt marsh managers.

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Appendix A

Field Methods Manual: US Fish and Wildlife Service (Region 5) salt marsh study

James-Pirri et al. (2002)

Field Methods Manual: US Fish and Wildlife Service (Region 5) salt marsh study

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(version 2)

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USGS Patuxent Wildlife Research Center
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TABLE OF CONTENTS

LIST OF FIGURES

LIST OF TABLES

INTRODUCTION

Salt marshes are a common ecosystem type within coastal Refuges of the US Fish and Wildlife Service from Maine to Virginia (Region 5). Most of these marshes have been parallel ditched for mosquito control purposes, and to a lesser extent, to facilitate salt hay farming. Although ditching of salt marshes has occurred since Colonial times, most extensive ditching was in the 1930s, with programs to maintain ditches continuing for three decades or more. Documented impacts of parallel ditching on salt marshes include lowered water table levels, drainage of marsh pools and panes, vegetation changes, and associated impacts on habitat support functions for fish, birds, and other trophic components. Recognizing the detrimental impacts associated with ditching, the practice of Open Marsh Water Management (OMWM), considered a more ecologically appropriate mosquito control method, was introduced in the late 1960s.

OMWM is a common practice on coastal Refuges, especially in New Jersey (Forsythe NWR) and Delaware (Prime Hook NWR). In brief, OMWM involves physical alteration of the parallel ditched marsh, through creation of pools and other hydrologic alterations, to establish a marsh that is unsuitable for mosquito egg deposition and larval development, and that promotes establishment of habitats for larvivorous fishes. Traditional OMWM includes plugging of ditches, creation/excavation of ponds within intense breeding areas, construction of radial ditches to facilitate fish access to breeding areas, and other manipulations. As noted, traditional OMWM practices are common at Forsythe and Prime Hook NWR. At more northern Refuges (Long Island Complex, S.B. McKinney, Parker River, and Rachel Carson) a modification of OMWM is being implemented, which is generally limited to ditch plugging, with limited or no pond excavation and radial ditches. The objective of ditch plugging is to re-establish a hydrologic regime on the ditched marsh that is characterized by permanent water on the marsh surface, thereby restoring fish and wildlife habitat functions while controlling mosquito production.

A cooperative research project of the USGS-Patuxent Wildlife Research Center and US FWS-Region 5 was initiated in 2001 to quantitatively evaluate the response of salt marshes to OMWM and associated practices. The objectives of the three year study are to compare parallel ditched salt marshes with OMWM marshes. Specifically, this study is designed to evaluate the effects of OMWM and/or ditch plugging on marsh hydrology (marsh water table levels, soil salinity, and extent of surface water flooding), sedimentation and marsh development processes, vegetation patterns, utilization by nekton (fish and decapod crustaceans) and birds, and mosquito control. Research study sites were established at Rachel Carson NWR (ME) in 1999. Parker River NWR (MA), Long Island Complex (Wertheim NWR, NY), and Prime Hook NWR (DE) were established in 2001. Research will begin at Forsythe NWR (NJ) in 2002 and at Stewart B. McKinney (CT) in 2002/03.

The purpose of this document is to serve as a ***Field Methods Manual*** for the study. Details are provided on how to collect and record the data on appropriate field data sheets. Guidelines on data management, at least with respect to entering data into spreadsheets, are also provided. This manual does not address data analysis techniques. This document is

presented as Version 1 of the Fields Methods Manual. Subsequent versions will include marsh sedimentation methods as well as minor modifications as needed.

GENERAL STUDY DESIGN

This study employs a BACI study design (before, after, control, impact). At each study Refuge we have (or will) selected pairs of sites that include a ditched marsh and a control marsh. The ditched marsh and control marsh are sampled for one year prior to any OMWM activities (Before). Then in year two, OMWM is performed on the ditched marsh and sampling proceeds (After). In the BACI design, the practice of OMWM is the "impact." With this kind of study design it is possible to evaluate, with a degree of statistical certainty, the initial response of the marsh to OMWM. Continued monitoring in successive years will track the long-term response of the marsh to OMWM. It is important to monitor the control marsh simultaneously with sampling the ditched/OMWM marsh. If after OMWM a particular parameter such as water table level changed, and water level did not change in the control marsh, then it could be suggested with some degree of certainty that the change in water table was due to the OMWM and not some other factors. Inclusion of a control marsh serves to document any changes that are occurring in response to regional or local factors that are independent of OMWM manipulations.

To date, the following study sites have been established. Experimental refers to the OMWM or ditch-plugged marsh. Control and Experimental study areas vary in area, ranging from 0.6ha to 12.2ha.

Rachel Carson NWR

- Granite Point Rd. Marsh (control and experimental)
- Moody Marsh (control and experimental)
- Marshall Point Rd. Marsh (control and experimental)

Parker River NWR

- Control
- Experimental B1
- Experimental B2
- Experimental A

Long Island Complex

- Wertheim East experimental and Smith County Park control
- Flanders (control and experimental)

Forsythe NWR

- Oyster Creek, Atlantic County (control and experimental)
- AT&T, Ocean County (control and experimental)

Prime Hook NWR

- Petersfield Ditch (control and experimental)
- Slaughter Beach (control and experimental)

VEGETATION

SUMMARY

The salt marsh vegetation monitoring protocol recommends sampling vegetation community composition and abundance with permanent plots using the point intercept method. At least 20 vegetation plots are required per study area to detect differences in community composition and abundance. Permanent quadrats should be arranged in transects and spaced a minimum of 10-20m apart to maintain independence. Transects should be randomly located within each study area with the first permanent plot randomly located and subsequent plots systematically placed along each transect. Vegetation is sampled once during a season, usually at the end of the growing season (late summer or early fall) when plants are easily identifiable. At least 2 people are required to sample vegetation, one to place the bayonet and the other to record data. It is estimated that 2 people, who are familiar with salt marsh plant identifications, can sample approximately 10-20 plots per day. This does not include time spent re-locating plots that have lost their stakes. Details of the salt marsh vegetation monitoring protocol, including justification of the suggested field method and sample size, are found in Roman *et al.* (2001).

Materials for Site Selection and Location of Vegetation Plots

- Oak stakes to mark vegetation plots
- Mallet to pound stakes into ground
- Black permanent markers to mark transect and plot number on stakes
- Compass
- Meter tape (preferably 100m long)
- Random number table
- Aerial photos of study sites
- Draft map of study site showing boundaries of study areas and approximate location of transects

Site Selection and Sample Location

- Define boundaries of the control and experimental areas.
- Systematically divide each study area into segments to adequately sample the marsh. Segmentation is done to insure dispersion of the vegetation plots throughout the study area. Usually, a study area is segmented into 3 or 4 similarly sized areas.
- Randomly locate transects that traverse the main gradient (*e.g.*, elevation) from creek bank to upland edge of the marsh. The starting point for each transect is randomly located along the creek bank. The random location of the starting point for each transect is selected by measuring the total distance of the creek bank (within each segment) and then randomly selecting points along the bank where each transect will start. These measurements are best done from aerial photography.

- There is no definitive number of transects that should be established per marsh segment, however each transect should be at least 10m apart, to maintain independence of the replicate plots. Transects be dispersed throughout the study areas to ensure that the vegetation plots are representative of the entire study area.
- All transects within a marsh segment should be parallel to each other (*i.e.*, should run along the same compass heading) for ease in re-locating plots.
- Locate vegetation plots along each transect. Regardless of the size of the area a minimum of 20 plots are required for each study area.
- The first plot of each transect is randomly located within the low marsh zone. Measure the width of the low marsh and then place the plot at the distance selected by the random number (0 being on the edge of the bank). For example, if the low marsh zone is 5m wide, a random number between 0-5 would be selected. If there is no discernable low marsh zone, or if the vegetation zone at the creek bank is very broad, (*i.e.*, more than 10m), then the first plot should be located by a random number between 0 and 10.
- After the first plot is located, all subsequent plots are then systematically placed, at least 10m apart, along the length of the transect. The spacing of plots along each transect will be variable depending on the area of the marsh. For example, if the marsh is 8-9 hectares in area, then 4 transects, with 40m spacing between plots along each transect would be appropriate. For smaller marshes, 20m spacing between plots may be necessary. However, all plots should be at least 10m apart to maintain independence of the replicate plots. (refer to Fig 1.).
- Each plot should be marked with stakes labeled clearly with transect and plot number. Construction of permanent stakes is at the discretion of the investigator. Our experience suggests that 4 ft oak stakes (1 ft into the marsh sediment, 3 ft above marsh; about 1^2 inch square) are adequate. Naming convention for vegetation plots is the transect number followed by the distance along the transect where the plot is located. For example, 1-00 would be the first plot on transect 1 (00 meters along the transect), 1-20 would be a plot on transect 1 located 20m from the start of the transect.
- Plot location and distance between plots should be carefully noted on a map so that plots can be re-located in future surveys in the event that the stake is missing. The coordinates (latitude and longitude, UTM coordinates, *etc.*) of all plots should be recorded, preferably with a GPS unit that has sub-meter accuracy.

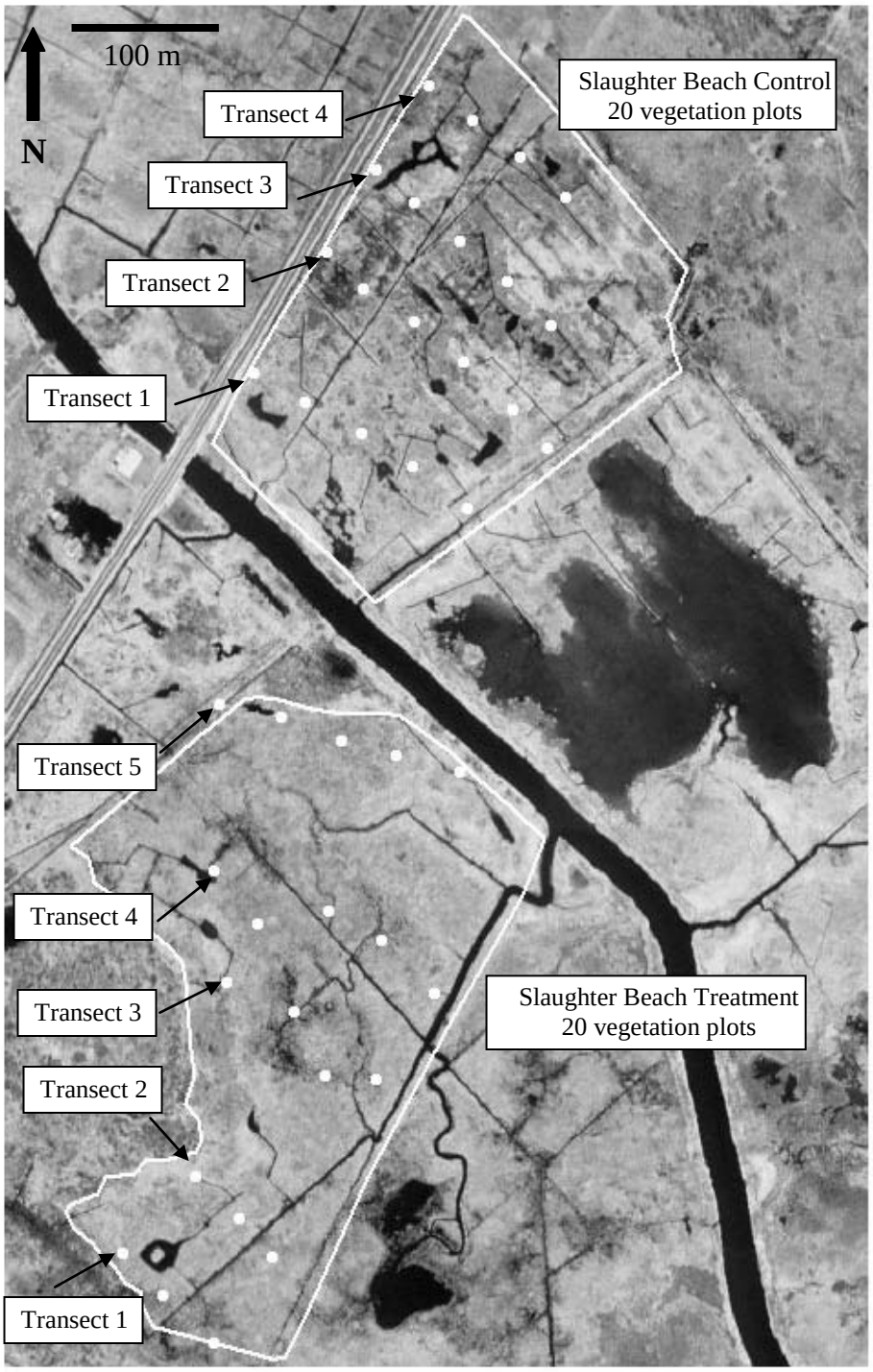


Figure 1. Aerial photo showing location of vegetation plots (white circles) within two study areas at Prime Hook NWR. White line indicates delineation of study areas.

Materials for Sampling Permanent Vegetation Plots

- 1 m^2 quadrat. The quadrat can be made from four 1m lengths of thin diameter (3-5mm) doweling. Dowels should be marked with 10 evenly spaced (11.1 cm apart) increments
- Bayonet or thin rod for point intercept method (less than 3mm diameter).
- Meter stick
- Map of vegetation plots
- Plant identification book
- Plastic bags for voucher specimens
- Data sheet and pencils (Table 1)

Sampling Procedure

- Locate permanent stake marking the vegetation plot.
- In order to sample vegetation that has not been trampled during the establishment of transects, the quadrat is offset 1m from the stake.
- Facing the direction of the transect (from the first plot towards the remaining plots of the transect) set the quadrat 1m to the right of the stake and orient the plot towards the direction of the transect. Be sure to maintain the same offset for all plots and record a detailed description of the offset (Fig. 2). Note: if existing plots are oriented differently that is acceptable. Continue with the existing layout, but be certain to carefully document the orientation with a schematic such as that shown in Fig. 2.

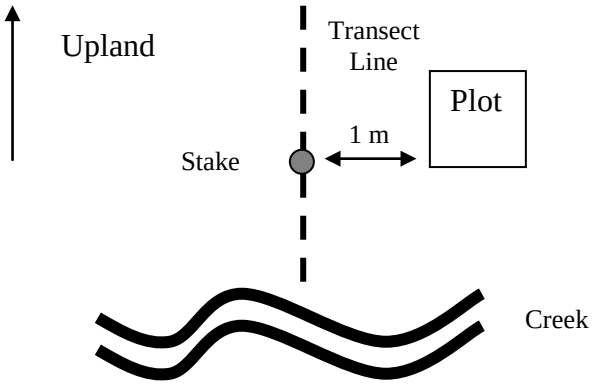
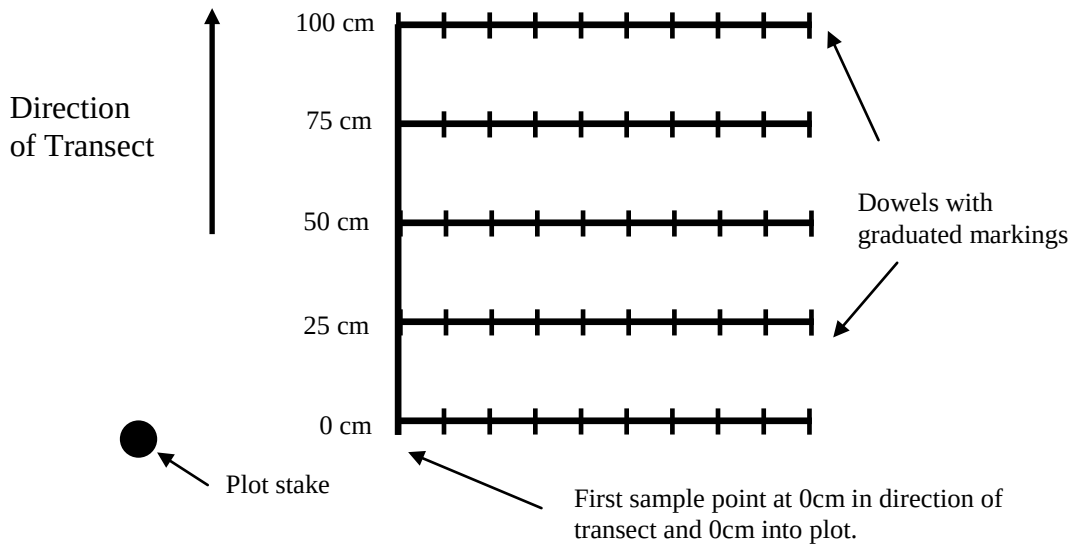


Figure 2. A schematic of the orientation of the sampling quadrat relative to plot stake.

- A meter stick is placed on the marsh surface and then at 0, 25, 50, 75, and 100cm intervals along the meter stick, dowels (≤3mm in diameter) are placed perpendicular to the meter stick. Each dowel is 1m in length and has a total of 10 marks, each spaced 11.1cm apart (Fig. 3). Thus, the $1m^2$ quadrat is divided into a grid of 50 evenly spaced points. In dense vegetation it may be necessary to weave the dowels through the vegetation.
- List all species that are present within the sample quadrat on the data sheet for that plot (a sample data sheet is presented in Table 1).
- Hold a thin rod (≤3mm in diameter) vertical to the first sampling point and lower the rod through the vegetation canopy to the sample point on the ground.
- All species that touch the rod are recorded as a "hit" on the data sheet for that point. Categories other than plant species, such as "water", "bare ground", "wrack or litter," and others are also recorded if they are "hit" by the rod. Table 2 provides definitions of cover type categories that should be included.
- More than one cover type category can touch the rod at each point, and thus multiple hits for each sample point should be recorded if appropriate. However, it is not necessary to count the number of hits for each individual species. For example, if *S. alterniflora* touches the rod in 3 places, it is recorded as one hit of *S. alterniflora* for that point. At least one cover type should be recorded for each point (*i.e.* if there is no vegetation, "bare ground" or "water" may be the appropriate cover type).
- After the first point is completed, the process is repeated for all remaining points on the sampling quadrat until all 50 points have been sampled.
- Tally the total number of hits per species for each plot on the data sheet (Table 1). This can be done after returning to the laboratory.
- All marsh study areas should be sampled within the same time frame (within 1-2 weeks of each other) and occur when the marsh surface is not flooded so that tidal waters do not conceal vegetation.
- This method can also be successfully used in marshes with taller vegetation canopies, like *Phragmites* and *Typha* marshes, or marshes dominated by shrubs (*e.g.*, *Iva*). The observer needs to be careful to use a long rod and to look up to determine if higher vegetation touches the rod.



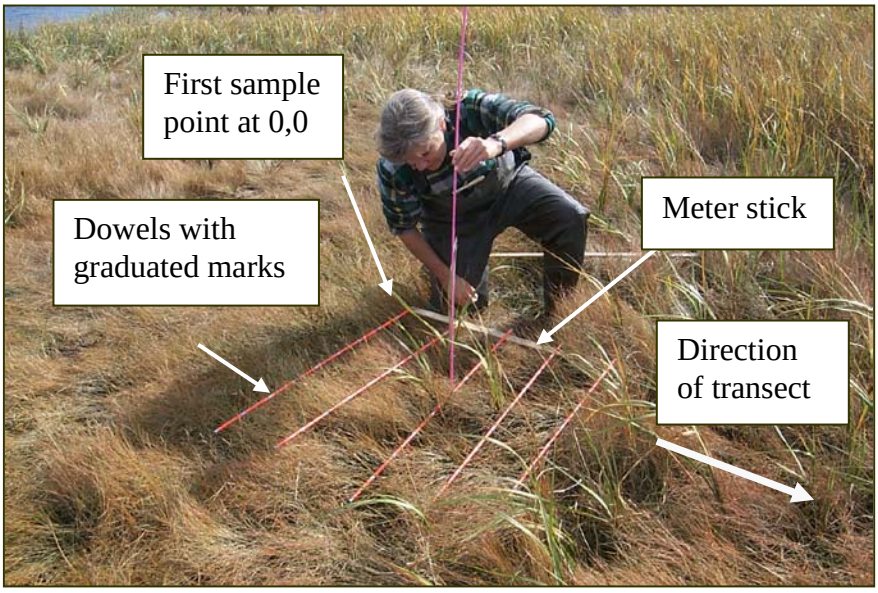


Figure 3. Schematic and photo of the sample quadrat and arrangement of dowels used in the point intercept method.

Table 1. Example of a field data sheet for the point intercept method.

Marsh _____ Field Crew _____ Date _____

Transect & Plot number _____ GPS Coordinates N:_____ E:_____

Record species, first row is for points 1-25, second row is for points 26-50.

		Point																							
SPECIES	Total Tally	1 26	2	3	4	5 30	6	7	8	9	10 35	11	12	13	14	15 40	16	17	18	19	20 45	21	22	23	24	25 50
1.																									
Species # 1 -- pts. 26-50	XXXX																								
2																									
Species # 2 -- pts. 26-50	XXXX																								
3																									
Species # 3 -- pts. 26-50	XXXX																								
4																									
Species # 4 -- pts. 26-50	XXXX																								
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Species # 7 -- pts. 26-50	XXXX																								
8.																									
Species # 8 -- pts. 26-50	XXXX																								
9.																									
Species # 9 -- pts. 26-50	XXXX																								

Table 2. Cover type categories to be included in the point-intercept salt marsh vegetation program.

Live vascular plants (herbaceous and shrubs) identified by species

Standing dead vascular plants identified by species (*e.g., S. alterniflora* dead). This category only includes standing dead (attached) plants that are from a previous year's growth. There may be some dead leaves from this year's growth (*e.g.,* the ends of leaves or leaves that are being replaced by new growth, *etc.*). If you are sure these dead leaves are from the current growing season, then record as live. Dead plant material from a previous growing season is recorded as "liter" (see below).

Macroalgae identified by species. This category generally includes the rockweeds (*e.g.*, *Fucus*, *Ascophyllum*). Microalgae (*e.g.*, diatom mats) and fine filamentous algae are not included in this category.

Bare. Includes mud, sand, microalgae cover, *etc*. These are areas that are not flooded with water and are devoid of standing live, standing dead, or macroalgae. There can be a thin film of surface water within the bare category.

Water. Permanent standing water is identified in plots that are partly within a creek, ditch, marsh pool, or flooded panne.

Wrack/Litter. Wrack is material that has floated into the plot. This is generally dead (not attached) plant material, but could also be trash. Litter is dead plant material that is highly decomposed, if from a previous years growing season, and may or may not be attached. It is not identified to species, as is standing dead (see above).

Trash. Items such as logs, old piers, tires, etc.

Rock. Boulders or rocks can be found on the surface of northern New England marshes.

NOTES:

- If an intercept point has standing water that is covering a bare mud bottom, this point should be recorded as standing water. It is assumed that the bottom is bare and there is no need to record this.
- If macroalgae or submerged aquatic vegetation are hit at the intercept point in a standing water habitat, then both the plant and water should be recorded.
- If a plot is at the edge of a marsh pool (water), *Spartina* overhangs the water, and the intercept point hits the *Spartina* and water, then both *Spartina* and water should be recorded.

WATER TABLE LEVEL

SUMMARY

Water table level provides information on the amount of waterlogging or drainage that is occurring in a marsh. Water table level is an important parameter to use when attempting to understand why vegetation is changing. Water table level is measured using ground water wells. It is recommended that a water table level well be placed in association with each vegetation sampling plot.

For water table level and soil salinity, approximately 20-30 sample stations could be visited within a low tide period. Sampling can be accomplished by one person, but teams of people are always recommended when conducting field work. More details on water table monitoring are found in Roman *et al.* (2001)

Construction and installation of the wells is outlined below. Wells can also be purchased, pre-made, from hydrological supply companies.

Materials for Groundwater Wells

- 1.5 inch (4 cm) interior diameter, schedule 40, PVC Tubes (comes in 10ft lengths and can be purchased at home goods stores)
- PVC caps to fit the tubes. Two caps (rounded preferably) are required for each well
- ¼ inch drill bit
- Meter sticks
- Black permanent markers to mark well number on caps
- Mallets to pound wells into ground and blocks of wood to place on well top when wells are pounded
- All weather copier paper for field data sheets

Groundwater Well Fabrication

- Cut PVC into 70 cm lengths (4 wells per 10 ft of tube), 10 cm will be aboveground, 60cm will be belowground.
- Drill ¼ inch holes in the belowground section of the well (along the 10–60cm length of the well). Drill enough holes to allow water to percolate into the well. The top of the well is the 0-10cm section that has no drill holes; the bottom of the well is the section with the drill holes. To prevent surface water from entering the well the top 0-10cm section of the well is left intact.
- Place a cap on the bottom of each well. Well bottoms should fit snugly, but do not need to be glued.
- Draw a line 10cm down from the top of the well. In the field, this line will serve as a guide for how deep the well should be driven into the peat.

- The remaining caps are for the top of the wells.
- Drill a ¼ inch hole in the center of the remaining top well caps. These caps are used to prevent rainwater from entering the well. A hole is drilled in the center of the top cap for venting.
- Well top caps are installed in the field.

Well Installation (refer to Fig. 4)

- Locate vegetation plot stake.
- Place groundwater well 1m away from the plot stake in the direction of the transect and pound well into the marsh.
- Pound well until only 10cm of well is above ground and all drill holes are below the marsh surface. Use 10cm mark on well as a guide.
- Label top cap (cap with center drill hole) with plot identification number. The well number will be the same as the vegetation plot number.
- Place top cap loosely on well top. Do not jam the cap onto the well top. These caps must be removed to measure the water table level.

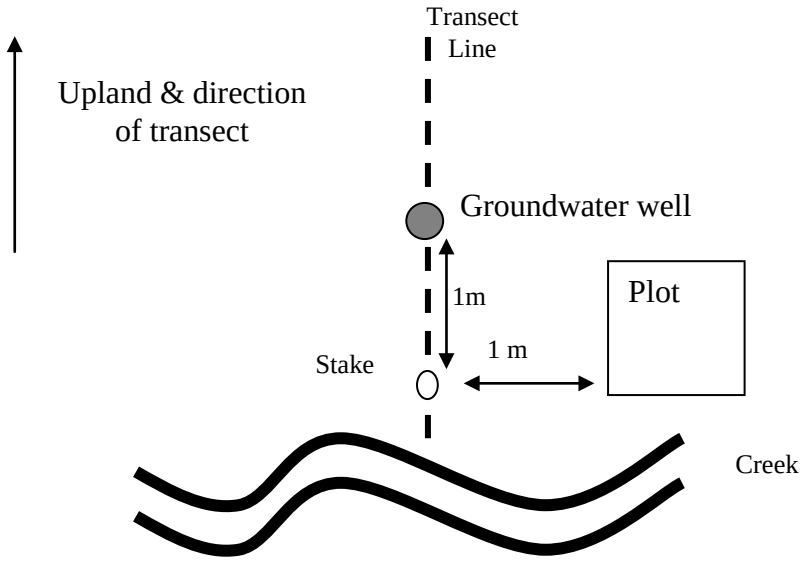


Figure 4. Schematic showing the location of groundwater well relative to stake and vegetation plot.

Materials for Sampling Ground Water Wells

- Meter stick
- Map of ground water well station locations
- Datasheet and pencils (Table 3)

Sampling Procedure (refer to Fig. 5)

- Water table level should be measured within 2 hours of low tide.
- Sampling should occur throughout the growing season, 10-14 intervals.
- Record well number.
- Remove well cap.
- Insert the meter stick into well (0mm end first) until the meter stick barely touches the water surface. By peering into the well as the meter stick is lowered you will be able to see the surface tension of the water break as the meter stick reaches the water surface.
- Record the measurement off the meter stick at the top of the well (Measurement A in Fig. 5 and Table 3).
- Record the height of the well from the marsh surface (Measurement B in Fig. 5 and Table 3). This measurement is important because the well could move from freezing/thawing, trampling, vandalism, *etc*.
- The height of the well from the marsh surface is subtracted from the total distance of the top of the well to the water level. This will give the distance of the water level below the marsh surface. This calculation will be done back in the office and should not be done in the field. The above two numbers are all that is required to be recorded in the field.
- If the well is dry (no water in the well at all), record "dry" on the data sheet
- If the marsh surface is flooded, measure the depth of the water from the marsh surface to the water surface. Be sure to write "surface" on the data sheet next to this measurement.
- Replace the top cap. Be sure not to jam the cap onto the well top.

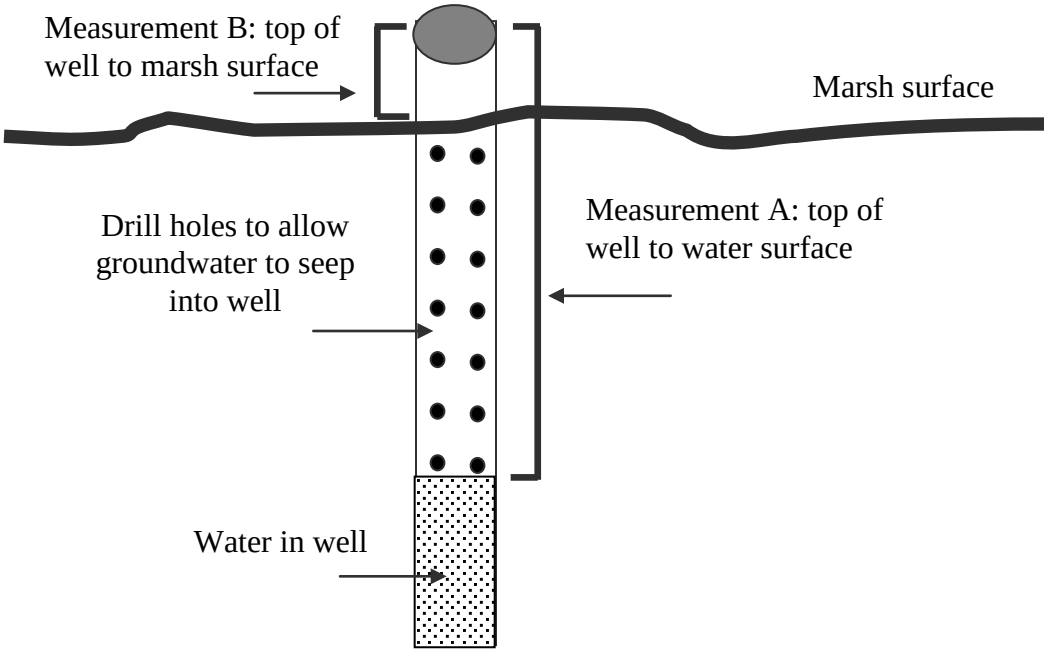


Figure 5. Schematic of groundwater well in place in the marsh

SOIL SALINITY

SUMMARY

In addition to water table level, soil water salinity is an important factor controlling the patterns of salt marsh vegetation. It is not appropriate to sample soil water salinity from water within the groundwater wells for several reasons. First, most useful measurements will be from the portion of sediment that has the most active roots and rhizomes. This is generally from the marsh surface to 10-15cm deep. The groundwater wells are integrating soil water from the surface to a greater depth (60cm). Second, water collected within the groundwater wells tends to stratify over-time, with denser high salinity water near the bottom of the well and fresher water near the surface of the well. The well could be pumped dry before each sampling event, allowed to fill, and then the water in the well sampled for salinity; however, the process of filling could take several hours (although filling is quite rapid for some wells, depending on soil porosity). To avoid these problems with sampling water from the groundwater wells, a soil probe in recommended for collecting soil water.

In conjunction with water table level measurements, sampling of soil water salinity with a soil probe is recommended as follows.

Materials for Soil Salinity

- Soil probe, constructed of stainless steel tubing, 1/16 inch (1.6mm) inner diameter, 1/8 inch (3.2mm) outer diameter, wall thickness 1/32 inch (0.8mm) (e.g., gas chromatography tubing), cut to about 70 cm length, with one end crimped and slotted to allow entry of soil water. Any rigid tubing that approximates these dimensions is satisfactory to use (Fig. 6).
- 10-15cc plastic syringe, or larger volume syringe up to 50cc
- 5cm length of plastic tubing to attach the soil probe to the syringe
- Salinity hand-held refractometer
- Filter paper (cut-up coffee filters are fine)
- Plastic squeeze bottle with freshwater to rinse and calibrate refractometer
- Data sheets and pencils (Table 3)

Soil Probe Fabrication

- Make 3 – 4 slits approximately 5mm apart and 2.5cm from one end of the metal tubing. The slits can be made with a roto-tool or a fine blade hacksaw. The slits are to allow water to be drawn up into the tube (Fig 6).
- Close the end of the metal tube (nearest to the slits) by crimping with pliers.
- Attach a short length of plastic tubing to the uncrimped end of the metal tubing.
- Attach the syringe to the other end of the plastic tubing.

- Make sure that water can be drawn up into the tubing by pulling the plunger on the syringe.
- Mark increments of 15cm, 30cm, and 45cm on the metal tube with tape so that depth of the soil salinity sample can easily be determined.

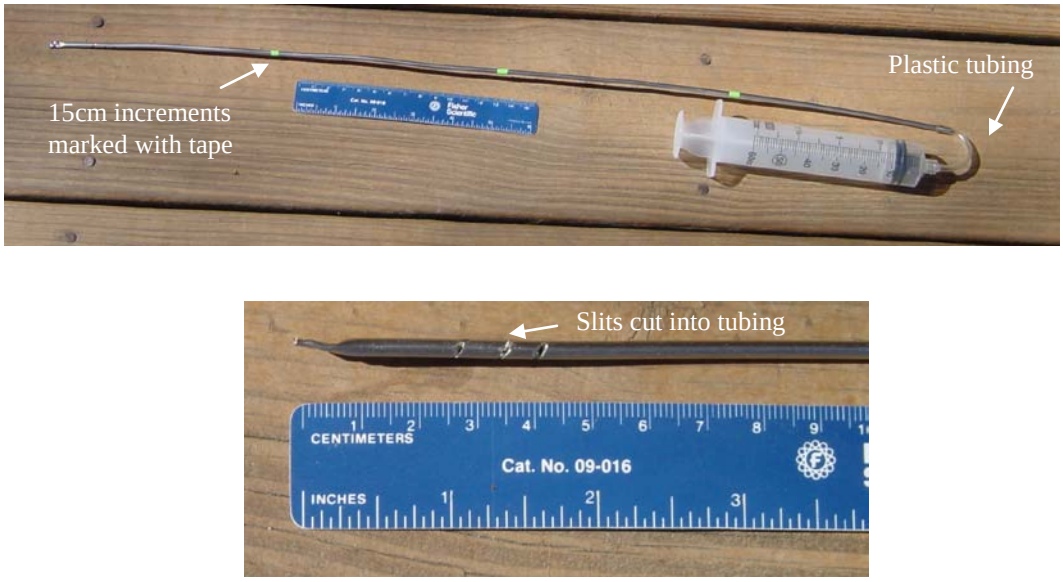


Figure 6. Photograph of a soil probe used to sample soil water salinity.

Sampling Procedure

- Sampling should coincide with groundwater well sampling. Always measured within 2hrs of low tide.
- Calibrate (zero) hand-held salinity refractometer with fresh water (tap is okay) before EACH field day.
- At a location near the groundwater well, insert the soil salinity probe (crimped end downward) 15cm into the sediment (Tape can be used to mark 15cm). The plastic syringe is attached to the top of the probe. Carefully withdraw the plunger to collect soil water.
- Once several milliliters (just a few drops) of water have been withdrawn into the syringe, detach it from the probe. If the marsh is dry at 15cm, then insert the probe deeper (30cm, then 45cm) until soil water is collected. Record the depth that soil water was collected. Record dry if no soil water was collected at 45cm.
- Place a piece of filter paper over the nozzle of the syringe. Depress the syringe plunger and let the water pass through the filter paper and onto the glass plate of the refractometer.
- Read and record the soil water salinity (Table 3). The station location for the soil salinity is the same as the water table level station and vegetation plot.

- Clean-up. Discard (never re-use) the filter paper. Using water from the groundwater well or a nearby creek, rinse silt and sediment from the probe by drawing up water into the syringe. Discard all the water in the syringe and probe before sampling the next station. Rinse refractometer with freshwater; dry refractometer.
- SAMPLING FREQUENCY: Soil salinity should be sampled in conjunction with groundwater sampling (10-14 day intervals during the growing season).

Table 3. Example of a field data sheet for water table level and soil salinity monitoring.

Water Table Level & Soil Salinity Monitoring

SITE _____ DATE _____

Data Collector(s) _____

Note: *If water is below marsh surface indicate water table level with a negative sign in "Depth Column". If water is on the marsh surface, write "surface" in Column A, measure water depth and record depth with a positive sign in "Depth" column. If water is below marsh surface than water table depth will be negative. If water is on marsh surface than depth will be positive.*

Plot No.	Time	A. Top of Well to Water (cm)	B. Top of Well to Marsh (cm)	Depth to Water Table (B-A)[1]	Salinity (ppt)	Depth if other than 15 cm

[1] calculate in lab.

SOIL SULFIDES (An Optional Monitoring Variable)

SUMMARY

Under waterlogged and anaerobic soil conditions sulfide concentrations can rise to levels that are toxic to root metabolism, often inhibiting nitrogen uptake and plant growth (Howes et al 1986, Koch *et al.* 1990). When restoring salt marshes by re-introducing tidal flow it is suggested that soil porewater sulfide be monitored as a measure to help understand why vegetation patterns are changing. For example, if tidal flow is re-introduced to a wetland site and the soils become waterlogged then high sulfide levels could result. Sulfide toxicity from waterlogged soils can stress *Phragmites* growth, while *Spartina alterniflora* is more tolerant of high sulfide (Chambers *et al.* 1998).

Field and laboratory methods for total sulfides in salt marsh porewaters are described in detail by Portnoy and Giblin (1997) and will only be summarized here.

Materials for Sampling Soil Sulfides

- Soil probe (same probe as used for salinity as described above), constructed of stainless steel tubing,1/16 inch (1.6mm) inner diameter, 1/8 inch (3.2mm) outer diameter, wall thickness 1/32 inch (0.8mm) (e.g., gas chromatography tubing), cut to about 70 cm length, with one end crimped and slotted to allow entry of soil water. Any rigid tubing that approximates these dimensions is satisfactory to use. See Soil Salinity section for fabrication instructions.
- 10-15cc plastic syringe, or larger volume syringe up to 50cc
- Small volume pipette

Sampling Procedures for Soil Sulfides (Field and Lab)

- Sampling should coincide with groundwater well and soil salinity sampling. Always measured within 2hrs of low tide.
- At a location near the groundwater well, insert the soil salinity probe (crimped end downward) 15cm into the sediment (tape can be used to mark 15cm). The plastic syringe is attached to the top of the probe. Carefully withdraw the plunger to collect soil water. Be certain that air is purged from the probe and syringe prior to sampling using a three-way valve.
- Once several milliliters of water have been withdrawn into the syringe, detach it from the probe. If the marsh is dry at 15cm, insert the probe deeper until soil water is collected. Record the depth that soil water was collected.
- In the field, the porewater sample is collected from the syringe with a pipette and discharged into 2% zinc acetate and stored on ice. Volume of the pipette depends on expected sulfide concentration, but 0.1 ml is often appropriate.
- Sulfide is determined colorimetrically after Cline (1969).
- SAMPLING FREQUENCY: Soil sulfide should be sampled at least monthly during the growing season in conjunction with groundwater and soil salinity sampling.

MOSQUITO LARVAL SAMPLING

SUMMARY

Mosquito larvae will be sampled using the standard "dip count" method along the established vegetation transects. Mosquito larvae should be sampled 4 to 5 days after a tide that has flooded the surface of the marsh. This is usually a full or new moon high tide, but can also be associated with rainfall events and storms. This time frame corresponds to the period when mosquito larvae are present on the marsh. Adult mosquitoes deposit their eggs on moist soil or stems of grasses where the eggs must dry for at least 24 hours. The eggs hatch when flooded by the monthly high tides or a rainfall event. After hatching, the larvae pass through five developmental stages, or instars, that reside in small, stagnant pools before emerging in one to two weeks as adult mosquitoes. Larval sampling is conducted during this development period. Larvae will be sampled at approximately every 15-20 m along the transect, this corresponds to samples in the vicinity of every vegetation plot and in between each vegetation plot.

Materials for Sampling Mosquito Larvae

- Mosquito dipper
- Sample vials for mosquito larvae
- Labels for sample vials
- Map of vegetation transect locations
- Datasheet and pencils (Table 4)

Sampling Procedure for Mosquito Larvae

- Sample for mosquito larvae 4 to 5 days after a tide has flooded the marsh surface. This could be after new or full moon high tide, storm surge, or a rainfall event that floods the marsh. Four to five days after flooding, larvae should be in the 3rd or 4th instar and are therefore easier to count and identify.
- Optimal sampling time will not always be as predictable as the full and new moon tides. Investigators should be ready to sample 4 to 5 days after major rainfall events as well.
- Sample larvae at least once per month from May to October (twice per month is better).
- Mosquito larvae sampling stations are located at approximately 15m to 20m intervals along each vegetation transect. This should correspond to sampling in the vicinity of each vegetation plot as well as in between each plot. A meter tape should be used to measure the distance between vegetation plots to locate stations between each vegetation plot. To ensure that the same approximate location is sampled each time, sample stations could be marked with flags or another identifying marker.
- Mosquito sampling stations located at vegetation plots should be identified as the vegetation plot ID. Those stations in between plots should be identified as the meter distance along the vegetation transect. For example, a mosquito sampling

station located at vegetation plot 1-120 (transect 1, distance 120m along the transect) would be identified as 1-120, a mosquito sampling station located between vegetation plots 1-120 and 1-140 would be identified as 1-130.

- When walking the vegetation transect for mosquito sampling be sure not to walk in the permanent vegetation plots. Offset the mosquito larvae sampling transect several meters to the left or right of the plot stake.
- Transects should be walked facing the sun, so the sampler's shadow is not cast onto the standing water that is sampled. Mosquito larvae will quickly disperse if a shadow is cast over them.
- Once at a sampling location (a permanent vegetation plot or in between 2 adjacent plots) locate the nearest standing water within a 3m radius. If no standing water is present record "dry" on the data sheet. Record the type of water that was sampled on the data sheet using the following categories:
    - creek
    - pool
    - panne
    - SW: standing water in vegetation
    - Other: be sure to define what "Other" represents
- If standing water is present, sample the edges of the standing water with a standard mosquito dipper. Attempt to get a full dipper of water. If this is not possible, estimate the amount of water using the following coded categories and record the amount on the data sheet:
    - 5: full dipper
    - 4: ¾ full
    - 3: ½ full
    - 2: ¼ full
    - 1:less than ¼ full
    - 0: no water in dipper

  Some mosquito dippers have volume increments on the inside of the dipper than can be used to estimate the water volume.
- Count all larvae up to 100 in the dipper. Larvae are be counted by letting the water in the dipper slowly flow out of the dipper lip and back on to the marsh surface. If there are more than 100 larvae estimate the number using the following categories:
    - 100 to 200 larvae
    - 200 to 300 larvae
    - 300 to 500 larvae
    - > 500 larvae
- In the field, save a sub-sample of the larvae (approximately 5 individuals from each sample station) and place them in a vial with water from the dipper. Record the sample location on the vial. These larvae will be brought back alive to the lab for species identification.

Method to Preserve Mosquito Larvae for Identification

- If the mosquito larvae are not identified immediately they must be preserved for later identification upon return to the laboratory.
- First the mosquito larvae should be killed in hot water. Boil a small amount of water and let it cool until it stops bubbling. Then put the mosquito larvae in the water and remove them after about 50 seconds. This heating is necessary to maintain taxonomic characteristics for identification.
- Remove larvae from water either by straining or individually with forceps.
- After heating the larvae should be preserved in either a Dietrich's solution or an AGA solution, both are described below.
- Dietrich's solution: The formula for Dietrich's solution is:
    - 30 parts distilled water (=58%)
    - 15 parts 95% ethanol (=29%)
    - 6 parts formaldehyde (=11%)
    - 1 part glacial acetic acid (=2%)

  Specimens should be transferred from Dietrich's solution to a long-term preservative (*e.g.*, 75% ethanol) after 24 hours. However, they can be left in Dietrich's solution for months before transferring them, if necessary.
- AGA solution: AGA solution is a 1:1:8 solution (by volume) of glacial acetic acid:glycerin:75%ethanol. The formula for AGA solution is:
    - 1 part glacial acetic acid (=10%)
    - 1 part glycerin (=10%)
    - 8 parts 75%ethanol (=80%)
- Record sample date, site, and station location on vial.

Table 4. Example of a field data sheet for mosquito larvae monitoring.

Mosquito Larvae Sampling Data Sheet

Site ________________________ Date ___________ Sampler __________

Dipper volume codes: 0=no water, 1= < ¼ full, 2=¼ full, 3= ½, 4=¾, 5=full

Station #	# Mosquito Larvae	Water volume in Dipper	Area dipped (dry, pool, panne, standing water in vegetation)	Larvae saved for ID? (yes or no)

NEKTON SAMPLING IN PONDS

SUMMARY

This estuarine nekton protocol recommends sampling exclusively with $1m^2$ throw traps in shallow salt marsh ponds. The species composition and abundance (density) of nekton (fish and decapod crustaceans) is measured with a $1m^2$ enclosure trap in shallow water (< 1m) habitats such as marsh ponds. Enclosure traps provide a repeatable, quantitative estimate of nekton utilization of specific habitats. Beach seines are useful for determining species composition and relative abundance, but provide a less repeatable method. Raposa and Roman (2000 and 2001) provide details on the method, sample size requirements, and information on data analyses. There should be two daytime sampling efforts per year; one in early summer (June-July) and another in late summer-early fall (August-October), unless there are species or processes unique to other seasons that are of interest. The number of throw trap samples required depends on the habitat under examination, but generally between 25 and 50 samples should be collected from each sampled habitat during each sample period. This protocol recommends sampling twice in summer and is presented as a minimum for nekton monitoring.

Materials for Identifying Station Locations

- Oak stakes to mark station locations
- Mallet to pound stakes into ground
- Black permanent markers to mark transect and plot number on stakes
- Colored flagging (optional) to tie to oak stakes
- Compass
- Random number table (to determine specific station location at each pond)
- Aerial photos of study sites
- Draft map of study site showing boundaries of study areas and approximate location of ponds

Nekton Pond Station Location Procedure

All pond sampling stations should be randomly selected prior to monitoring. The best method is to obtain either an aerial photo or GIS map of the study area and randomly selecting pond sampling stations using the procedure outlined below.

- Obtain an aerial map of the study area.
- Identify all ponds within the study area.
- If there are more than 25 ponds, sequentially number each pond within the study area. Random numbers between one and the total of number of ponds are then generated using a random number generator found in several spreadsheet

programs. The first 25 random numbers from the generated list are chosen, each of these random numbers correspond to a pond that will be sampled.
- If there are 25 or fewer ponds, all ponds will be sampled.
- Locate the pond in the field.
- Pace the perimeter of the pond to estimate the length of the perimeter.
- Select a random number between one and length of the perimeter.
- Locate the station (point where the throw trap will be released from) by pacing the distance of the random number. For example, if the pond perimeter is 25 meters, and the random number is 17, the station location will be 17m around the perimeter.
- Alternatively, a random compass bearing (between 0 and 360) can be chosen to location the station location on the pond.
- Locate the station in the field and mark with a 1m oak stake.
- Label the oak stake with the station number. The naming convention should be "P" for pond and the station number, one through the total number of stations sampled in that area (*e.g.* P1, P2, *etc*.).
- If a pond is large, more than one station may be located at the pond if additional stations are needed. However, stations located on the same pond should not be sampled at the same time and sampling time should be spaced apart by at least 30min. If two stations are located on the same pond, the naming convention is the same as explained above. For example, P1 and P2 can be on the same pond.

Materials for $1m^2$ Throw Trap Construction

- The throw trap measures 1m wide x 0.5m high. The bottom and top of the trap are open
- Eight, 1m long by 2.5cm aluminum bars
- Four, 0.5 m long by 2.5cm angle aluminum bars
- Nuts, bolts and lock washers to attach aluminum bars to angle bars
- 3mm hardware cloth (when reporting results from this method, investigators should cite a 3-mm mesh size, the mesh size of the throw trap)
- thin gauge wire or cable ties to attach hardware cloth to aluminum frame
- Nylon netting, 3mm mesh, 4m long by 0.5m width (for skirt)
- 4m of float cord (for skirt)

Materials for Dip Net Construction

- The dip net measures 1m long x 0.5m wide, and fits snugly within the throw trap (Fig 7).
- Approximately 4m of 1.25cm diameter aluminum rod that can be bent into the shape of the dip net with a 0.5m handle.
- 0.5m length of 2.5-5.0cm diameter steel pipe (to fits over the aluminum rod to strengthen the handle).
- Cable ties or sewing thread.

Throw Trap and Dip Net Fabrication

- The throw trap measures $1m^2$ x 0.5m high (Fig. 7).
- Construct the frame of the throw trap by attaching the 0.5m long 2.5cm angle aluminum angle bars (forms the corners of the trap) to the 1m long 2.5cm straight aluminum bars (forms the sides of the trap) with nuts, bolts, and lock-washers.
- Once the frame is built, the four sides of the trap are surrounded by 3mm mesh hardware cloth that is attached to the horizontal frame bars with thin gauge wire. Attach hardware cloth (with thin gauge wire or cable ties) to the 4 sides of the trap, leaving the top and bottom of the trap open.
- If water depths are expected to exceed 0.5m, the height of the trap can be extended to 1 m by attaching a skirt (3mm mesh nylon netting) to the top of the trap. The skirt is equipped with float-cord along the top edge to ensure that the top of the skirt floats at the waters surface.
- Bend the aluminum rod into the shape of the dip net (1m long by 0.5m wide) with a 0.5m handle.
- 0.5m length of 2.5-5.0cm diameter steel pipe can be fit over the aluminum rod handle of the dip net to strengthen the handle.
- Attach the 1mm mesh nylon netting (1.25m by 0.75m), to the dip net frame either with numerous small cable ties, or by sewing with twine or wire cable ties. Use of a 1mm mesh dip net facilitates collection of all nekton within the $1m^2$ frame.
- When reporting results from this method, investigators should cite a 3-mm mesh size, the mesh size of the hardware cloth.

Figure 7. $1m^2$ throw trap. The investigator is sweeping the trap with the 1m x 0.5m dip net. Note the skirt of 3mm nylon mesh net attached to the top of the trap for sampling in deeper water.

Materials for Sampling Nekton in Ponds

- $1m^2$ throw trap
- small ruler (to measure nekton)
- Map of pond station locations
- Data sheet and pencils (Table 5)
- Identification keys
- Any other equipment necessary for taking environmental variables (*e.g.* refractometer, oxygen probe, thermometer)

Sampling Procedure for Nekton Using $1m^2$ Throw Trap

- Nekton sampling should occur at the same relative tide stage.
- Sampling salt marsh ponds should occur only after the marsh surface is drained of tidal water. We generally begin sampling in seaward habitats where the marsh surface drains first, and then proceed to landward areas following the tidal prism. This method ensures that samples are collected at similar water depths throughout the marsh, and is thus one way to control for the effects of tide stage.
- Samples are collected by approaching to within 4 to 5m of a marked station with the throw trap. Approach the station quietly so as not to not disturb or startle the nekton. Only the person throwing the throw trap should approach the station, all others should remain at a distance (>10m) from the station to avoid startling the nekton. Pond stations are approached by crouching low and walking over the marsh surface, then waiting about 2 minutes before throwing the trap.
- The trap is thrown into the water by tossing it from the hip like a giant Frisbee (Fig. 8). The trap is then quickly pushed into the sediment to prevent escape of nekton from under the trap. In order to minimize disturbance, replicates are never taken from the same station in a single sampling period.
- Once the sample is secured, nekton is removed by the large dip net.
- The net is slid downward into the trap, flush against the side of the trap nearest the researcher. The net is then moved across the trap with the forward edge of the net always remaining flush against the sediment until the opposite side of the trap is reached. In muddy sediments the dip net often goes through a thin layer of surface sediment, capturing buried nekton. The net is then moved upward out of the trap, again keeping the leading edge flush against the far wall of the trap.
- The dip net should be used from at least three sides of the trap because nekton may be hiding in the trap corners.
- The dip-netting procedure is repeated until three consecutive dips do not capture any animals or if the first four dips come up empty. At this point the trap is considered empty.
- Animals are processed as they are captured. All animals are identified to species in the field and immediately released at the same station.
- In each sample, up to fifteen individuals of every species are measured to the nearest mm for total length (from the tip of the snout to the tip of the caudal fin for fishes; from the tip of the rostrum to the tip of the telson for shrimp) or carapace width for crabs (the distance between the two furthest points across the

carapace). Generally, dominant fish species (*i.e.* the mummichog, *Fundulus heteroclitus*) is counted and measured as two categories, juveniles (<45mm) and adults (>45mm). Juveniles and adults can be entered on the data sheets under separate species (Table 5).

- Nekton may be identified using any number of guides that are specific to the Atlantic coast and New England regions, including Bigelow and Schroeder (1953), Gosner (1978), and Robins *et al.* (1986), Eddy and Underhill (1978).
- Individuals that are difficult to identify may be humanely sacrificed by a strong blow to the head, preserved in 70% ethanol, and returned to the laboratory for identification.
- Any associated environmental variables should be measured at this time (see section entitled "Other Environmental Variables Monitored During Nekton Sampling").

Figure 8. Sampling technique for 1m^2 throw trap. The trap is tossed like a frisbee into the pond that is being sampled.

Table 5. Sample nekton throw trap field data sheet.

THROW TRAP DATA SHEET

SITE:_____ DATE: _____ TIME:_____

STATION #:_____ SAMPLING CREW: _____

GPS Coordinates: N_____ E_____

Water temp: _____ Salinity: _____ DO: _____

Water Depth: _____ Tide (circle one): Flood Ebb Vegetation (circle one): Yes No

Vegetation Species #2 _____ Veg. % Cover: <1% 1-5% 5-25% 50-75% >75%

NEKTON SPECIES & MEASUREMENTS

SPECIES #1 _____ Total # of individuals: _____

Talley (include measured fish): _____

LENGTHS: _____
(15)

SPECIES #2 _____ Total # of individuals: _____

Talley (include measured fish): _____

LENGTHS: _____
(15)

SPECIES #3 _____ Total # of individuals: _____

Talley (include measured fish): _____

LENGTHS: _____
(15)

SPECIES #4 _____ Total # of individuals: _____

Talley (include measured fish): _____

LENGTHS: _____
(15)

SPECIES #5 _____ Total # of individuals: _____

Talley (include measured fish): _____

LENGTHS: _____
(15)

NEKTON SAMPLING IN DITCHES and SMALLER TIDAL CREEKS

SUMMARY

Common features on salt marshes are grid ditches that were created for mosquito control purposes in the 1940's. These ditches vary in width, (usually 45cm and 100cm in wide) and on some marshes, especially those in the southern New England, are the only water habitat within the marsh. The throw trap is not a good sampling gear for the grid ditch habitat, as the trap is too large. Even smaller versions of a throw trap would not sample these areas as the trap would have to land precisely in the ditch in order to enclose the nekton. This protocol outlines an enclosure sampling gear, the ditch net, that we have designed to sample these narrow tidal channels. The center body of the net lines the sides and bottom of 1 linear meter (approximately) of ditch. There are two doors on the open ends of the net, which when pulled, rise up to close off the ends of the net, enclosing an area of water that is 1m long and as wide as the ditch. This sampling gear is designed to sample mosquito ditches and smaller tidal creeks up to 1m wide and 1m deep. There should be two daytime sampling efforts per year; one in early summer (June-July) and another in late summer-early fall (August-October), unless there are species or processes unique to other seasons that are of interest. The number of ditch net samples required depends on the habitat under examination, but initially we recommend at least 10 samples should be collected from each sampled habitat during each sample period.

Materials for Identifying Ditch Station Locations

- Oak stakes to mark station locations
- Mallet to pound stakes into ground
- Black permanent markers to mark transect and plot number on stakes
- Colored flagging (optional) to tie to oak stakes
- Random number table (to determine specific station location along ditches)
- Aerial photos of study sites
- Map of study site showing boundaries of study areas and approximate location of ditches

Nekton Ditch Station Location Procedure

All sampling stations should be randomly selected prior to monitoring. This is best accomplished by obtaining an aerial photograph of the study location to identify the ditches and smaller tidal creeks (<1m wide).

- Estimate length of ditch.
- Choose a random number (from a random number table) between zero and the total ditch length.

- Locate station location at the random number and mark with a 1m oak stake and colored flagging. For example, if the ditch is 120m long and the random number is 33, the station would be located at 33m along the ditch.
- Label stake with ditch net station ID. The naming convention for ditch stations is "D" for ditch and the station number, one through the total number of stations sampled in that area (*e.g.* D1, D2, *etc*.).
- Establish at least 10 ditch net stations per study area.
- Generally, only one station is located per ditch.
- If there are few ditches, more than one station can be located on the same ditch, but the station should be at least 20m apart.
- Record geographic coordinates (UTM, latitude/longitude) of all station locations using a sub-meter accurate GPS unit.

Materials for Ditch Net Construction (for 1 net)

- NYLON netting (24lb test), 1/8in mesh, at least 1m deep. Each net takes 5 meters of netting – a 1m X 3m section for the center of the net (sides & bottom) and two 1m X 1m sections the doors
- 20m of nylon rope, 3/16in diameter. Each net takes 20m of line – four 4m lengths for rip cords and four 1m lengths for runner lines of the doors
- 5m of leadcore line; m for the top of each door (total 2m) and 3m for the floor of the net
- 4 eye-hooks with 1in eyes
- 4 oak stakes – 1.5 to 2m long, 2.5cm square
- Staple gun and 3/8 in stainless steel staples
- D-ring hand pliers and 9/16 in C-ring fasteners
- 25 to 30 plastic rings or rubber O rings, approximately 2.5cm diameter

Ditch Net Fabrication

- Cut a 1m by 3m section of the nylon netting for the center of the net.
- Cut two 1m by 1m sections of nylon netting for the doors of the net.
- Attach the doors of the net to the center section. The doors should be centered on the main body of the net along the 3m length (Fig 9a). To attach the doors take a 1m length of leadcore line and wrap the nylon netting from the leading edge of the door and the center 1m middle section of the net body around the leadcore and fasten the two pieces of nylon netting to the leadcore line with the D-ring pliers and 9/16 in C-ring fasteners.
- Attach 5 to 7 nylon rings or rubber O-rings to sides of the doors (side A in Fig 9a). Use the D-ring pliers to attach the rings to the nylon netting. The rings should be attached to the edge of the netting so the center of the ring is clear of the netting. The draw cord that pulls the doors up passes through these rings.

- Attach 3 to 5 plastic rings to the top of the door (side B in Fig. 9a). Use the D-ring pliers to attach the rings to the nylon netting. The rings should be attached to the edge of the netting so the center of the ring is clear of the netting.
- Attach a short length of lead core line to the top of each door (Fig. 9a, side B) using either cable ties or the D-ring pliers and C-ring fasteners. This is to weigh down the top of the net so it does not float up, and impede the passage of fish through the net.
- Attach a length (approximately 1m) of leadcore line to the bottom center of the net (Fig. 9b) on the outside of the net using either cable ties or the D-ring pliers and C-ring fasteners. This is to weigh down the center of the net so it does not float up when placed in the ditch.
- Attach the net to the four oak stakes using a staple gun and stainless steel staples. The free edges of the net (Fig 9a, side C, and Fig. 9b between points 1 and 2) are stapled to the oak stakes. The portion of the net closest to the doors should be stapled starting at approximately 30cm (1 ft) from the bottom of the oak stake, and continue up towards the top of the stake. The bottom 1ft of the stake should be free of the net so that the stake can be pushed into the ground to hold the net in place while it is deployed.
- The runner lines are attached next. The runner lines hold the plastic rings close to the stake, so when the door is pulled up the net remains close to the stake.
- Attach the bottom of runner lines to the interior of the stakes (on top of the stapled netting). The runner lines are approximately 1m in length. The bottom of the runner line should be attached at the intersection of the doors and main body of the net. Tie a few knots in the end of the line and staple the line to the stakes using several staples close together on each side of the knot so the line will not pull loose.
- Pass the free end of the runner line through the 5 plastic rings that are attached to side A (Fig. 9a) of the door closest to the stake (Fig. 9b, runner line (3) and plastic rings (4)). The bottom most ring is added first, then the next ring, until all rings for that door side are on the runner line. The runner line is then pulled taut against the stake and the free end is stapled approximately 5 to 8cm above the end of the net. After stapling a knot should be tied in the free end of the line and stapled again on either side of the knot to ensure the runner line does not come loose.
- Attach the rip cord to the center ring on the top of the door, and pass the rip cord through one of the rings on the corner of the door. Then pass the rip cord through the top ring of the door that is attached to the runner line. Attach another rip cord to the same center ring, and pass it through the other corner ring, and the top ring on the other side of the door. When these lines are pulled, they will pull on the top rings attached to the doors, which in turn will pull the sides of the doors up the stakes to enclose the sides of the net.
- Attach the rip cords to the other side of the net as described above.
- Attach the eye-hook to the oak stake. When the net is held upright, with the 4 stakes sticking into the ground, the eye-hook should be placed on the outside of the stake. The free end of rip cord is passed through the eye-hook. When the rip

Fig. 9a

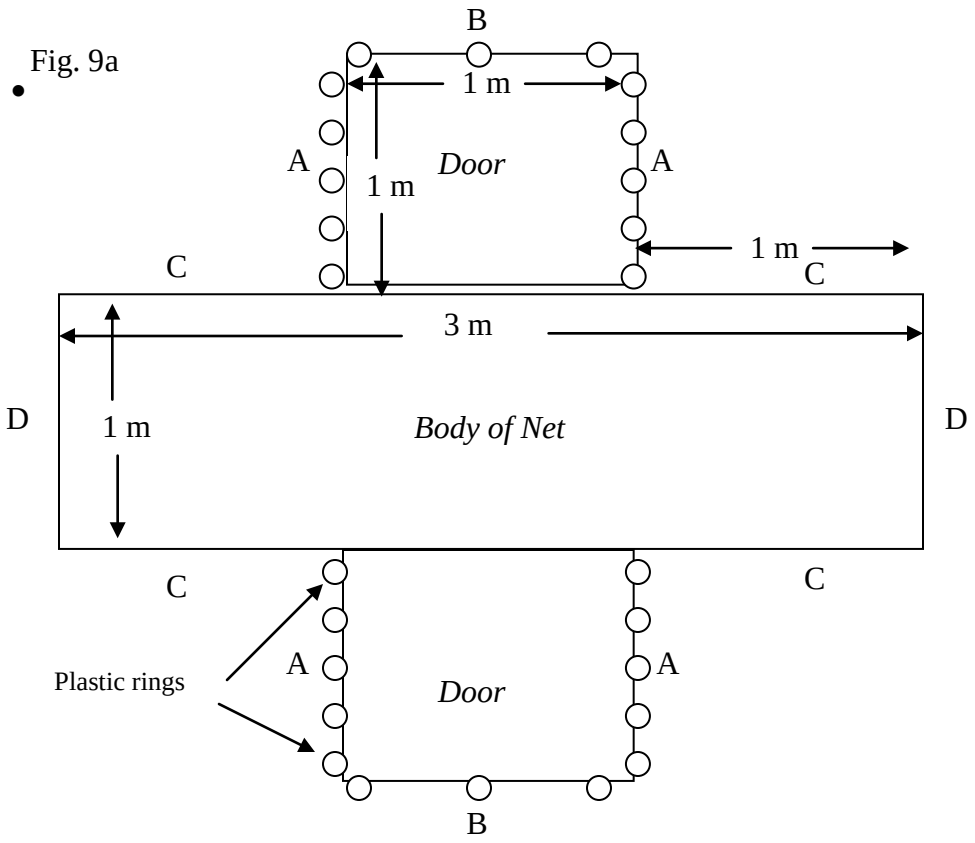


Fig. 9b.

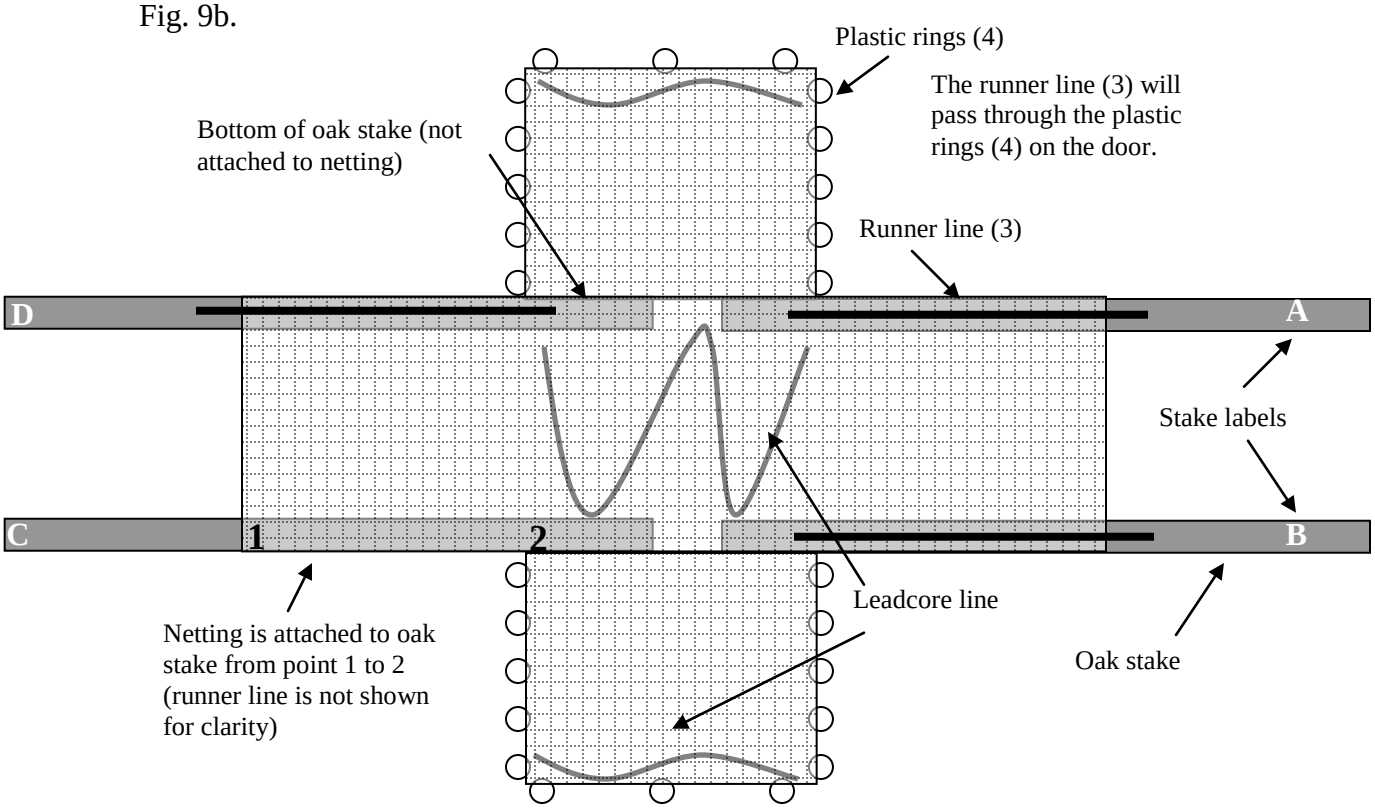


Figure 9a and b. Schematic of ditch net showing dimensions of nylon netting and attachment points for leadcore line, plastic rings, runner lines, and oak stakes. Fig. 9b is looking down into the interior of the net.

is pulled the line should pass easily through the eye-hook, so the doors are pulled up smoothly.

- Label the stakes. We usually label the stakes A, B, C, and D. Be sure to label each net exactly the same. The labels are used to set the net correctly in the ditch and to measure the distance between the stakes in order to determine the area of the water that the net was fishing (refer to data sheet). For example, stakes A and B are place on one side of the ditch and stakes C and D are place on the opposite side of the ditch (Fig. 9b).
- Test each net to be sure that the rip cords pull up the doors smoothly and quickly.

Materials for Sampling Nekton in Ditches

- Ditch nets
- small ruler (to measure nekton)
- meter stick (to measure net, creek and water depths)
- Map of ditch station locations
- Data sheet and pencils (Table 6)
- Identification keys
- Any other equipment necessary for taking environmental variables (*e.g.* refractometer, oxygen probe, thermometer)

Sampling Procedure for Nekton Ditch Net

- Nekton sampling in ditches should occur at the same relative tide stage. Sampling salt marsh ditches should occur only after the marsh surface is drained of tidal water. Sampling should occur on a high slack or ebb tide, when the marsh surface has drained.
- Nets are placed at the station locations in the ditches at least 30min before sampling. This usually means that the nets are placed at flood or slack tide.
- Set up a ditch net usually requires 2 people, each standing on opposite sides of the ditch. One person will take stakes labeled “A” and “B” and place the stakes into the bottom of the ditch close to the side of the ditch. The other person will take stakes labeled “C” and “D” and place them on the opposite side of the ditch. The net should be stretched tight between stakes “A” and “B” and stakes “C” and “D” so that approximately a 1m section of ditch is sampled. (Fig 10a).
- The rip cords should be pulled to make sure that the lines are not fouled and that the doors will pull up smoothly and quickly.
- Push the doors and the center of the net down into the bottom of the ditch with the meter stick. Make sure that the net lays down on the bottom of the ditch, so that fish passage through the net is not impeded.
- Measure the distance between all the stakes (*e.g.* “A” to “B”, “B” to “C”, “C” to “D”, and “D” to “A”) and the diagonal distance between stakes “A” and “C” and record these on the datasheet (Table 6). These distances are measured when the

net is placed in the ditch and are necessary to calculate the area of water that is sampled.

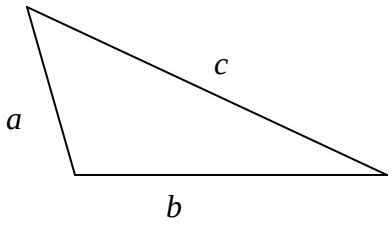
- Lay the rip cords out on the marsh surface as far from the net as possible without pulling on the doors.
- Note the time that the net is deployed on the data sheet.
- Ditch nets should not be sampled until they have been deployed for at least 30min. This time period is necessary to minimize any disturbance to nekton caused by placing the net in the ditch.
- At high slack or ebb tide, the ditch nets are pulled. Two people are required to pull the ditch nets. The nets are quietly approached from opposite sides of the ditch, one person on each side. Upon reaching the rip cords, each person kneels and waits quietly for approximately 2min. The rip cords should not be handled during this time, as the vibrations on the cords can be transmitted to the stakes and possibly disturb nekton that are in the net. At a pre-determined signal, both people quickly pull on the rip cords and run towards the net. The doors of the net will pull up, enclosing nekton within the net (Fig. 10b and 10c).
- The net is then quickly lifted out of the ditch and onto the marsh surface. The best way to do this is to have both people pull the stakes out simultaneously (while still maintaining pressure on the rip cords). All four stakes are then handed to one person who will lift the net out of the ditch and onto the marsh surface. It is important to quickly pull the stakes and net out of the ditch, since this creates a bag of netting in the center of the net where the fish are trapped.
- The net is then laid out on the marsh surface and the nekton are identified, counted, and measured.
- The collection time should be recorded.
- In each sample, up to fifteen individuals of every species are measured to the nearest mm for total length (from the tip of the snout to the tip of the caudal fin for fishes; from the tip of the rostrum to the tip of the telson for shrimp) or carapace width for crabs (the distance between the two furthest points across the carapace). Generally, dominant fish species (*i.e.* mummichog, *Fundulus heteroclitus*) is counted and measured as two categories, juveniles (<45mm) and adults (>45mm). Juveniles and adults can be entered on the data sheets under separate species.
- Nekton may be identified using any number of guides that are specific to the Atlantic coast and New England regions, including Bigelow and Schroeder (1953), Gosner (1978), and Robins *et al.* (1986), Eddy and Underhill (1978).
- Any associated environmental variables should be measured at this time (see section entitled “Other Environmental Variables Monitored During Nekton Sampling”).

Calculating the Area of a Ditch Net

- The area of a ditch net is calculated as the sum of two irregular triangles.
- The areas of the 2 irregular triangles are calculated from the 5 distances measured in the field.
- The formula for an irregular triangle is:

$$\sqrt{[s*(s-a)(s-b)(s-c)]}$$

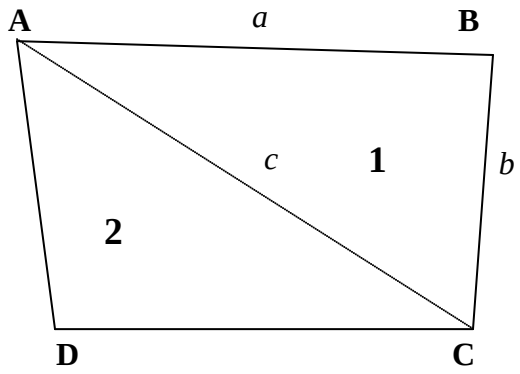
Where: $s = \frac{(a+b+c)}{2}$



- For example, a net with the following dimensions:

Where:

A to B = 81cm
B to C = 73cm
C to D = 71cm
D to A = 76cm
A to C (diagonal) = 109cm



s for Triangle 1: $s = \frac{(81+73+109)}{2} = 131.5$

The area of Triangle 1:

$\sqrt{[131.5*(131.5-81)(131.5-73)(131.5-109)]} = 2684.9\text{cm}^2$

s for Triangle 2: $s = \frac{(71+76+109)}{2} = 128$

The area of Triangle 2:

$\sqrt{[128*(128-71)(128-76)(128-109)]} = 2956.5\text{cm}^2$

The total area of the net would be: $2956.5\text{cm}^2 + 2684.9\text{cm}^2 = 5641.4\text{cm}^2$ or 0.56m^2

Figure 10a-c. Photos of ditch net in the field showing correct deployment (Fig 10a), doors being pulled up (Fig. 10b), and the net once the doors have been pulled (Fig 10c).

Table 6. Sample nekton ditch net field data sheet.

Nekton Ditch Sampler Data Sheet

SITE:_____ DATE: _____

STATION #:_____ SAMPLING CREW: _____

GPS Coordinates: N_____ E_____

Habitat Type: Tidal Creek (TC) Open Ditch (OD) Plugged Ditch (PD)

Deployment Time: _____ Collection Time: _____

Net Distance Measurements:

A to B:_____ B to C:_____ C to D: _____ D to A: _____ A to C (diagonal): _____

Water temp: _____ Salinity: _____ DO: _____

Water Depth: _____ Creek/Ditch Depth: _____ Tide: Flood or Ebb

NEKTON SPECIES & MEASUREMENTS

SPECIES #1 _____ Total # of individuals: _____

Talley (include measured fish): _____

LENGTHS: _____

SPECIES #2 _____ Total # of individuals: _____

Talley (include measured fish): _____

LENGTHS: _____

SPECIES #3 _____ Total # of individuals: _____

Talley (include measured fish): _____

LENGTHS: _____

SPECIES #4 _____ Total # of individuals: _____

Talley (include measured fish): _____

LENGTHS: _____

SPECIES #5 _____ Total # of individuals: _____

Talley (include measured fish): _____

LENGTHS: _____

BIRD SURVEYS

SUMMARY

The bird guilds using salt marsh areas within Region 5 that may be affected by OMWM practices include: (1) cryptic marsh passerine species such as marsh wrens and salt marsh sparrows (Seaside, Coastal Plain, Swamp, and Sharp-tailed primarily) and non-passerine rails and bitterns; (2) conspicuous, large waterbirds such as waterfowl (ducks, geese, and swans), colonial species such as herons and egrets, gulls, terns, Black Skimmers, and Double-crested Cormorants; and (3) migrating and wintering shorebirds, including sandpipers, plovers, and relatives. Many of these species are of high priority in state and national bird conservation plans, Partners in Flight, and Region 5 FWS. These groups however require different survey methods.

Because previous work on breeding salt marsh sparrows had begun in New England in 1998, the same protocol used then, developed by James Gibbs and Greg Shriver, SUNY-Syracuse, should continue at the northern refuges, Parker River and Rachel Carson NWRs. It is probably necessary to add points specifically in the OMWM study areas at these northern refuges, although usually only one point could fit into each treatment area (points have a 100m radius, and adjacent points must be 300 m apart, according to the Shriver protocol). At ALL refuges, starting in 2002 or 2003, the marsh sparrow surveys will be combined with those for the other bird groups. The surveys will be conducted both in the breeding and non-breeding seasons. Although the same fixed walking route (see details below) can be used as with the Group 2 and 3 species, the bird survey method varies somewhat.

Methods: Group 1 Species:

- A *trained* observer should conduct the survey (see Training section below). If possible, having a second person as a recorder is desirable.
- The observer should maintain a slow pace following a flagged walking route, recording all *passerine bird species* (sparrows, wrens) seen (flushed or perched) or heard within a 25 m distance from the survey line.
- Detections that are visual (V) or auditory (A) should be recorded on the form. Auditory detections will be the most critical for a few of the marsh passerines as well as the rails and bitterns.
- For birds that flush, try to mentally record their new location to avoid double-counting individuals. This method will certainly underestimate Sharp-tailed Sparrows which are not vocal (except during periods after spring tides) and are non-territorial, however it should be more effective for Seaside and Swamp sparrows.
- Because the surveys will be conducted with the larger Group 2-3 species and will focus on low-tide periods, time of day will vary from morning to afternoon. (Again, this is not optimal for passerine surveys, but Seaside and Swamp sparrows are very vocal species in the breeding season).

- The order of visitation of the treatment and control areas should be varied each survey, so that no systematic time bias results; *e.g.*, avoid going to only controls early, treatments late in the day.

Methods Groups 2 and 3 Species:

- A four-season survey design is desired.
- At least 5 replicates for each season should be performed: Spring/breeding (May 10 – June 30), late Summer (July 20 – September 10), late fall (October 15 – December 10) and Winter (January 10 – March 10).
- For these groups, detailed maps of the study area (2 + ha plot) should be used to set up the survey route and discrete habitats (creek segment, panne, pond, OMWM ditch) should be marked.
- Later, data analyses will require converting bird estimates to densities using both water (or basin) area and total plot area.
- Surveys should be timed to coincide with falling (>3hrs past high tide) or low tide (2hr before to 2hr after low), during daylight hours between 1hr after sunrise to 1 h before sunset.
- A fixed point should be established along one side of the area, preferably elevated (blind or scaffolding), that will allow the observer to use a spotting scope (15-20 X) and scan the entire area.
- All birds seen or heard are recorded, including those flying over the area *that are feeding in the area, not simply transiting over* (includes northern harriers, tree swallows, peregrine falcon, *etc*.).
- Birds should be assigned to one of the habitats listed on the form.
    - Water bodies without daily tidal influence are considered "nontidal," therefore plugged ditches would be nontidal.
    - Unplugged ditches would be tidal.
    - For the aerial feeders, the habitat should be "Air".
- It is easiest to cover different sectors of the plot in sequence (*e.g.*, N to S) recording all species by habitat use.
- We recommend using tick marks in the field form block as sectors accumulate, then in each block, at the end of the survey (10min minimum to 15min maximum) total the ticks and circle the number (see field sheet example).
- The notation of "FP" should be used in the left-hand column for all birds detected (A or V) from the fixed point observation.
- At the end of the 15min, draw a line across the form under the line for the last species entered. This will prevent confusion between entries for the FP and the walking route parts of the survey.
- A walking route (WR) transect marked with wire flagging (different colors than those used for marking nekton stations and vegetation plots) should be followed to survey each water body, panne, ditch, or creek as potential habitat.
- At regular intervals along the WR, GPS locations should be recorded in case flags disappear.

- Water levels in ditches and ponds should be crudely estimated and averaged for the entire plot area, not each water body (scores of 1-4, where 1=<25% basin full, 2=25-50% *etc*.); this will allow more refined analyses of bird densities.
- The observer should maintain a slow, steady pace, but should stop at larger water bodies to record, or at long tidal ditches to inspect for rails, and cryptic birds with binoculars.
- The observer should record the number of individuals of each species within a given habitat, with careful attention noted of species movements; *i.e.*, avoid double counting birds that flush ahead and land in the next location visited.
- As before, we recommend using tick marks in each habitat block as one walks along, then totaling and circling the number in each habitat block after the transect is completed.
- Because the surveys are conducted around low tides, most birds are expected to be using ditches, creek banks, or pools/pannes.
- Also recorded should be those species that feed aerially (Habitat = Air), such as harriers, terns, falcons. (NOTE: We no longer recommend recording behavior during the surveys).
- The data should be recorded on field forms (Table 7) and if possible entered the same day in electronic format.
- A second individual should cross check the field forms and electronic database. The data should be maintained in a Microsoft Access database format that is being developed by PWRC personnel in consultation with other personnel involved with the OMWM project.

Training

- Observers need to be able to identify all species of waterbirds and marsh birds by sight and sound. If necessary, they may need to listen to tape recordings of certain species (rails, sparrows) or may require field reinforcement with an expert. Tapes can be provided by the USGS Patuxent Wildlife Research Center if necessary.
- Ideally, at the beginning of each refuge study, a local expert should be used to conduct a series of 20-30 point counts (radius 25m) at marsh pannes, tidal creeks, impoundments, and emergent marsh plots. Accompanying the expert should be the refuge biologist and field technicians expected to conduct the bird surveys.
- They should record on field forms all the birds seen or heard during 10min point counts, keeping records independently and not talking during the sessions.
- After the sessions, a tally of total species and numbers should be compared among the observers.
- A "qualified" field observer must have identified > 90% of the total species correctly, and > 80% of the total numbers of individuals compared to the "expert."
- If not, that individual should receive more training and be retested, or not used.
- We prefer this method to the much more time-consuming alternative of the double-observer method where two observers are required on every survey and where a rigorous data collection protocol is required using primary and secondary observers.

- We *strongly* encourage that data collection for birds be conducted by the *same observer at all the sites* and that the same individuals be used between years to reduce observer variability, a major source of variation.
- Observers should be reinforced using markers at 10, 20, 25, …up to 50m distances so that they may accurately determine bird detections within 25m of the transect.
- Stakes should be set out at these fixed intervals in one field site.
- During training, objects can be placed at varying distances for observers to estimate. After estimating each distance, they are then told the actual distance, and the two distances recorded for side by side comparison.
- If the observer can consistently (> 90% of occurrences) estimate within 5 m of the 20-30 distances, he/she should be considered qualified.

Instructions for Filling Out Bird Survey Field Data Sheet

These instructions are to be used for the OMWM waterbird data surveys. For continuation of the special marsh sparrow surveys at Rachel Carson and Parker River, Greg Shriver has provided the protocol for those, begun in 1999, and data for New England is being compiled by Greg Shriver.

OMWM WATERBIRD DATA: We have set up the field data forms in a manner that will be most efficient when entering into Microsoft Access. We plan on creating a data entry form in Access that will look very much like your field datasheets. Many of the fields on the Access data entry form will contain bound dropdown lists (for example, when you enter your bird species codes, there will be a set list that you must choose from) and automated calculations (total survey minutes will be calculated; bird totals will be summed across the habitat types and compared with your bird totals as a data check). Migration from the field datasheets to electronic form (Access) will be done at each refuge, and then forwarded to Mike Erwin's group.

TOP SECTION OF OMWM DATA FORM – Survey Description

- Refuge Name and Site Code: This is a combination of Refuge Code and Site Code. For example, Prime Hook Petersfield Treatment would be PHPT. Refer to Table 8 in the section entitled "Data Headers for Spreadsheets" for site codes.
- Date: two digit month and day, four digit year (e.g. 05/24/2001)
- Observer: Use 3 initials.
- Time: First conduct a Fixed Point (FP) survey at each unit where one vantage point can cover the entire unit (elevated if possible). We decided to allow a 10-15 min window depending on numbers of birds and location. Do not exceed 15min. Time of day is not being standardized, but tide is. Use military time (*i.e.*, 1:00 pm = 1300h). For walking surveys, try to stay within about 30 minutes. This will vary among survey units, but keep a slow, steady pace. The emphasis is on WATERBIRDS using ditches and waterbodies; other birds using marsh grasses

are to be recorded within 25m of the transect. Total survey times will be calculated by Access, based on the times that you enter in these fields.

- Time of Low Tide: Record the predicted time of low tide. Of course, factors may differ for each survey, depending on local conditions (wind, *etc*.). It is beneficial to note water levels on the main data form (Water Level column, second from the right edge of the form) at each waterbody surveyed (see Water Level below).
- Temperature: in degree F.
- Wind speed: Check with nearest weather station, USCG, or military airport if possible. Windspeed codes are listed on the datasheet: L(<5mph), M(6-12mph), S(>12mph).

MAIN SECTION OF OMWM DATA SHEET – Bird Data

- Survey type: Use abbreviations FP for Fixed Point and WR for Walking Route. When transitioning from conducting the Fixed Point to the Walking Route, draw a line sectioning off all FP data, and below the line write WR to start the WR section of data.
- Species: On your field datasheets, you may record the bird species in whatever way is best for you, although we recommend using the ABA 4-letter codes. When the data is entered in Access, a drop down list containing 4 letter codes and the species common name will be provided in this field so that all refuges will be using the same 4-letter codes. Abbreviation conventions are as follows: For common names with one word, take the first 4 letters, example: Dunlin = DUNL; common names with 2 words, take the first 2 letters from each word, example: Greater Yellowlegs = GRYE; For multiple species confusion, use the following conventions: For small unidentified sandpipers (the Least, Semipalmated, Western complex), use "PEEP"; for small terns (Forster's or Common), use TERN, for unidentified dowitchers, use DOWI; for Greater or Lesser Yellowlegs, use YELL; for unidentified sparrow, use SPAR; for unidentified (usually immature) gulls, use GULL.
- Total Number of Birds: Tally this up at the end of the session. We suggest using tick marks in pencil as you count birds by species in each habitat type. After the survey is complete, add up the ticks in each cell, write the total in the cell, and circle it. The sum of all cells in a row is what you should write in the Total Number of Birds Column. We will use this column as a data check (it will be compared with a sum that Access calculates for each row).
- Habitat types: Self explanatory. To avoid double counting of individuals, record individuals in the first habitat that they were seen in ONLY (if a dunlin moves from a tidal pond to a ditch, record it in the tidal pond only). For analysis, we will create GIS layers and each habitat type will be measured for area. When analyses are underway, we will convert total numbers of birds by habitat type into density values. **For birds seen in the air, only list species that are aerially feeding or hunting, not those obviously flying over. For example, you may list a northern harrier that is hovering**.

- A or V: Record whether a bird was observed audially or visually. You don't need to do this for the waterbirds, but please be sure to distinguish for the sparrows and secretive species such as rails and bitterns.
- Water Level: Note water levels in each waterbody (ditch, tidal pool, panne, *etc*.) using the codes provided on the sheet (1=25% full, 2=25-50%, *etc*.).
- Notes: Record any anomalies such as disturbances, use of heavy equipment, *etc*. Because the cell is pretty small, you might have to write over a couple of cells; be sure to demarcate which record the note refers to.

PAGE 2 - MAIN SECTION OF OMWM DATA SHEET – Bird Data
Use this if you fill up the first sheet. Be sure to fill in the page number and the total number of pages for that survey in the top right corner (*e.g.*, page 2 of 2).

Table 7. Sample bird survey field data sheets. The second data sheet is to be used if the first sheet is filled.

OMWM WATERBIRD DATA FORM

page ___ of ____

Refuge Name and Site Code ___________________________ Time of Low Tide _________________________
Date (mo/da/yr) ___________________________________ Temperature (F) _________________________
Observer Initials ___________________________________ Wind Speed ___________ L(<5mph), M(6-12mph), S(>12mph)
Time(military) **Fixed Point:** Start___________End_______________ **Walking Route:** Start____________ End _________________

Survey Type Fixed Point (FP) or Walking Route (WR)			HABITAT TYPE													
	SPECIES (4 letters)	Total # Birds	Tidal pool	Non Tidal Pool-panne	Creek (natural)	Ditch (Open)	Ditch (Plugged)	Low Marsh (S.altern.)	High Marsh (S.patens)	Phrag-mites	Woody Veg.	Air	Other:	A or V (Audial or Visual Detection)	Water Level 1=<25% 2=25-50% 3=50-75% 4=75-100%	Notes (Disturbance, etc.)
			*** To avoid double counting of individuals, record numbers within habitat where they were first observed ONLY.													

Table 7. continued.

OMWM WATERBIRD DATA FORM

Page ___ of ____

Refuge Name and Site Code __________________ Date (mo/da/yr) _________________________ Temp.(F)__________

Survey Type Fixed Point (FP) or Walking Route (WR)	SPECIES (4 letters)	Total # Birds	HABITAT TYPE											A or V (Audial or Visual Detection)	Water Level 1=<25% 2=25-50% 3=50-75% 4=75-100%	Notes (Disturbance, etc.)
			Tidal pool	Non Tidal Pool-panne	Creek (natural)	Ditch (Open)	Ditch (Plugged)	Low Marsh (S.altern.)	High Marsh (S.patens)	Phrag-mites	Woody Veg.	Air	Other:			
			*** To avoid double counting of individuals, record numbers within habitat where they were first observed ONLY.													

OTHER ENVIRONMENTAL VARIABLES MONITORED DURING NEKTON SAMPLING

Once nekton is removed from a trap or ditch net sample, environmental variables can be measured. Measures of water temperature, salinity, water depth, and plant cover are essential environmental data to collect in conjunction with each throw trap and ditch net sample. Some investigators may elect to also collect other variables. Sediment composition (*e.g.* grain sizes and organic content) can be measured by extracting sediment cores and then processing according to Dean (1974). This information helps describe the habitat available to the nekton. Other variables such as creek width, creek order (*e.g.*, 1st order, 2nd order), pond size, adjacent shoreline type, distance of seagrass bed to shoreline, are easy measures and can enhance interpretation of the nekton data.

The following water quality measurements should be taken from an area of the pond or ditch that has not been disturbed (*e.g.* sediment re-suspension) from the throw trap or ditch net activity.

- Water temperature, to the nearest degree C, is measured using a stick thermometer or temperature probe.
- Salinity is measured, to the nearest part per thousand, using either a refractometer or water quality probe.
- Dissolved oxygen is measured using a dissolved oxygen probe.
- Water depth within the trap is measured to the nearest cm using a meter stick. Alternatively, the sides of the trap can be marked off in centimeters and readings taken directly from the trap. The trap is often located on an uneven bottom, and thus, depth should be measured near each corner of the trap to obtain a mean depth value. Water depth is a simple measure and is useful for documenting changes in water depth over time. When monitoring restoration sites, where hydrology has been altered, this is a particularly important measure.
- Water depth in the ditch is measured after the net is removed from the ditch. The depth of the ditch is also measured.
- If macroalgae, marsh grass, or eelgrass are present within the trap, cover and species composition should be quantified. **Prior to dip netting for nekton**, the percent cover of each plant species should be visually estimated according to the following cover class categories (<1% cover, 1-5%, 5-25%, 25-50%, 50-75%, >75%). These data provide a measure of the complexity of habitat available to the estuarine nekton. If sampling in regions with turbid waters and the vegetation can not be seen, then vegetation should be quantified by a biomass technique after Raposa and Oviatt (2000).

DATA MANAGEMENT AND QUALITY ASSURANCE AND QUALITY CONTROL

Field data should be recorded in waterproof notebooks or on datasheets that are previously developed and printed on waterproof paper. Datasheets can be organized to the preference of individual researchers, but should include all information described previously in these protocols (*e.g.*, study site, date, station identification, habitat, species name, total number of individuals captured by species, lengths, comments, and environmental parameters). Examples of sample datasheets are provided after each protocol.

- Be sure to write legibly on the datasheet.
- Make sure that all sections of the data sheet are filled out before moving on to the next sampling station.
- If a variable cannot or was not measured (*e.g.* instrument failure, fish escaped) explain on the data sheet for that station the reason for missing data. Record the reason/explanation in the field. Do not wait until you return to the office to record the reason or explanation.
- All field data should be transferred to digital format soon after sampling. Field data are easily incorporated into common spreadsheet programs that are designed for comprehensive data management.
- After the data are entered it is important to carefully check the data for typos and mis-entries to insure the data are correct and to maintain quality assurance and quality control of the data.

DATA HEADERS FOR SPREADSHEETS

The following are the suggested column headers for entering data into computer spreadsheets. Column headers should be limited to 8 characters in length. Refuge and site codes given in Table 8 should be used for all data types. The usual naming convention for species (except for birds, see section entitled "Instructions for Filling Out Bird Survey Field Data Sheet") is to use the first 3 letters of the species, an underscore for the 4th character, and the first 4 letters of the genus. A separate key to all species codes should be kept (preferably in another spread sheet page) with the electronic data.

Table 8. Site codes for Refuges and study areas. Note: Some codes are not yet available (n/a).

Refuge	Refuge/Site Code	Site Name
Rachel Carson	n/a	n/a
Parker River	PR_A	Parker River Area A (plugged)
	PR_B1	Parker River Area B1
	PR_B2	Parker River Area B2
	PR_C	Parker River Area C
S.B. McKinney	n/a	n/a
Long Island Complex	LI_FT1	Long Island, Flanders Site 1, Treatment
	LI_FT2	Long Island, Flanders Site 3, Treatment
	LI_FC	Long Island, Flanders Site 2, Control
	LI_WTE	Long Island, Wertheim East, Treatment
	LI_WTW	Long Island, Wertheim West, Treatment
	LI_WC	Long Island, Smith Point, Wertheim Control
E.B. Forsythe	F_OC	Forsythe, Oyster Creek Control
	F_OT	Forsythe, Oyster Creek Treatment
	F_ATTC	Forsythe, AT&T Control
	F_ATTT	Forsythe, AT&T Treatment
Cape May	n/a	n/a
Prime Hook	PH_PC	Prime Hook, Petersfield Control
	PH_PT	Prime Hook, Petersfield Treatment
	PH_SC	Prime Hook, Slaughter Control
	PH_ST	Prime Hook, Slaughter Treatment

Vegetation Data Headers

All vegetation data for a refuge should be entered into a single spread sheet (*i.e.*, do not use separate spreadsheets for each study area). The first and second columns identify the study area and the vegetation plot. The remaining columns are for the vegetation cover types that were observed at all study locations. It is helpful to first make a master list of all vegetation cover types that were observed within the Refuge study areas. Be sure to make a list of all vegetation cover type codes used and what they represent, so that others can interpret the codes. In each cell under the appropriate cover type for that plot, the total tally of "hits" would be entered. Table 9 shows an example of vegetation data entered for study sites at Prime Hook NWR for Slaughter Control (PH_SC) plot 2-40 and Petersfield Treatment (PH_PT), plot 1-100. At Slaughter Control, plot 2-40, *S. alterniflora* (Spa_alte) had 14 hits, *S. patens* (Spa_pate) had 45 hits, and bare ground had 3 hits, while at Petersfield Treatment, plot 1-100 had 50 hits of *Phragmites australis* (Phr_aust) and 15 hits on bare ground. Note that since all vegetation data for a refuge are entered into one spreadsheet, entries of "0" are entered if that species was not present at a particular plot. Only four species are shown in this example, however there will be more species for any particular Refuge.

Table 9. Example of vegetation data entry.

Study Site	**Plot ID**	**Spa_alte**	**Phr_aust**	**Spa_pate**	**Bare**
PH_SC	2-40	14	0	45	3
PH_PT	1-100	0	50	0	15

Water Table Level and Soil Salinity Data Headers

Headers for data entry are shown in Table 10. Calculations can be done by hand and entered on the spreadsheet or can be entered as additional columns and the spreadsheet program can perform the calculation. For example, water table level is calculated as the top of the well measurement (A) minus the top of the well to the marsh surface (B). In the example below only the finished calculation is shown. Note that at Flanders treatment 1 (LI_FT1), stations 1-00 and 1-30, the water table depth was negative since the water table was below the surface, but at Wertheim control (LI_WC), station 1-60, there was water on the surface of the marsh and therefore the water table level was positive number. Missing data are entered as a "." as at LI_WC, station 1-90.

Table 10. Example of water table level and soil salinity data entry.

Date	Site	Station #	Water Table Depth (cm)	Soil Salinity (ppt)	Depth of Soil Salinity (cm)
7/5/2001	LI_FT1	1-00	-2.8	25	15
7/5/2001	LI_FT1	1-30	-14.9	26	45
7/5/2001	LI_WC	1-60	3.7	20	15
7/5/2001	LI_WC	1-90	.	.	.

Mosquito Larvae Data Headers

Mosquito data headers are shown in Table 11. If the station is dry, then a missing value represented by "." should be entered as the number of larvae observed. If water was present at the station, but no larvae were in the dipper, then "0" is entered for the number of larvae observed. For example, at Forsyth NWR's Oyster Creek control site (F_OC), station 4-120, the station was dry and since there was no water present the number of larvae is entered as ".". At the AT&T control site (station 1-40) water was present, but no larvae were in the dipper, so a "0" is entered. Missing values are entered for dry stations so as not to bias the counts by including habitats that are not appropriate for larval mosquitoes. If larvae were present, then enter "yes" or "no" in the appropriate species column indicating what species were found. This information will be entered after the larval samples have been identified. Be sure to keep information on species identification with the appropriate Mosquito Larvae Sampling Data Sheet. Only two mosquito species are shown for example purposes, it may be necessary to add more species columns. Note that codes for the amount of water volume in the dipper are entered instead of the actual volume.

Nekton Data Headers

Nekton data from both collection methods (throw trap and ditch net) are entered on the same spread sheet. Nekton data require two different spreadsheets since one data type deal with station information and the other with individual nekton (length) information. Table 12 illustrates the header information for example data from Parker River NWR. Note that the station ID also identifies the type of gear. For example, P1 stands for Pond 1 which is sampled with a throw trap, whereas D2 is a ditch net station which is sampled with a ditch net. Environmental variables such as water depth, creek depth, tide phase, water temperature, salinity, and DO are also entered. For throw trap samples, a missing value represented by "." is entered for the creek depth since only the water depth of the pond is measured. Habitat type for ditch nets should be entered as TC (tidal creek), OD (open ditch) or PD (plugged ditch). Habitat type for throw traps is always "pond". The area of the ditch net is from the measurements of the net (refer to section "Calculating the Area of a Ditch Net"). The area of throw trap samples is always $1m^2$. The remaining columns are for the nekton species that were observed at all study locations. The total count of individuals for that station is entered under each appropriate species. Note that species names are coded into an 8 letter code. Be sure to include a key to the codes with the database. If a nekton species was not present, represent that by a "." not "0" as this will influence the final density calculation. Density is calculated as the sum total number of individuals per trap or ditch net divided by the area of the trap or net.

The individual nekton (fish and decapods) data are entered in another spreadsheet. Headers for individual nekton data are shown in Table 13. In this database there is one "species" column and the species coded name is entered and the corresponding length of that fish is entered in the corresponding cell under "length". Carapace width (for crabs) and total length (for shrimps) are also entered under the length. Note that *Fundulus heteroclitus* adults (Fun_hetA) and juveniles (Fun_hetJ) are given separate species codes. Be sure to include a key to the codes with the database.

Bird Survey Data Headers

An Access Database with drop down menus has been designed for bird survey data entry by Diann Prosser, USGS, Patuxent Wildlife Research Center. All refuges will receive a copy of this database for bird survey data entry.

Table 11. Example of mosquito data entry. Note that the Och_soli and Och_cant stand for *Ochlerotatus sollicitans* and *O. cantator* which were formally *Aedes sollicitans* and *A. cantator*.

Date	Site	Station #	Total # of larvae	Dipper volume	Area dipped	Larvae ID'ed?	Och_soli	Och_cant
7/02/02	F_OC	4-120	.	.	dry	no	.	.
7/05/02	F_OT	3-40	72	3	panne	yes	yes	no
7/06/02	F_ATTC	1-40	0	2	pool	no	.	.

Table 12. Example of nekton data entry.

Date	Site	station	Water depth (cm)	Creek depth (cm)	tide	habitat	Area (m^2)	Water Temp	Salinity (ppt)	DO (mg/L)	Fun_hete	Cyp_vari	Men_bery
6/1/01	PR_B1	P1	50	.	ebb	pond	1	26.9	29.8	7.36	1	.	.
8/15/01	PR_A	D2	46	48	ebb	OD	0.409954	25.8	27.8	5.10	.	.	.

Table 13. Example of individual nekton data entry.

Date	Location	Station	Species	Length (mm)
9/28/01	PR_C	P6	Fun_hetA	50
9/28/01	PR_C	P6	Fun_hetA	50
9/28/01	PR_C	P6	Fun_hetJ	45
9/28/01	PR_C	D3	Fun_hetA	79
9/28/01	PR_C	D3	Fun_hetA	76
7/12/01	PR_B2	D5	Car_maen	15
7/12/01	PR_B2	D5	Pun_pung	40

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Appendix B

Properties and Behaviors of Soils On-line Lab Manual, Chapter 3

Hossain et al. (2021)

CHAPTER 3.

SIEVE ANALYSIS

INTRODUCTION

The grain size analysis test is performed to determine the percentage of each size of grain that is contained within a soil sample, and the results of the test can be used to produce the grain size distribution curve. This information is used to classify the soil and to predict its behavior. The two methods generally used to find the grain size distribution are:

- Sieve analysis which is used for particle sizes larger than 0.075 mm in diameter and
- Hydrometer analysis which is used for particle sizes smaller than 0.075 mm in diameter

Sieve analysis is a method that is used to determine the grain size distribution of soils that are greater than 0.075 mm in diameter. It is usually performed for sand and gravel but cannot be used as the sole method for determining the grain size distribution of finer soil. The sieves used in this method are made of woven wires with square openings. The list of the U.S. standard sieve numbers with their corresponding opening sizes are provided in Table 3.1.

Table 3.1: U.S. Sieve Size

Sieve No.	Opening (mm)	Sieve No.	Opening (mm)
4	4.75	35	0.500
5	4.00	40	0.425
6	3.35	45	0.355
7	2.80	50	0.300
8	2.36	60	0.250
10	2.00	70	0.212
12	1.70	80	0.180
14	1.40	100	0.150
16	1.18	120	0.125
18	1.00	140	0.106
20	0.85	200	0.075
25	0.71	270	0.053
30	0.60	400	0.038

PRACTICAL APPLICATION

- This test method is used primarily to grade aggregates. The results are used to determine the compliance of the particle size distribution with applicable specification requirements and to

provide necessary data for controlling the production of various aggregate products and mixtures containing aggregates.

- The data may also be useful in developing relationships concerning porosity and packing. Information obtained from the particle size analysis (uniformity coefficient C_u, coefficient of curvature, C_c, and effective size, D_{10}, etc.) is used to classify the soil.
- Particle size is one of the criteria used to ascertain whether the soil is suitable for building roads, embankments, dams, etc.
- Information obtained from particle size analysis can be used to predict the soil-water movement if the permeability test is not available.

OBJECTIVE

- To obtain the grain size distribution curve for a given soil sample.

EQUIPMENT

- Stack of sieves with a cover,
- Mortar and pestle or a mechanical soil pulverized
- Balance, sensitive to 0.1 g
- Oven
- Mechanical sieve shaker
- Brush

STANDARD REFERENCE

- ASTM D6913: Standard Test Methods for Particle-Size Distribution (Gradation) of Soils Using Sieve Analysis.

METHOD

1. Obtain a representative oven-dried soil sample.

Figure 3.1: Weighing some representative oven dried samples

Figure 3.2: Washing the sieves before the test

2. Pulverize the soil sample as finely as possible, using a mortar and pestle or a mechanical soil pulverizer.
3. Obtain a soil sample of about 500 g and determine its mass W_0 (g).
4. Stack the sieves so that those with larger openings (lower numbers) are placed above those with smaller openings (higher numbers). Place a pan under the last sieve (#200) to collect the portion of soil passing through it. The #4 and #200 sieves should always be included in the

stack.

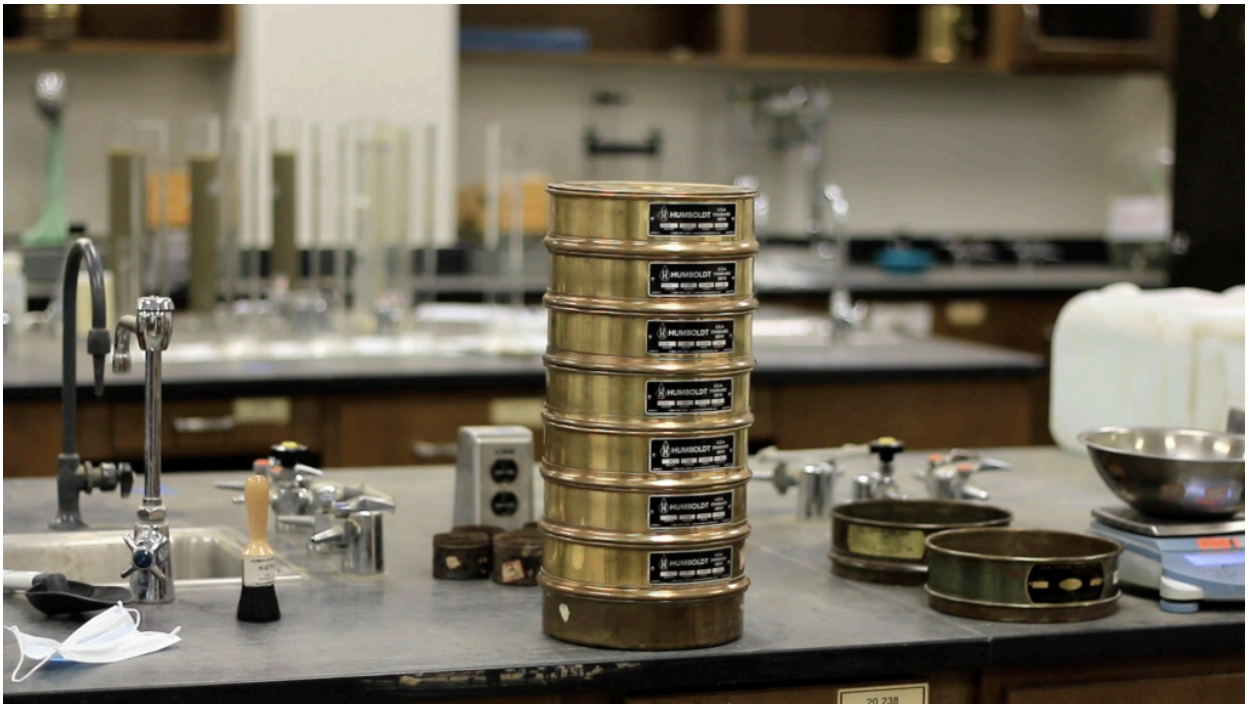

Figure 3.3: Stack of sieve in order

5. Make sure the sieves are clean, If soil particles are stuck in the openings, use a brush to poke them out.

Figure 3.4: Pouring the soil sample at the top of the sieves

6. Weigh the pan and all of the sieves separately.

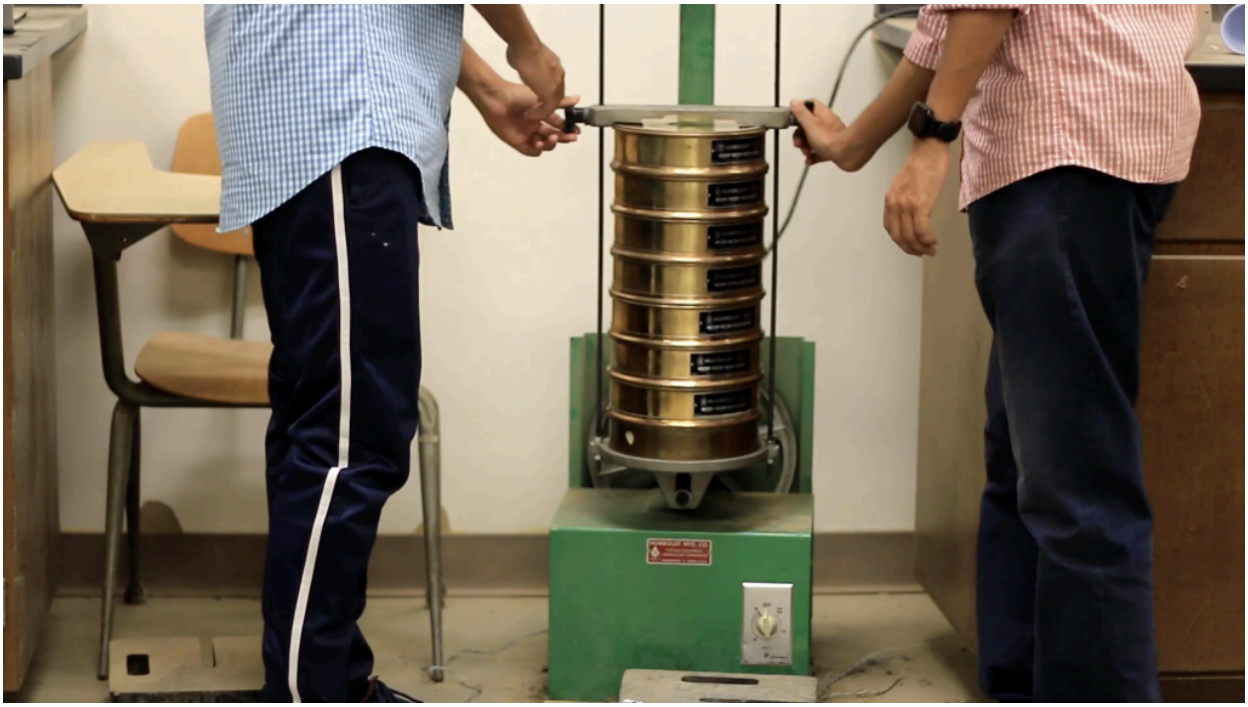

Figure 3.5: Sieve shaker

7. Pour the soil from above into the stack of sieves and place the cover on it. Put the stack in the sieve shaker, affix the clamps, set a timer for 10 to 15 minutes, and start the shaker.

Figure 3.6: Weighing of each sieve after shaking

8. Stop the sieve shaker and measure the mass of each sieve and retained soil.

VIDEO MATERIALS

LECTURE VIDEO

A PowerPoint presentation is created to understand the background and method of this experiment.

One or more interactive elements has been excluded from this version of the text. You can view them online here: https://uta.pressbooks.pub/soilmechanics/?p=178#oembed-1

DEMONSTRATION VIDEO

A short video is executed to demonstrate the experiment procedure and sample calculation.

One or more interactive elements has been excluded from this version of the text. You can view them online here: https://uta.pressbooks.pub/soilmechanics/?p=178#oembed-2

RESULTS AND DISCUSSIONS

Sample Data Sheet

Sieve No	Opening (mm)	Sieve Wt. (gm)	Sieve + Soil Wt. (gm)	Wt. of soil retained (gm)	Percent retained	Cumulative percent retained	Percent finer
4	4.75	521	521	0	0	0	100
8	2.36	491.8	504	12.2	4.07	4.07	95.93
16	1.18	426	450.5	24.5	8.17	12.24	87.76
30	0.60	401.8	490	88.2	29.4	41.64	58.36
50	0.297	375.5	478	102.5	34.17	75.81	24.19
100	0.149	355.3	410	54.7	18.23	94.04	5.96
200	0.075	351.1	368.2	17.1	5.7	99.74	0.26
Pan	-	364.2	365	0.8	-	-	-

Sample Calculation

For #8 sieve,
Sieve weight = 491.8 gm
Sieve + soil weight = 504 gm
Weight of soil retained = (504 – 491.8) = 12.2 gm
Percent retained= $2.2/300 × 100 = 4.07%
Cumulative percent retained= 0 + 4.07 = 4.07%
Percent finer= 100 – 4.07= 95.93%

The grain-size distribution of the soil sample can be obtained by plotting the percent finer with the corresponding sieve on semi-log graph paper, as shown below. An example of the grain-size distribution curve is shown in Figure 3.7.

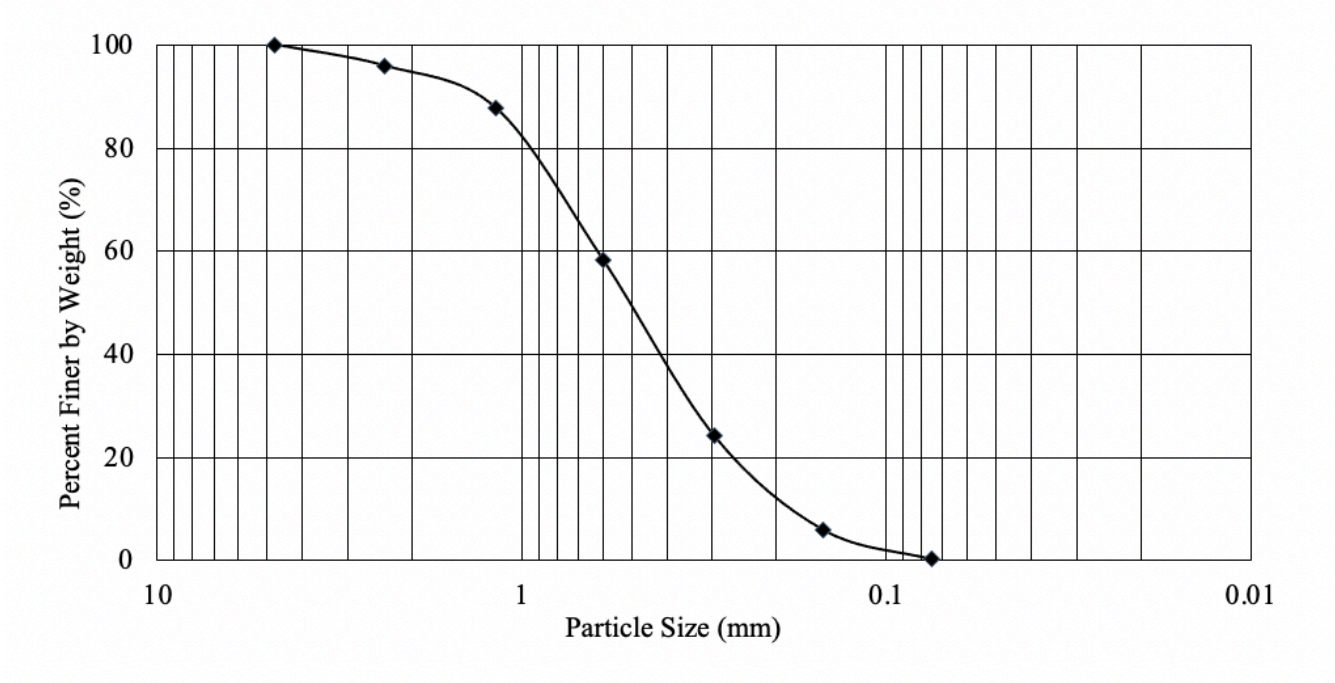

Figure 3.7: Particle size distribution curve

he values of D_{10}, D_{30}, and D_{60}, which are the diameters that correspond to the percentfiner of 10%, 30%, and 60%, respectively can be determined from the grain-size distributioncurve. The values of the uniformity coefficient C_u and the coefficient of gradation C_c can be calculated using the following equations:

$$C_c = \frac{D_{30}^2}{D_{60} \times D_{10}}$$

$$C_u = \frac{D_{60}}{D_{10}}$$

The values of C_u and C_c are used to classify whether the soil is well-graded or not. Sand isconsidered well-graded, if C_u is greater than 6 and C_c is between 1 and 3. For gravel to be considered as well-graded, C_u should be greater than 4 and C_c should be between 1 and 3.

From Figure 3.5,

D_{10}= 0.18, D_{30}= 0.35, and D_{60}= 0.61

Uniformity coefficent, C_u=D_{60}/D_{10}=0.61/0.18=3.39

Coefficent of gradation, C_c= $(D^2_{30})/(D_{60}\times D_{10})$=$(0.35)^2/(0.61\times0.18)$ =1.12

Blank Data Sheet

Sieve No	Opening (mm)	Sieve Wt. (gm)	Sieve + Soil Wt. (gm)	Wt. of soil retained (gm)	Percent retained	Cumulative percent retained	Percent finer
4	4.75						
8	2.36						
16	1.18						
30	0.60						
50	0.297						
100	0.149						
200	0.075						
Pan	-						

REPORT

Use the template provided to prepare your lab report for this experiment. Your report should include the following:

- Objective of the test
- Applications of the test
- Apparatus used
- Test procedures (optional)
- Analysis of test results – Complete the table provided and show one sample calculation. Draw the grain size distribution curve. Calculate C_u and C_c
- Summary and conclusions – Comment on the shape of the grain size distribution curve of the given soil sample. Comment on whether the soil is well graded or poorly graded.

Appendix C

Soil Quality Test Kit Guide

USDA (2001)

United States
Department of
Agriculture

Agricultural
Research Service

Natural Resources
Conservation Service

Soil Quality Institute

Soil Quality Test Kit Guide

July 2001

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Table of Contents

Appendix: **Page**

Section II - Background & Interpretive Guide for Individual Tests:

PREFACE

Soil quality is simply defined as "the capacity of a specific kind of soil to function." It is generally assessed by measuring a minimum data set of soil properties to evaluate the soil's ability to perform basic functions (i.e., maintaining productivity, regulating and partitioning of water and solute flow, filtering and buffering against pollutants, and storing and cycling nutrients). This guide describes a kit of selected field procedures to evaluate or indicate the level of one or more soil functions.

When measuring soil quality, it is important to evaluate the physical, chemical, and biological properties of the soil. Physical properties addressed by the kit include bulk density, water content, infiltration rate, aggregate stability, slaking, and morphological estimations. Biological properties measured include soil respiration and earthworms. Soil chemical properties measured include pH, electrical conductivity (EC), and soil nitrate levels. The chemical tests are also useful to evaluate water quality of well-water, tile drainage waters, and other water bodies related to farm activities.

Section I of this guide provides a list of supplies and instructions for conducting a number of on-farm tests to assess soil quality. Section II provides background and interpretive information for each test described in Section I. These tests, or indicators, are designed as a screening tool to provide immediate results for comparing management systems, monitoring changes in soil quality over time, and for diagnosing possible soil health problems due to land use and management.

These tests can be easily conducted on the farm by NRCS field personnel or by landowners themselves to assess the quality of their soil. Use of the kit allows NRCS staff to be an active participant with the landowner in the assessment of soil health. The assessment will provide the opportunity to discuss management options when the need arises.

The kit was developed by John Doran and associates, Agricultural Research Service, Lincoln, NE. The Soil Quality Institute has continued the development, enhancement and testing of the kit (with NRCS field staff) by adding tests, modifying the manual, and writing an interpretations guide. The NRCS Soil Quality Team in Akron, CO (Manuel Rosales, Josh Saunders, and Mike Sucik) were instrumental in the field testing of the test kit and this guide. The Soil Quality Test Kit Guide is a dynamic document. The Institute welcomes suggestions for additional tests and interpretive information to incorporate in future versions of the guide.

The Institute gratefully acknowledges the contributions of the following individuals: John Doran, USDA-ARS, Lincoln, NE, for the development of the original soil quality test kit from which this guide is based. Bob Grossman, USDA-NRCS, NSSC, Lincoln, NE, for the development of the soil structure index and penetration resistance tests. Jeff Herrick, USDA-ARS, Las Cruces, NM, for the development of the soil slake test procedure and aggregate stability test design. Dennis Linden, USDA-ARS, St. Paul, MN, for the development of the earthworm procedure. Bob Hanafin, Auburn University, for the development of the design and layout of this guide. Cathy Seybold and Lee Norfleet, USDA-NRCS, Soil Quality Institute, for the development of this guide and testing of kit procedures.

The mission of the Soil Quality Institute is to cooperate with partners in the development, acquisition, and dissemination of soil quality information and technology to help people conserve and sustain our natural resources and the environment.

For more information about the Soil Quality Institute and its products and services, visit our website at **http://www.statlab.iastate.edu/survey/SQI/**.

Soil Quality Institute Staff

1. Measuring Soil Quality

Soil quality integrates the physical, chemical, and biological components of soil and their interactions. Therefore, to capture the holistic nature of soil quality or health, all of the parameters in the kit should be measured. However, not all parameters have equal relevance to all soils and situations. For example, the EC test for salinity may not be useful in the eastern part of the U.S. where salinity is not a problem. A minimum data set of soil properties, or indicators, from each of the three soil components are selected based on their ability to indicate the capacity of the soil to function for a specific land use, climate, and soil type. Indicators in the soil quality kit are selected primarily for agricultural soil quality assessments. The kit should be used as a screening tool to give the general trend or direction of soil quality--whether current management systems are maintaining, enhancing, or degrading the soil. Proper use of the kit and interpretation of results depends on how well the indicators are understood with respect to the land use and environmental goals.

There are two fundamental ways to assess soil quality:

- take measurements periodically over time to monitor changes or trends in soil quality;
- compare measured values to a standard or reference soil condition.

By making use of the two ways of assessing soil quality, the kit can be used to:

- make side-by-side comparisons of different soil management systems to determine their relative effects on soil quality;
- take measurements on the same field over time to monitor trends in soil quality as affected by soil use and management;
- compare problem areas in a field to the non-problem areas;
- compare measured values to a reference soil condition or to the natural ecosystem.

Field or Site Characterization

It is important to gain as much information about the area and soils as possible. Indicators of soil quality must be evaluated within the context of site and climatic characteristics. A "**Soil Quality Site Description**" recording sheet, located in the appendix, should be completed during the soil quality assessment. The following are items that should be considered when making an on-farm soil quality assessment:

Soil series - The soil series name can be found in the county soil survey.

Signs of erosion - Signs of erosion include gullies, rills, development of pedestals, exposed areas of subsoil, damage to plants caused by wind blown materials, etc.

Management history - This item includes a description of past and present land and crop management; kind, amount, and method of fertilization; prior tillage; and land leveling.

Slope and topographical features of the field - Record percent slope at the sampling sites within the field, and note any hills, knolls, ridges, potholes, depressions, etc.

Location of the field and sampling areas - Record longitude and latitude (if GPS unit is available), a description of the location (feet from landmarks), and a drawing of the field showing sampling areas.

Climatic information - This item includes precipitation and high and low average tempera-

tures for each month (data from a county or watershed level will often be sufficient).

Location of environmentally sensitive areas - This item includes location of ponds, creeks, wetlands, or other environmentally fragile sites adjacent to the field.

Sampling Guidelines

Important: When, where, and how deep to sample and how many samples to take is primarily dependent on the questions being asked or problems being addressed by the farm or land manager.

When to sample?

Timing of sampling is important, because soil properties vary within a season and with management operations, such as tillage. In general, for the overall assessment of soil quality, an **annual sampling of the field** is recommended. Sampling once a year will allow for the detection of long-term changes in soil quality. A good time of year to sample is when the climate is most stable and there have been no recent disturbances, such as after harvest or the end of the growing season.

Where to sample?

An important consideration in determining where to sample in a field is the variability of the area. Soil properties naturally vary across a field and even within the same soil type. Soil variability across a field is also affected by management operations. General field characteristics to consider are:

- row versus inter-row areas,
- differences in soil type,
- differences in management,
- wheel versus non-wheel tracked areas,
- differences in crop growth,
- salt affected versus non-salt affected areas,
- eroded versus non-eroded areas,
- differences in slope, and
- wet versus non-wet areas (drainage).

Some general guidelines on selecting sampling sites are as follows:

(1) For a general assessment of soil quality, select sample sites within a field that are representative of the field. Refer to soil maps of the area (Soil Survey) to identify soil type differences and variations within the map unit

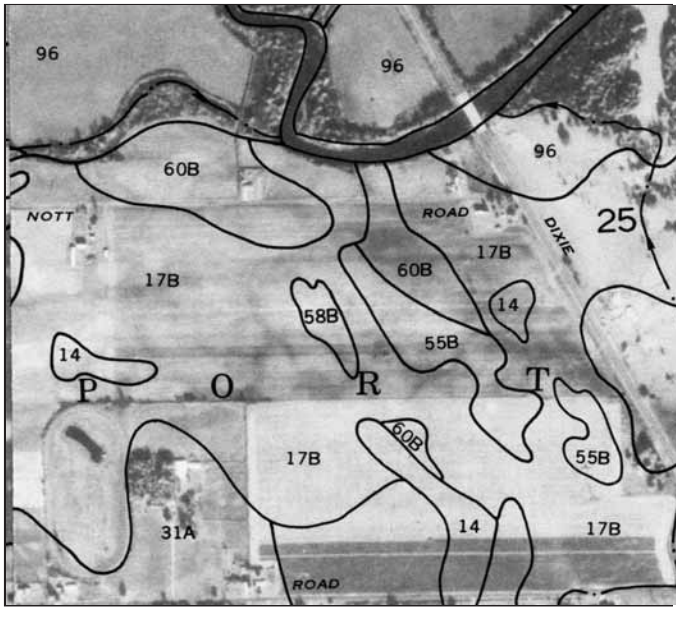


Figure 1.1

(Figure 1.1). A hand auger can be used to make a number of borings to establish locations of the most representative areas of the field.

(2) For assessment of trouble spots within a field, sample areas that are representative of the trouble spots **(Figure 1.2)**.

(3) When comparing management systems, make sure sites selected for comparison have the same soil type and are located on the same topographical features in both fields. For example, if sites are measured in the wheel tracks in one field, wheel tracks sites should be selected in the comparison field.

(4) When monitoring changes in soil quality over time, make sure the same sites within the field are measured at each sampling time. Also, try to take measurements at the same soil moisture conditions at each sampling time to reduce variability.

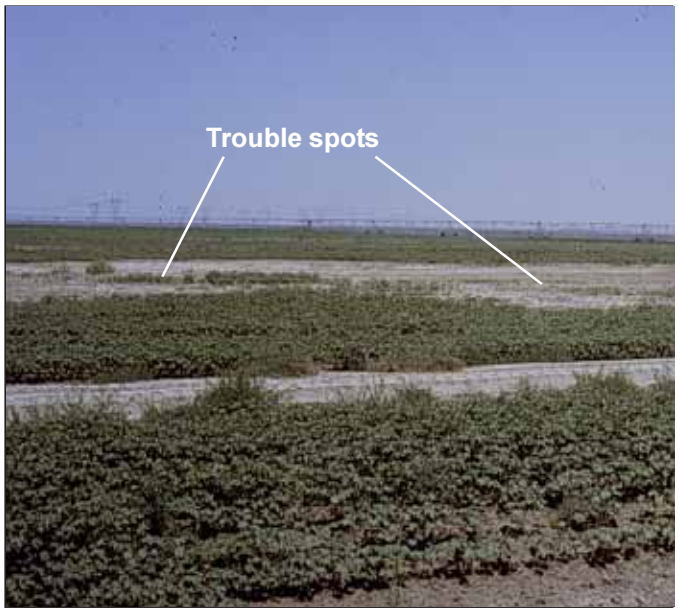


Figure 1.2

In some cases it might be helpful to compare sampling points if the field is sampled at different points across gradients of soil type, soil moisture, slope, or other factors rather than just at a fixed point **(Figure 1.3)**.

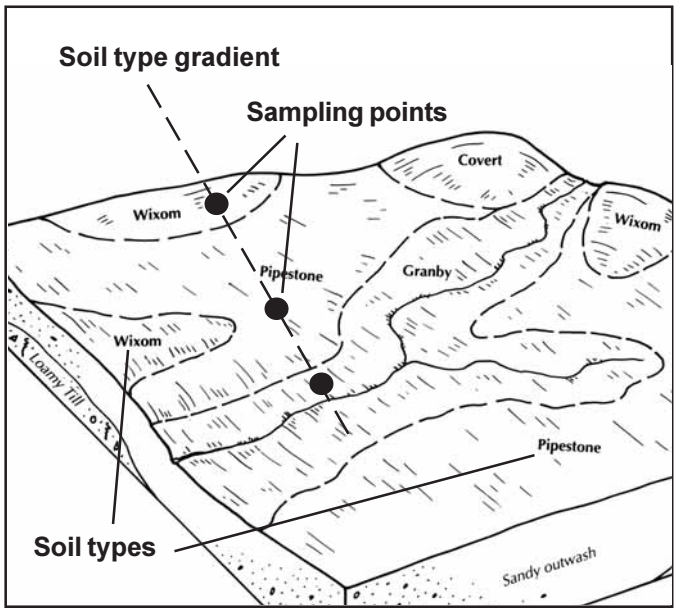


Figure 1.3

How many samples?

The number of samples or measurements to take will depend on the variability of the field. It is recommended that **a minimum of three samples or measurements** be collected on any one soil type and management combination. In general, the greater the variability of the field, the greater the number of measurements are needed to get a representative value at the field scale. When measuring EC, pH, and soil nitrates at the field scale, eight or nine sample cores from across the field could be bulked and mixed, and two subsamples from the mixed cores could be analyzed. When taking cores from across the field, stay away from areas that are distinctly different and are not representative of the field, such as farm lanes and field borders, fertilizer bands, areas within 150 feet of a gravel road, potholes, eroded spots, etc.

2. Soil Respiration Test

For efficient sampling, the soil respiration test is performed first, followed by the infiltration test (Chapter 3) without removing the 6-inch diameter ring. The best time to run the soil respiration test is when soil moisture is at field capacity (the amount of water the soil can hold after drainage). Otherwise, soil respiration should be measured before and after the infiltration measurement or soil wetting (6 to 24 hours after wetting).

Materials needed to measure respiration:

- **6-inch diameter ring**
- **lid with rubber stoppers**
- **hand sledge and wood block**
- **soil thermometer**
- **two sections of plastic tubing**
- **2 needles**
- **Draeger tubes**
- **140 cc syringe**
- **stopwatch or timer**

Did You Know?

Soil breathes! Soil respiration is an indicator of biological activity (i.e., microbial and root), or soil life. This activity is as important to the soil ecosystem as healthy lungs are to us. However, more activity is not always better; it may indicate an unstable system (i.e., after tillage).

Considerations: Microbial activity is greatest when the soil is moist (at or near field capacity). If the soil is dry, a second respiration measurement should be made at a minimum of six hours (preferably 16 to 24 hours later) after the infiltration test or wetting of the soil. If the soil is saturated, soil respiration is inhibited, and this test should not be run.

1 **Drive Ring into Soil**

- Clear the sampling area of surface residue, etc. If the site is covered with vegetation, trim it as close to the soil surface as possible.
- Using the hand sledge and block of wood, drive the 6-inch diameter ring, beveled edge down, to a depth of three inches (line marked on outside of ring) **Figure 2.1.**

Figure 2.1

- If the soil contains rock fragments, and the ring can not be inserted to depth, gently push the ring into the soil until it hits a rock fragment. Measure the height from the soil surface to the top of the ring in centimeters (cm). **[See note below]**

NOTE: For a more accurate measurement of soil respiration, the chamber head-space should be measured. Inside the ring, take four measurements (evenly spaced) of the height from the soil surface to the top of the ring, and calculate the average. Record average on the Soil Data worksheet.

② Cover Ring with Lid and Wait

- Cover the ring with the lid as depicted in **Figure 2.2** and note the time.
- Wait exactly 30 minutes* (to allow CO_2 to accumulate in the chamber).

Figure 2.2

[If this is the SECOND respiration measurement, briefly remove the lid and replace it before timing to allow the release of gases that have built up over the 6-24 hour waiting period. Proceed with Step 3.]

***NOTE: During the 30-minute wait, other tests such as Bulk Density (Chapter 4) can be run.**

③ Insert Soil Thermometer

- Insert the soil thermometer into the soil adjacent to the ring with lid (about one inch away from ring and one inch deep). If the thermometer can easily be inserted into the rubber stoppers, insert it into one of them to a 1-inch depth into the soil.

④ Assemble Draeger Tube Apparatus

- Assemble the Draeger tube apparatus just before the end of the 30-minute wait.
- Connect a needle to one of the sections of tubing.
- Break open **both** ends of a CO_2 Draeger tube, either by using the hole at the end of the syringe handle as depicted in **Figure 2.3**, or by clipping the tube ends with a finger nail clipper.

Figure 2.3

- Connect the Draeger tube to the **other** end of the needle's tubing**.** The arrow on the side of the Draeger tube should point **away** from the needle.
- With the second piece of tubing, connect the Draeger tube to the syringe as shown in **Figure 2.4**

Figure 2.4

5 **Insert Apparatus Needle into Stopper**

After 30 minutes, insert the Draeger tube apparatus needle into a stopper as shown in **Figure 2.5**. Insert a second needle into one of the other stoppers on the lid to allow air flow into the head space during the gas sampling. The second needle should be inserted just before the head space is sampled.

Figure 2.5

6 **Draw Head Space Sample**

Over a 15-second span, draw the syringe handle back to the 100 cc reading (1 cc = 1 mL) as shown in **Figure 2.5.** [If the reading is less than 0.5%, take four additional 100 cc samples of the head space through the same Draeger tube. To do this, disconnect the tube from the syringe to remove the air, and reconnect the tube to the syringe. Take another 100 cc sample. Repeat.]

7 **Record Soil Temperature and % CO_2**

On the Soil Data worksheet, record the temperature in Celsius at the time of sampling. On the Draeger tube, read the "n=1" column if 100 cc was sampled or the "n=5" column if 500 cc was sampled. The % CO_2 reading should be an estimate of the highest point that the purple color can be easily detected. Enter this reading on the Soil Data worksheet. In the example in **Figure 2.6**, the reading would be approximately 0.75%.

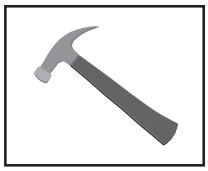
Figure 2.6

8 **Remove Lid**

Remove the thermometer, Draeger apparatus needle, air flow needle, and the lid from the ring.

If this is the **first** respiration measurement, leave the ring in the soil for the **infiltration measurement** (Chapter 3).

Maintenance Tips: Seal any holes in the chamber lid that may cause leakage. Also to prevent leaks, replace the stoppers in the lid if they become worn or loose.



CALCULATIONS:

Soil Respiration (lb CO_2-C/acre/day) = PF x TF x (%CO_2 - 0.035) x 22.91 x H

PF = pressure factor = 1
TF = temperature factor = (soil temperature in Celsius + 273) ÷ 273
H = inside height of ring = 5.08 cm (2 inches)

3. Infiltration Test

The infiltration test is generally performed after the **first** respiration measurement. The same 6-inch diameter ring left in place from the soil respiration test can be used for the infiltration test. If soil respiration was not determined, follow the instructions in Step 1 of the soil respiration procedure (Chapter 2) for inserting the 6-inch diameter ring.

Materials needed to measure infiltration:

- **6-inch diameter ring (left in soil from respiration test)**
- **plastic wrap**
- **500 mL plastic bottle or graduated cylinder**
- **distilled water**
- **stopwatch or timer**

Did You Know?

Infiltration rate is a measure of how fast water enters the soil. Water entering too slowly may lead to ponding on level fields or to erosion from surface runoff on sloping fields.

Considerations: If the soil is saturated, infiltration will not occur. Wait for one or two days to allow for some drying. Also, if the respiration test is not performed, make sure the sampling area is free of residue and weeds or that vegetation is trimmed to the soil surface before inserting the ring.

1 **Firm Soil**

With the 6-inch diameter ring in place, use your finger to gently firm the soil surface **only** around the **inside edges** of the ring to prevent extra seepage. Minimize disturbance to the rest of the soil surface inside the ring.

2 **Line Ring with Plastic Wrap**

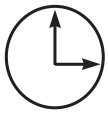
Line the soil surface inside the ring with a sheet of plastic wrap to completely cover the soil and ring as shown in **Figure 3.1.** This procedure prevents disturbance to the soil surface when adding water.

Figure 3.1

3 **Add Water**

- Fill the plastic bottle or graduated cylinder to the 444 mL mark with distilled water.
- Pour the 444 mL of water (1" of water) into the ring lined with plastic wrap as shown in **Figure 3.1.**

4 Remove Wrap and Record Time



- Remove the plastic wrap by gently pulling it out, leaving the water in the ring **(Figure 3.2)**. N**ote the time.**

- Record the amount of time (in minutes) it takes for the 1" of water to infiltrate the soil. Stop timing when the surface is just glistening.

Figure 3.2

- If the soil surface is uneven inside the ring, count the time until half of the surface is exposed and just glistening **(Figure 3.3)**.

- Enter the amount of time in minutes on the Soil Data worksheet.

Figure 3.3

5 Repeat Infiltration Test

In the same ring, perform Steps 2, 3, & 4 with a second inch of water. On the Soil Data worksheet, enter the number of minutes elapsed for the second infiltration measurement. If soil moisture is at or near field capacity, the second test is not necessary.

[The moisture content of the soil will affect the rate of infiltration; therefore, two infiltration tests are usually performed (if soil is dry). The first inch of water wets the soil, and the second inch gives a better estimate of the infiltration rate of the soil.]

6 Replace Lid

If a second respiration measurement will be performed, set the lid loosely on the ring and leave it covered for preferably 16 to 24 hours (6-hour minimum) before beginning the second test (Chapter 2). (Remove lid and replace it before beginning the second soil respiration measurement).

> **!** **Reminder**: If you still need to perform the second respiration measurement, remember to loosely place the lid back on the ring before leaving the field.

4. Bulk Density Test

The bulk density measurement should be performed at the soil surface and/or in a compacted zone (plow pan, etc.) if one is present . Measure bulk density near (between 1 and 2 feet) the site of the respiration and infiltration tests. To get a more representative bulk density measurement of the area, additional samples may be taken.

Materials needed to measure bulk density:

- **3-inch diameter ring**
- **hand sledge**
- **wood block**
- **garden trowel**
- **flat-bladed knife**
- **sealable bags and marker pen**
- **scale (0.1 g precision)**
- **1/8 cup (30 mL) measuring scoop**
- **paper cups**
- **18-inch metal rod**
- **access to a microwave oven**

Did You Know?
Bulk density is the weight of soil for a given volume. It is used to measure compaction. In general, the greater the density, the less pore space for water movement, root growth and penetration, and seedling germination.

Considerations: For rocky or gravelly soils, use the alternate procedure on page 11.

1 **Drive Ring into Soil**

- Using the hand sledge and block of wood, drive the 3-inch diameter ring, beveled edge down, to a depth of 3 inches **(Figure 4.1)**.

- The exact depth of the ring must be determined for accurate measurement of soil volume. To do this, the height of the ring above the soil should be measured. Take four measurements (evenly spaced) of the height from the soil surface to the top of the ring and calculate the average. Record the average on the Soil Data worksheet.

Figure 4.1

NOTE: Use the metal rod to probe the soil for depth to a compacted zone. If one is found, dig down to the top of this zone and make a level surface. Proceed with Step 1.

2 **Remove 3-inch Ring**

Dig around the ring and **with the trowel underneath it,** carefully lift it out to prevent any loss of soil.

3 Remove Excess Soil

Remove excess soil from the sample with a flat-bladed knife. The bottom of the sample should be flat and even with the edges of the ring **(see Figure 4.2).**

Figure 4.2

4 Place Sample in Bag and Label

Touch the sample as little as possible. Using the flat-bladed knife, push out the sample into a plastic sealable bag. Make sure the entire sample is placed in the plastic bag. Seal and label the bag.

NOTE: Steps 5-7 can be done in a lab or office if a scale is not available in the field. Step 8 requires access to a microwave.

5 Weigh and Record Sample

- Weigh the soil sample in its bag. [If the sample is too heavy for the scale, transfer about half of the sample to another plastic bag. The weights of the two sample bags will need to be added together. Enter the weight (sum of two bags, if applicable) on the Soil Data worksheet.

- Weigh an empty plastic bag to account for the weight of the bag. Enter the weight (sum of two bags, if applicable) on the Soil Data worksheet.

6 Extract Subsample to Determine Water Content and Dry Soil Weight

- Mix sample thoroughly in the bag by kneading it with your fingers.

- Take a 1/8-cup level scoop subsample of loose soil (not packed down) from the plastic bag and place it in a paper cup (a glass or ceramic cup may be used).

7 Weigh and Record Subsample

- Weigh the soil subsample in its paper cup. Enter the weight on the Soil Data worksheet.

- Weigh an empty paper cup to account for its weight. Enter the weight on the Soil Data worksheet.

8 Dry Subsample

Place the paper cup containing the subsample in a microwave and dry for two or more four-minute cycles at full power. Open the microwave door for one minute between cycles to allow venting. Weigh the dry subsample in its paper cup and enter the weight on the Soil Data worksheet.

NOTE: To determine if the soil is dry, weigh the sample and record its weight after each 4-minute cycle. When its weight does not change after a drying cycle, then it is dry.

CALCULATIONS (See page 13)

Bulk Density Test for Gravelly and Rocky Soils

This method is to be used when rocks or gravels prevent sampling bulk density by the core method described in the first part of this Chapter. This excavation method will require the user to sieve out the coarse material greater than 2 mm in size.

Materials needed to measure bulk density:

- **Plastic wrap**
- **140-cc syringe**
- **water**
- **garden trowel**
- **sealable bags and marker pen**
- **2-mm sieve**
- **scale (0.1 g precision)**
- **1/8-cup (30 mL) measuring scoop**
- **paper cup or bowl**
- **access to a microwave oven**

Considerations: Choose a spot that is as level as possible to allow water to fill the hole evenly. If the soil is too wet to sieve, ignore the part in Step 2 about replacing rocks, and proceed to Step 3. Soil will have to be dried and sieved later. The volume of gravel will need to be determined and subtracted from the total volume of the soil sample taken in the field.

1 **Dig Hole**

- Dig a bowl shaped hole three inches deep and approximately five inches in diameter using the trowel (**Figure 4.3)**. Avoid compacting the soil in the hole while digging. Place **all** of the soil and gravel removed from the hole in a plastic bag.

- Using the 2-mm sieve, sieve the soil in the plastic bag to separate the gravel. Collect the soil in a plastic sealable bag. Put the gravel aside to be used in Step 2. Seal and label the plastic bag. **[Note: See Considerations above if soil is wet.]**

Figure 4.3

2 Line the Hole

Line the hole with plastic wrap as shown in **Figure 4.4**. Leave some excess plastic wrap around the edge of the hole. Place the sieved rocks and gravel carefully in the center of the hole on top of the plastic wrap. Assure that the pile of rocks **do not** protrude above the level of the soil surface.

Figure 4.4

3 Add Water to Hole

- Use the 140 cc syringe to keep track of how much water is needed to fill the lined hole. The level of the water should be even with the soil surface.

- The amount of water represents the volume of soil removed. Record the total amount of water in cubic centimeters (1 cc = 1 cm^3) on the Soil Data worksheet.

NOTE: Steps 4-6 can be done in a lab or office if a scale is not available in the field. Step 7 requires access to a microwave.

4 Weigh and Record Sample

- Weigh the soil sample in its bag. [If the sample is too heavy for the scale, transfer about half of the sample to another plastic bag. The weights of the two sample bags will need to be added together. Enter the weight (sum of two bags, if applicable) on the Soil Data worksheet.

- Weigh an empty plastic bag to account for the weight of the bag. Enter the weight (sum of two bags, if applicable) on the Soil Data worksheet.

5 Extract Subsample to Determine Water Content and Dry Soil Weight

- Mix sample thoroughly in the bag by kneading it with your fingers.

- Take a 1/8-cup level scoop subsample of loose soil (not packed down) from the plastic bag and place it in a paper cup (a glass or ceramic cup may be used).

6 Weigh and Record Subsample

- Weigh the soil subsample in its paper cup. Enter the weight on the Soil Data worksheet.

- Weigh an empty paper cup to account for its weight. Enter the weight on the Soil Data worksheet.

7 Dry Subsample

Place the paper cup containing the subsample in a microwave and dry for two or more four-minute cycles at full power. Open the microwave door for one minute between cycles to allow venting. Weigh the dry subsample in its paper cup and enter the weight on the Soil Data worksheet.

NOTE: To determine if the soil is dry, weigh the sample and record its weight after each 4-minute cycle. When its weight does not change after a drying cycle, then it is dry.

CALCULATIONS (for both bulk density methods):

Soil water content (g/g) = $\frac{\text{(weight of moist soil - weight of oven dry soil)}}{\text{weight of oven dry soil}}$

Soil bulk density (g/cm^3) = $\frac{\text{oven dry weight of soil}}{\text{volume of soil}}$

Soil water-filled pore space (%) = $\frac{\text{volumetric water content x 100}}{\text{soil porosity}}$

Volumetric water content (g/cm^3) = soil water content (g/g) x bulk density (g/cm^3)

Soil porosity (%) = $1 - \left(\frac{\text{soil bulk density}}{2.65}\right)$

Volume of Rocks (cm^3) = Fill 1/3 of a graduated cylinder with water, and record the amount. Add the rocks to the cylinder and record the change in the water level. The difference is the volume of rocks (1 mL = 1 cm^3).

Volume of Soil (cm^3) = Total soil volume - volume of rocks

5. Electrical Conductivity Test

Soil samples for the electrical conductivity (EC) test are taken from the 0- to 3-inch depth. Bulked soil samples from across the field can be collected, and two subsamples can be taken for analysis (See Chapter 1, Sampling Guidelines). **Electrical conductivity, pH, and soil nitrate are all measured from the same soil subsample.**

Materials needed to measure electrical conductivity (EC):

- **1/8-cup (30 mL) measuring scoop**
- **120-mL plastic containers with lid**
- **EC pocket meter (blue with black cap)**
- **squirt bottle**
- **calibration solution (0.01 M KCl)**
- **distilled water**

Did You Know?
Excess salts in soil can be a detriment to plant health. Salts can also hamper water movement into the soil and increase the occurrence of surface compaction.

1 Extract Subsample

The soil sample should be thoroughly mixed before taking a subsample. Measure a 1/8-cup level scoop subsample of soil and place it in the plastic container. If soil nitrates will be measured on this subsample (Chapter 7), weigh the subsample for a more accurate estimate of soil nitrates. Enter the subsample weight on the Soil Data worksheet.

2 Add Water to Subsample and Mix

- Add 1/8-cup (30 mL) of distilled water to the container with the subsample. The resulting soil/water mixture equates to a 1:1 soil to water ratio on a volume basis.
- Put the lid on the container and shake vigorously about 25 times.

Calibration Tip: Make sure the EC meter is calibrated before making a measurement. See Appendix C for calibration instructions.

3 Measure and Record EC (See Calibration Tip)

- Open the container and insert the EC pocket meter into the soil-water mixture. Take the reading while the soil particles are still suspended in solution. To keep the soil particles from settling, stir gently with the EC pocket meter. Do not immerse the meter above the immersion level (See Appendix C, Figure 1c). Allow the reading to stabilize (stays the same for about 10 seconds).
- Enter the EC reading on the Soil Data worksheet in decisiemens per meter (dS/m). The DiST WP 4 meter gives readings directly in dS/m. For the Microsensor 4 meter, divide the reading by 10, and for the Microsensor 3 meter, divide the reading by 100 to get readings in dS/m.
- Save the soil-water mixture for the pH measurement (Chapter 6).

4 Turn the meter off. Thoroughly rinse meter with distilled water and replace cap.

6. Soil pH Test

Use the same soil-water mixture prepared in the EC test to conduct the pH Test. **If you are starting with a fresh soil sample, read the introduction and follow Steps 1-3 in the EC Test Chapter on preparing the sample.**

Materials needed to measure pH:

- **1/8-cup (30 mL) measuring scoop**
- **plastic specimen bottle**
- **calibration buffer solutions**
- **squirt bottle**
- **pH pocket meter (red with black cap)**
- **distilled water**

Did You Know?
Soil acidification can also be an indication of excessive N fertilizer applications and N leaching loss.

Considerations: If the soil sample is saturated or very wet, a 1:1 ratio, on a volume basis, of soil to water will not be obtained in the soil-water mixture (See Step 2, Chapter 5). Let the soil dry before proceeding with Step 1 in Chapter 5. Also, a small amount of salts diffuse out of the pocket pH meter; therefore, **EC measurements should always be taken first when measuring both EC and pH on the same sample**.

① Measure and Record pH

- Make sure to periodically calibrate your pH meter (See Appendix C for instructions). If the meter has not been used in a while, place the meter in tap water for about 5 minutes before calibrating or taking a reading.

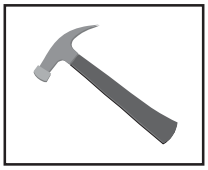
- Wait about 10 to 15 minutes after the EC measurement before measuring the pH. This gives the soil particles time to settle. Insert the pH pocket meter into the topmost portion of the solution and turn the meter on. Wait until the reading stabilizes (0-30 seconds), and record the digital reading on the Soil Data worksheet.

② Rinse Pocket Meter

- Thoroughly rinse the electrode with distilled water.

- Store the electrode with a few drops of the **pH 7** buffer solution and replace the cap. (See Appendix C on storage of pH meter)

Maintenance Tips: Check the batteries and calibrate the EC and pH meters periodically. Be sure to clean the meters thoroughly to keep them working properly.



7. Soil Nitrate Test (NO_3^-)

Use the same sample prepared for the EC and pH tests to measure soil nitrates. **If you are starting with a fresh soil sample, read the introduction and follow Steps 1-3 in the EC Test Chapter on preparing the sample.**

Materials needed to measure soil nitrate:

- **filter paper**
- **120-mL plastic container with lid**
- **eye dropper**
- **nitrate/nitrite test strips**
- **stopwatch or timer**
- **distilled water**

Did You Know?
Soil nitrates are good measures of plant-available nitrogen, but they can be readily lost from the soil by leaching and volatilization.

1 Fold Filter

Fold the filter paper in half (into a semicircle). Fold it again, but not quite into a quarter-circle. Leave the edges a little uneven as in **Figure 7.1** (A black line is drawn for demonstration purposes.)

Figure 7.1

2 Insert Filter Paper into Subsample

Open the filter paper into the shape of a cone and push it (pointed part first) quickly into the jar with the soil/water mixture until it touches the bottom of the jar **(Figure 7.2)**. **Wait** until about an eye dropper-full of the solution has seeped through to the inside of the filter paper. **(Note: Inserting the filter paper quickly prevents it from wetting up and tearing as it is inserted.)**

[For Steps 3 & 4, it would be helpful to first familiarize yourself with the directions on the side of the bottle of nitrate strips.]

Figure 7.2

3 Place Drops on Nitrate Strips

Using the eye dropper and one nitrate/nitrite test strip, place 1 or 2 drops of the filtered solution on each of the strip's two pads. **Note the time.**

NOTE: One pad measures the amount of nitrite, and the other measures the amount of nitrite and nitrate combined. Nitrite rarely occurs in measurable amounts in soils, so nitrite readings from the test strips are not recorded.

4 Measure and Record Nitrate

- Align the nitrate/nitrite test strip with the bottom of the bottle with your thumb corresponding to the diagram on the bottle.

- **After 60 seconds,** compare the first pad (furthest from your thumb) along the nitrate scale as shown in **Figure 7.3**. Estimate the nitrate amount according to the degree of color change. Enter the value from the nitrate scale on the Soil Data worksheet in ppm. This value is an estimate of nitrate-N concentration in the extract.

Figure 7.3

NOTE: The nitrate test strips have a shelf-life. Check the expiration date on the bottle.

CALCULATIONS:

Estimated (lb NO_3-N/acre) =

$$\frac{\text{(ppm extract } NO_3\text{-N) x (depth of soil sampled in cm) x bulk density x 0.89}}{10}$$

Exact (lb NO_3-N/acre) =

$$\frac{\text{(ppm } NO_3\text{-N) x (volume water used) x (depth of soil sampled, cm) x bulk density x 0.89}}{\text{(dry weight of soil) x 10}}$$

Volume water used = 30.0 mL + [dry weight of soil x soil water content (g/g)]

Note: The maximum nitrate-N reading on the nitrate/nitrite test strip container is 50 ppm. If the sample reading falls into the 50 ppm category, the sample can be diluted to get a better estimate of the actual amount over 50 ppm. To dilute the sample, fill the eye dropper with filtered solution and place five drops in a plastic container. Add five drops of distilled water; mix gently by swirling the container. Take a reading with a new test strip as stated in Step 4. Multiply the estimated nitrate-N in ppm by 2 before using the calculations. If the nitrate reading falls into the category of 50 ppm again, repeat the dilution steps, and multiply the estimated nitrate-N in ppm by 4.

Did You Know?

Water samples may be taken from drinking water, well water, tile drainage, drainage ditches, and ponds. Dip a nitrate/nitrite test strip into the water and estimate the nitrate or nitrite concentration from the color chart on the test strip bottle. This test can give you an idea of how much N fertilizer is lost from the soil. **(See Chapter 12).**

8. Aggregate Stability

Aggregate stability measures the amount of stable aggregates against flowing water. It is recommended that aggregate stability be determined on the top three inches of surface soil. The soil sample should be air-dried before determining aggregate stability.

Materials needed to measure aggregate stability:

- **2-mm sieve (3-inch diameter)**
- **0.25-mm sieves (2.5-inch diameter)**
- **terry cloths**
- **400-watt hair dryer and drying chamber**
- **calgon solution (about 2 tbsp of calgon per 1/2 gallon of tap water)**
- **bucket or pan**
- **scale (0.1 g precision)**
- **distilled water**

Did You Know?
Soil aggregates protect organic matter within their structure from microbial attack. Formation and preservation of aggregates allows organic matter to be preserved in the soil.

Considerations: If the soil is moist, air-dry a sample before determining aggregate stability. When taking a soil sample, care should taken not to disrupt the soil aggregates.

1 **Sieve the Soil Sample**

Transfer about a 1/4 cup of air-dried soil to the 2-mm sieve. Shake the sieve gently and collect the soil passing through the sieve. Try to pass all of the soil through the sieve by gently pressing the soil through with your thumb **(Figure 8.1)**.

Figure 8.1

2 **Weigh Sieved Soil Sample**

Weigh the 0.25-mm sieve, and record its weight on the Soil Data worksheet. Weigh out about 10 g of the sieved soil from Step 1 (make sure the soil is mixed well before taking a subsample). Record the exact weight on the Soil Data worksheet.

3 **Slowly Wet the Soil Sample in Sieve**

Saturate one of the terry cloth sheets with distilled water and lay it flat. Place the 0.25-mm sieve containing the soil on the wet cloth, allowing the soil to wet up slowly **(Figure 8.2)**. Wet the soil for five minutes.

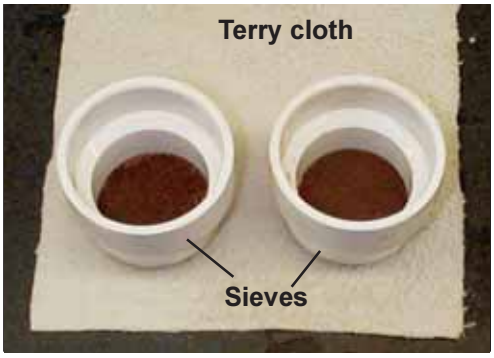


Figure 8.2

NOTE: A container (bucket or pan) of distilled water is needed for Step 4. The water temperature should be at or near the temperature of the soil.

4 Wet Sieve the Soil

- Place the 0.25-mm sieve with soil in the container filled with distilled water, so that the water surface is just above the soil sample.
- Move the sieve up and down in the water through a vertical distance of 1.5 cm at the rate of 30 oscillations per minute (one oscillation is an up and down stroke of 1.5 cm in length) for three minutes. **Important: Make sure the aggregates remain immersed in water on the upstroke.**

5 Dry Aggregates

After wet sieving, set the sieve with aggregates on a dry piece of terry cloth, which will absorb the excess water from the aggregates in the sieve. Then place the sieve containing the aggregates on the drying apparatus **(Figure 8.3)**. Allow the samples to dry using the low power setting.

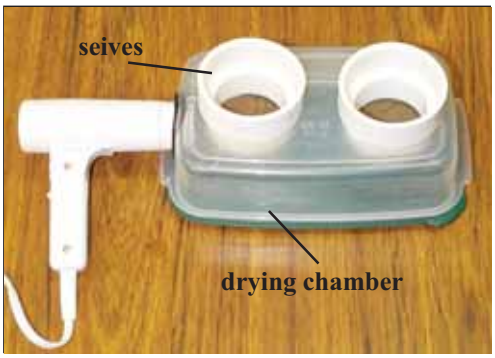


Figure 8.3

NOTE: Be careful when drying the soil to prevent particles from blowing out of the sieves. It may be necessary to put a cover over the top of the sieves to keep aggregates in place.

6 Weigh Aggregates

After drying, allow the aggregates and sieve to cool for five minutes. Weigh the sieve containing the aggregates. Record the weight of the sieve plus aggregates on the Soil Data worksheet.

7 Disperse Aggregates in Calgon Solution

- Prepare calgon solution. Immerse the sieve containing the dried aggregates in the calgon solution **(do not completely immerse the sieve)**. Allow the aggregates in the sieve to soak for five minutes, moving the sieve up and down periodically. Only sand particles should remain on the sieve.
- Rinse the sand on the sieve in clean water by immersing the sieve in a bucket of water or by running water through the sieve.

8 Dry and Weigh Sand

- Remove excess water by first placing the sieve containing the sand on the dry terry cloth, then placing it on the drying apparatus. Allow sand to dry.
- After drying, allow the sand and sieve to cool for five minutes. Weigh the sieve containing the sand. Record the weight of the sieve plus sand on the Soil Data worksheet.

CALCULATIONS:

Water Stable Aggregates (% of soil > 0.25mm) = $\frac{\text{(weight of dry aggregates - sand)}}{\text{(weight of dry soil - sand)}} \times 100$

9. Slake Test

The slake test measures the stability of soil when exposed to rapid wetting. This test is qualitative and should be measured on air-dried soil fragments or aggregates.

Materials needed to measure slaking:

- **complete soil stability kit**
- **sampling scoop**
- **distilled water (1 L)**

Did You Know?
Soil stability serves as a qualitative indicator of soil biological activity, energy flow, and nutrient cycling. Binding of soil particles must constantly be renewed by biological processes.

Considerations: The soil should be **air-dry** when performing this test. If the soil is not dry, collect surface fragments as described in Step 1 and allow them to dry. Be careful not to destroy the soil fragments while sampling.

1 Collect Surface Fragments

- Carefully remove soil fragments or aggregates from the soil surface. If there is a surface crust, carefully sample pieces of it. Use the **flat end** (handle) of the scoop to lift out surface and subsurface fragments. If the soil has been tilled, collect some aggregates (about 1 cm in size). Be careful not to shatter the soil fragments or aggregates while sampling.

- Collect 16 separate soil fragments. If there is a surface crust, collect eight fragments of the crust and eight fragments from below the crust.

Figure 9.1

2 Fill Box with Water

- Remove all sieve baskets from the stability kit.

- Fill the compartments in the box with water. The water should be 2 cm deep and at approximately the same temperature as the soil.

Figure 9.2

3 Test Soil Fragments

- Place soil fragments in the sieve baskets **(Figure 9.1).**

- Lower one of the sieves into a box compartment filled with water **(Figure 9.2)**.

4 Observe Fragments

- Observe the soil fragment for **five minutes**. Refer to the stability class table below to determine classes 1 and 2.
- After five minutes, raise the basket out of the water, then lower it to the bottom. It should take one second for the basket to clear the surface and one second to return to the bottom.
- Repeat immersion four times (total of five immersions). Refer to the stability class table below to determine classes 3 through 6.

5 Record Ratings

- Soil stability is rated according to the time required for the fragment to disintegrate during the five-minute immersion and the proportion of the soil fragment remaining on the mesh after the five extraction-immersion cycles. **[See table below.]**
- Record the stability ratings for all 16 soil fragments or aggregates on the Soil Data worksheet.

Stability class	Criteria for assignment to stability class (for "Standard Characterization")
0	Soil too unstable to sample (falls through sieve).
1	**50 %** of structural integrity lost within **5 seconds** of insertion in water.
2	**50 %** of structural integrity lost **5 - 30 seconds** after insertion.
3	**50 %** of structural integrity lost **30 - 300 seconds** after insertion or **< 10 %** of soil remains on the sieve after 5 dipping cycles.
4	**10 - 25%** of soil remaining on sieve after 5 dipping cycles.
5	**25 - 75%** of soil remaining on sieve after 5 dipping cycles.
6	**75 - 100%** of soil remaining on sieve after 5 dipping cycles.

10. Earthworms

Earthworms are most active during the spring and fall, which are the best times to observe their activity.

Materials needed to measure the number of earthworms:

- **tap water (2 L)**
- **hand trowel or shovel**
- **large jar or container for worm collection and cleaning**
- **mustard solution (2 tablespoons mustard powder in 2 liters of tap water)**

Did You Know?
Earthworm burrowing improves infiltration and their casts improve aggregation. Earthworms also break down larger bits of residue for use by other soil organisms.

Considerations: When examining the soil for earthworms, avoid places where their populations might be affected, such as near mulch or compost piles. The abundance of earthworms is usually patchy within a field and varies with season. Therefore, count earthworms several times during a season and use the average to gauge changes from year to year.

1 **Dig Plot**

Measure a square-foot plot and dig down 12 inches with the hand trowel or shovel (**Figure 10.1**). Try to minimize the number of cuts with the shovel to avoid damage to the earthworms. **Dig the hole first, then sort for earthworms.**

Figure 10.1

2 **Count the Number of Earthworms**

Sort the soil samples against a pale-colored background to help locate the earthworms. Separate and count the number of earthworms.

3 **Add Mustard Solution (optional)**

To facilitate extraction of deep burrowing earthworms, add two liters of mustard solution to the hole. **First**, make sure the bottom of the hole is level. The deep burrowing worms should appear within five minutes (**Figure 10.2**). Count the number of worms.

Figure 10.2

4 **Record Total Number of Earthworms**

Record the total number of earthworms (those found in the hole and after adding the mustard solution) on the Soil Data worksheet. **[The mustard solution should not harm the worms. Rinse them in water before returning them to the soil.]**

11. Soil Physical Observations and Estimations

Materials needed in observing the soil physical properties:

- **tape measure**
- **sharpshooter spade or shovel**
- **18-inch metal rod**
- **tap water**

1 **Dig hole**

Dig a hole to a depth of 1 foot. Make it wide enough to cut out a slice of soil.

2 **Cut Slice of Soil**

Using the shovel, cut a slice of soil from a wall of the hole and lay it on the ground.

Figure 11.1

3 **Measure Depth of Topsoil**

- Measure the depth of the topsoil. Look for color changes from the soil surface downward through the soil profile. The topsoil is usually distinguished by a darker color than the underlying material **(See Figure 11.1)**.

- Record the depth of topsoil on the Soil Data worksheet.

4 **Observe Plant Roots**

- Observe plant roots in the hole and the slice of soil. To get a better look at the roots, dig down along a plant stem. The roots should be well branched with lots of fine root hairs.

- Things to look for are balled up roots or roots growing sideways. A lack of fine root hairs indicates oxygen deprivation in the root zone. Lateral root growth indicates a hardpan, or compacted layer.

5 **Determine Resistance**

- Use the metal rod to probe one of the side walls, starting from the soil surface to the bottom of the hole. Determine changes or differences in penetration resistance as you probe the side wall **(See Figure 11.2)**.

- Look for compacted layers that may restrict root growth and water movement.

Figure 11.2

6 Examine Soil Structure

Observe soil structure in the slice of soil to a depth of about 12 inches. Measure and mark, starting at the surface and moving downward; depth increments of 0 to 4 inches, 4 to 8 inches, and 8 to 12 inches. Note and record the type, size, and grade of the soil structural units or aggregates for each depth increment.

Note: Soil structure is how particles of soil are grouped together in stable collections or aggregates.

6a Note the type of soil structure at each of the three depth increments.

- The three general types of soil structure are granular (**Figure 11.3**), blocky (**Figure 11.4**), and platy (**Figure 11.5**).

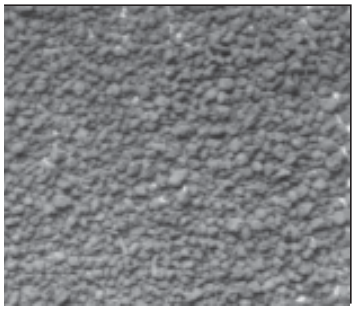


Figure 11.3 Granular: imperfect spheres, usually sand-size.

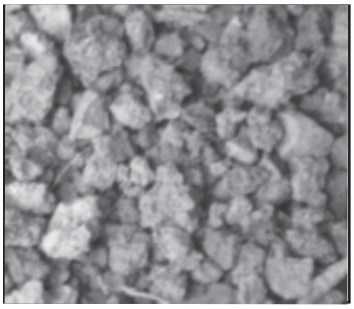


Figure 11.4 Blocky: imperfect cubes with angular or rounded edges.

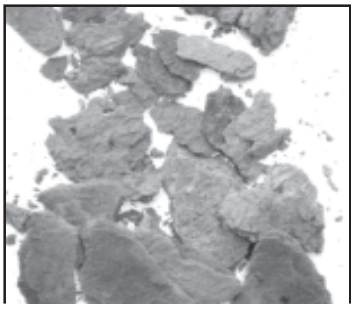


Figure 11.5 Platy: a flattened or compressed appearance.

- If there are no noticeable aggregates or peds, the soil has no structure. It is either single grained (**Figure 11.6**) or massive (**Figure 11.7**).

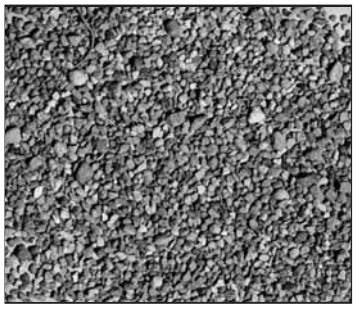


Figure 11.6 Single grain: unconsolidated mass such as loose sand.

Figure 11.7 Massive: cohesive mass.

- Record on the Soil Data worksheet the type of structure observed for each depth increment.

6b **Note the size of the aggregates or peds at the different depths**.

- Estimate the general size of the aggregates or peds. If the structure is granular, choose from fine (**Figure 11.8**), medium (**Figure 11.9**) and coarse (**Figure 11.10**) granule sizes.

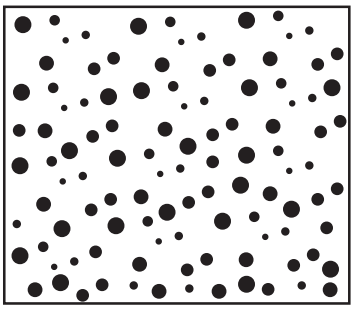


Figure 11.8
Fine: < 2 mm.

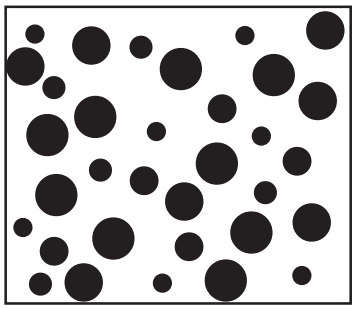


Figure 11.9
Medium: 2 to 5 mm.

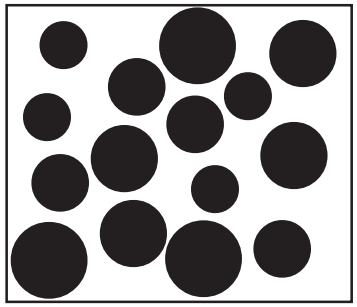


Figure 11.10
Coarse: 5 to 10 mm.

- If the structure is blocky, choose from very fine (**Figure 11.11**), fine (**Figure 11.12**), and medium (**Figure 11.13**) block sizes.

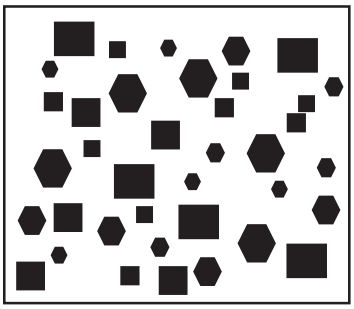


Figure 11.11
Very fine: < 5 mm.

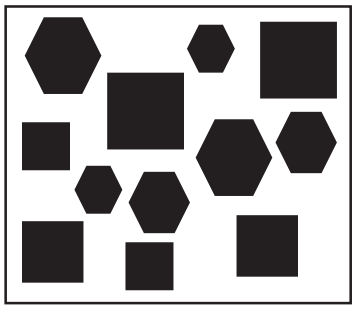


Figure 11.12
Fine: 5 to 10 mm.

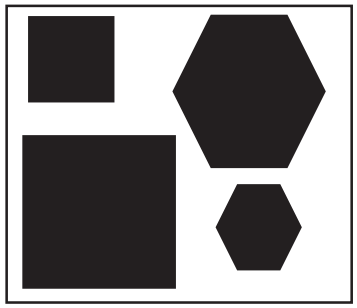


Figure 11.13
Medium: 10 to 20 mm.

- If structure is platy, choose from thin (**Figure 11.14**), medium (**Figure 11.15**), and thick (**Figure 11.16**) plate sizes.

Figure 11.14
Thin: < 2 mm.

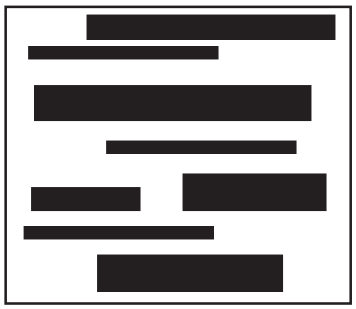


Figure 11.15
Medium: 2 to 5 mm.

Figure 11.16
Thick: 5 to 10 mm.

- Record on the Soil Data worksheet the size of the aggregates or peds observed for each depth increment.

6c **Note the distinctness (grade) of the aggregates in place and when removed from the slice of soil.**

The distinctness of the aggregates is either weak, moderate, or strong.

Weak structure:

- Aggregates or peds are barely observable in place in moist soil.
- When removed, the structure breaks into a few observable aggregates or peds **(Figure 11.17)**.

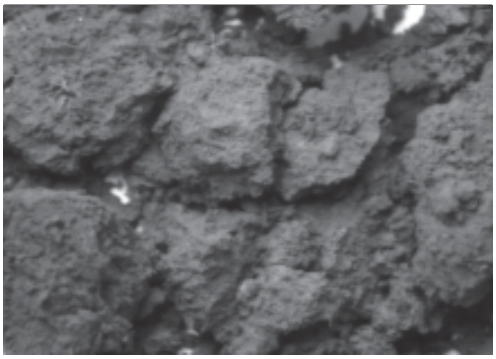


Figure 11.17

Moderate structure:

- Aggregates or peds are moderately well-formed and distinct in place.
- When removed, many well-formed aggregates are observable **(Figure 11.18)**.

Figure 11.18

Strong structure:

- Aggregates or peds are well-formed and very evident in place.
- When disturbed, the structure breaks into quite evident aggregates or peds **(Figure 11.19)**.

Record on the Soil Data worksheet the grade of the aggregates or peds observed for each depth increment.

Figure 11.19

7 **Determine soil textural class**

- Perform the Texture by Feel procedure **(See page 27)** on the top three inches of soil.
- Record on the Soil Data worksheet the soil textural class.

TEXTURE BY FEEL PROCEDURE

Making a Ribbon

Place approximately 25 grams in palm. Add water dropwise and knead the soil to break down all aggregates. Soil is at the proper consistency when plastic and moldable, like moist putty.

Add dry soil to soak up water

Yes

Yes

Does soil remain in a ball when squeezed?

No

Is the soil too dry?

No

Is the soil too wet?

No

Sand

Yes

Place ball of soil between thumb and forefinger, gently push the soil with the thumb, squeezing it upward into a ribbon. Form a ribbon of uniform thickness and width. Allow the ribbon to emerge and extend over the forefinger, breaking from its own weight.

Does the soil form a ribbon?

No

Loamy Sand

Yes

Does soil make a weak ribbon less than 1 inch long before breaking?

No

Does soil make a ribbon 1 inch long before breaking?

No

Does soil make a strong ribbon two inches or longer before breaking?

Yes

Yes

Yes

Excessively wet a small pinch of soil in palm and rub with forefinger.

Sandy Loam

Yes

Does soil feel very gritty?

No

Silt Loam

Yes

Does soil feel very smooth?

No

Loam

Yes

Neither gritty nor smooth?

Sandy Clay Loam

Yes

Does soil feel very gritty?

No

Silty Clay Loam

Yes

Does soil feel very smooth?

No

Clay Loam

Yes

Neither gritty nor smooth?

Sandy Clay

Yes

Does soil feel very gritty?

No

Silty Clay

Yes

Does soil feel very smooth?

No

Clay

Yes

Neither gritty nor smooth?

12. Water Quality Tests

A. Estimation of Water Nitrate and Nitrite levels

Materials needed to determine water nitrate (NO_3^-) and nitrite (NO_2^-) levels:

- **filter paper**
- **120-mL plastic containers with lids**
- **eye dropper**
- **nitrate/nitrite test strips**
- **stopwatch or timer**

Considerations: Water samples may be taken from drinking water, well water, tile drainage, drainage ditches, and ponds. Sample surface runoff from fields, which may be a contributing source of contaminates.

1 **Filter Water Sample (if cloudy)**

- Collect water sample in the plastic container. Fill to about 1/3 full.

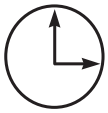
- Fold a piece of filter paper as described in Chapter 7--Soil Nitrate Test. Insert filter paper into the jar and allow the water to seep through the filter paper to the inside. **[If the water sample is clear (no cloudiness or suspended particles), the sample does not need to be filtered.]**

2 **Place Drops on Nitrate and Nitrite Strips**

Using the eye dropper, collect a sample of the filtered water. Place 1 or 2 drops of the filtered solution on each of the strip's two pads. Note the time.

[One pad measures the amount of nitrite and the other measures the amount of nitrite and nitrate combined.]

3 **Measure and Record Nitrate and Nitrite.**



- **After 30 seconds**, measure and record nitrite.
  Estimate the nitrite amount according to the degree of color change. Enter the value on the Soil Data worksheet in ppm from the nitrite scale on the bottle.

- **After 60 seconds**, measure and record nitrate.
  Estimate the nitrate amount according to the degree of color change. Enter the value on the Soil Data worksheet in ppm from the nitrate scale on the bottle.

[Note: Estimate results if colors on test pads fall between two color patches.]

B. Estimated Water Salinity Levels

Materials needed to estimate water salinity levels:

- **EC pocket meter**
- **120-mL plastic containers and lids**
- **distilled water**

Considerations: Water samples may be taken from drinking water, well water, tile drainage, ditches, irrigation water, and ponds.

1 Collect Sample

Collect water sample in plastic container. Fill to about 1/3 full.

2 Measure Electrical Conductivity

- Insert the EC pocket meter into the water sample. Allow the reading to stabilize (stays the same for about 10 seconds). Note the digital reading.

- Enter the EC reading on the Soil Data worksheet in decisiemens per meter (dS/m). The DiST WP 4 meter gives readings directly in dS/m. For the Microsensor 4 meter, divide the reading by 10, and for the Microsensor 3 meter, divide the reading by 100 to get readings in dS/m. Insert the EC pocket meter into the water sample until the reading stabilizes (stays the same for about 10 seconds). Note digital reading.

3 Rinse Pocket Meter

Turn off the meter. Thoroughly rinse the meter with distilled water, and replace cap.

Did You Know?

Healthy soil not only improves crop performance, it also cleans and stores water; and prevents runoff and erosion; and uses nutrients more efficiently, reducing the need for pesticides.

A. References

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B. Soil Respiration (Alternative Method)

This alternative method uses a kit produced by Woods End[1] known as the Solvita Soil Life Kit[1]. Instead of the Draeger tube apparatus, this procedure uses "paddles" inserted into a plastic container with the soil sample (See procedure on page 32). The use of this method eliminates the need for the Draeger tube (carbon dioxide adsorption tube), needle, and syringe. With the Solvita kits, results are given in 24 hours instead of 30 minutes with the Draeger method. The color change of the paddles may also be easier to distinguish than reading the color change off the Draeger tubes. The Solvita kit also requires the soil to be disturbed and will falsely stimulate microbial activity similar to the action of tillage. However, when used to compare sites, both soils are disturbed and the relative differences are noted. This procedure also reduces the effects of root respiration. Picking out as many roots from the sample as possible will further eliminate their CO_2 contribution. The Solvita kit may be preferred if immediate results are not necessary and the microbial activity differences without the influence of plant roots are desired.

The Solvita kit comes with well written and user friendly instructions and interpretations. There is also a trouble shooting guide to help the user. The kit consists of four parts: the sample jar to hold the correct volume of soil for the test **(Figure 1b)**; a foil-pack containing a special color gel paddle **(Figure 2b)**; instruction manual; and a color key for reading results **(Figure 2b)**.

Solvita Soil Life kits can be obtained from Woods End Research, Mt. Vernon, ME; solvita@woodsend.org.

Figure 1b

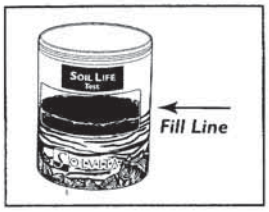
Figure 2b

[1]Trade names are used solely to provide specific information. Mention of a trade name does not constitute a guarantee of the product by the U.S. Department of Agriculture nor does it imply endorsement by the Department or the Natural Resources Conservation Service over comparable products that are not named.

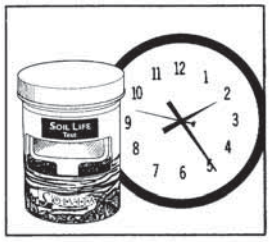
The following is part of the instructions from the SOLVITA SOIL LIFE KIT[1]:

RUNNING THE SOLVITA™ TEST

1. **SOIL SAMPLING:** Soil should be sampled from any garden or field in a fresh, moist condition just before the test is performed. Take many smaller samples from various locations and mix just well enough to be homogenous while removing large stones and organic debris.



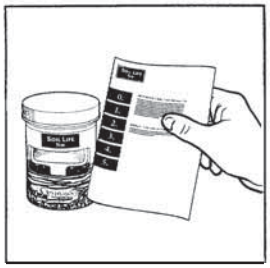
2. **IDEAL SOIL MOISTURE:** The soil should be at the ideal growing condition moisture before it is sampled. If the sample is very dry or very wet, it is best to wait until favorable conditions return. This may mean watering a dry soil and waiting 1-2 days again before sampling. If too wet, make a small pile to drain, or spread out to dry to a moderate moisture level. The idea is to disturb the natural state as little as possible.



3. **PUT SAMPLE INTO JAR:** Put the loose mix of soil into the jar just to the fill line. As you fill, tap the bottom of the jar sharply on a counter; this helps assure the correct density. Fill only to the indicated line. Record the time on the lid.

4. **START THE TEST:** When you are ready to start the test, open the foil-pack by tearing it along the top strip and carefully remove the paddle. *Do not touch the gel surface, and don't allow soil to touch it.* At the start of the test the paddle will be color #0 (bright blue). Once the foil pack is opened, the test should be started within about 30 minutes.

5. **INSERT THE PADDLE:** Push the paddle-stick point into the soil in the jar so that the gel-side can be seen through the back viewing side. Be careful not to jostle or tip the jar. Screw the lid on very tightly, and keep the jar at room temperature (68—77°F) *out of sunlight* for 24 hours.



6. **FIND THE GEL COLOR**: After 20 - 28 hours compare the color of the paddle to the Color Key provided. For this, the paddle should either be left in the jar with the lid on, or removed and laid face-up onto a white surface.

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C. EC and pH Meter Maintenance and Calibration

EC meter maintenance:

- Do not immerse the EC meter above the immersion level **(Figure 1c)**. Under no circumstances should the meter be immersed above the display level.

- When not in use, switch off the meter and replace the protective cap.

- To improve performance, clean the stainless steel electrodes periodically by rinsing them in alcohol for a few minutes.

- Replace all four batteries if the display becomes faint or disappears or if the readings are unstable or constant.

- To change batteries for DiST WP[1] models, unscrew the top with a coin and replace the batteries **(Figure 2c)**.

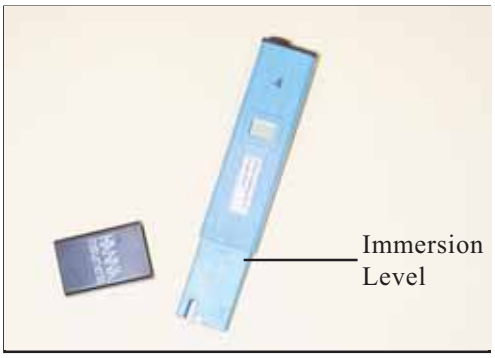

Figure 1c.

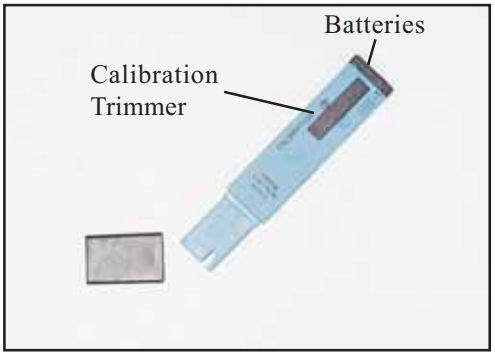

Figure 2c.

EC meter calibration:

- Immerse the meter into the calibration solution (1.41 dS/m).

- Allow the reading to stabilize. Using a small screw-driver, turn the Calibration Trimmer to match the solution value, 1.41 dS/m (normally at 25 C).

pH meter maintenance:

- Crystals may appear around the cap **(Figure 3c)**. This condition is normal. The crystals will dissolve when rinsed with water.

- After use, rinse the electrode with water to minimize contamination.

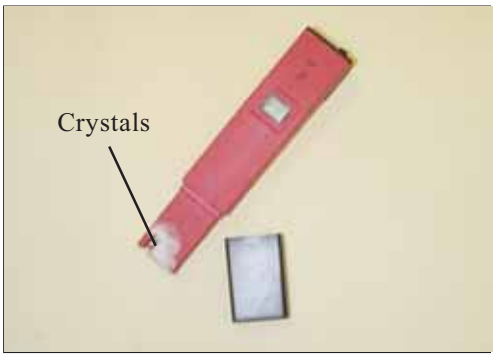

Figure 3c.

- Store the electrode with a few drops of storage solution (HI 70300L) or pH 7 solution in the protective cap. **DO NOT STORE IN DISTILLED OR DEIONIZED WATER.**

- Always replace the protective cap after use.

- Large differences in pH readings (± 0.5 pH) could be due to lack of calibration, dry electrode, or rundown batteries.

- If the pH meter cannot be switched on or if the display fades, unscrew the battery compartment and replace all four batteries, paying attention to their polarity **(Figure 4c)**.

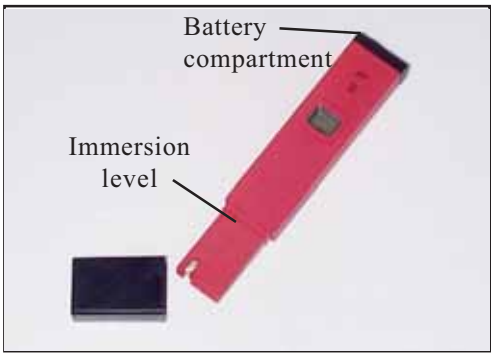

Figure 4c.

pH meter calibration (pHep 3[1]):

- Prepare buffer solutions. Only 2 buffers are needed, pH 7 and 4 or 10, depending on the pH range of your soils (see **Figure 5c**).

- Switch the unit on by pressing the ON/OFF button.

- With the meter on, press and hold the ON/OFF button for about three seconds. The display will start blinking "7.0o" to confirm that you have entered the calibration mode.

- Immerse the pH meter in the pH 7 buffer solution. Stir gently and wait approximately 20 seconds.

- If "Ec" appears on the display, the pH 7 solution is not fresh, or the electrode is not conditioned.

- The pHep 3[1] meter automatically confirms the pH 7 calibration after the meter is adjusted. The display will blink "4.0o". After a few seconds, it will display "Ec" to prompt you to use a second buffer solution.

- Rinse the electrode with water and immerse in pH 4 for acidic samples or pH 10 for alkaline samples. Allow approximately 20 seconds for the meter to auto-confirm the reading. Once the display stops blinking, the meter is calibrated and ready to use. ALWAYS USE FRESH BUFFERS FOR CALIBRATION.

Figure 5c.

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D. Building a Soil Quality Test Kit

Kit Inventory:

____ Two 6-inch diameter infiltration rings, 5 inches tall; cut from 1/8-inch thick aluminum irrigation pipe; beveled edge on one side **(Figure 1d)**. Mark a line around the outside of the ring, 2 inches from the top.
____ 12-inch wide roll of plastic wrap.
____ Two 500-mL plastic graduated cylinders. As a substitute, 500-mL plastic bottles or 16 oz. plastic soft drink containers can be used **(Figure 2d)**. Mark a line around the bottle at the 444 mL level.
____ 2-lb hand sledge or rubber mallet **(Figure 3d)**.
____ 7 3/4-inch long wood block, 2" by 4" **(Figure 3d)**.
____ Two 6-inch lids with septa (stoppers) for respiration chamber; 6-inch diameter stove-top caps can be used **(Figure 4d)**. As an alternative, the bottoms from a #10 can (coffee can) can be cut (1 inch lip) and used **(Figure 5d)**. The lids should be white or silver to reduce absorption of heat. Three holes fitted with red rubber stoppers, for serum or vaccine bottles, are drilled through the top of the lid to allow for gas sampling.
____ Two 6-inch long latex tubing (3/16" x 1/16").
____ Three 18- to 22-guage, 1.5-inch hypodermic needles.
____ 140-cc (mL) plastic syringe **(Figure 6d)**. A 2-mm hole can be drilled at the end of the syringe handle.
____ Pack of 10 Draeger tubes (0.1% CO_2 detection tubes) **[Figure 6d]**.
____ Soil thermometer (Celsius).
____ Two 3-inch diameter sampling tubes, 5 inches tall; cut from 1/8-inch thick aluminum irrigation pipe; beveled edge on one side **(Figure 1d)**. Mark a line around the outside of the ring, 2 inches from the top.
____ Flat-bladed knife.
____ Garden trowel (heavy duty) **[Figure 7d]**.
____ 1-qt sealable plastic storage bags.
____ 18-inch long metal rod, 1/8-inch in diameter. As a substitution, a straightened coat hanger can be used.
____ Calibrated scoop, 30.0 mL (1/8 cup) **[Figure 7d]**.
____ Squirt bottle.
____ Four 120-mL plastic containers with lids **(Figure 7d)**. As an alternative, any container with lid of similar size can be used; e.g., baby food jars.
____ Electrical conductivity meter (0.01-19.99 mS range) **[Figure 8d]**.

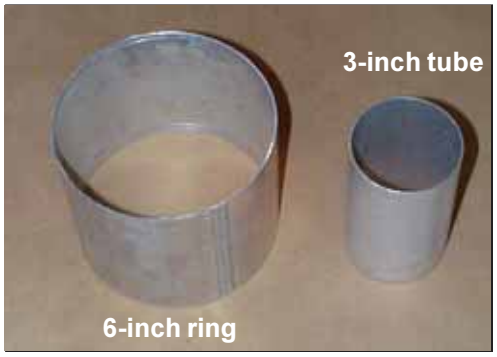

Figure 1d


Figure 2d


Figure 3d

Figure 4d

____ EC calibration standard, 1.41 dS/m (0.01 M KCl).
____ Small screw driver (for EC meter).
____ pH meter (model pHep3[1]) **[Figure 8d].**
____ Packets of 4, 7 and 10 pH buffers **(Figure 9d)**.
____ Bottle of 25 AquaChek[1] nitrate/nitrite test strips **(Figure 10d)**.
____ Box of filter paper, 12.5-cm diameter, Grade 2 - 5 can be used **(Figure 11d)**.
____Three standard plastic eyedroppers.
____Tape measure; 6-foot (metric and English units).
____ Small calculator
____ Permanent marker pen
____ 400-watt hair dryer **(Figure 12d)**.
____ 2-mm sieve, 3-inch diameter **(Figure 13d)**.
____ Two 0.25-mm sieves, 2-inch diameter **(Figure 14d)**.
____ Drying chamber, holds two 2-inch sieves **(Fig. 15d)**.
____ Two 6" x 6" sheets of terry cloth.
____ Calgon[1] (crystal form).
____ Soil stability kit (18 section tackle box with 18 1.5-mm sieve baskets) **[Figure 16d]**.
____ Stopwatch or timer.
____ Finger nail clipper.
____ Paper cups.

Other items needed:
____ Bucket or pan.
____ Mustard powder (optional).
____ Sharpshooter spade or shovel
____ Distilled water.

The kit requires the use of a scale with 0.1 g precision.

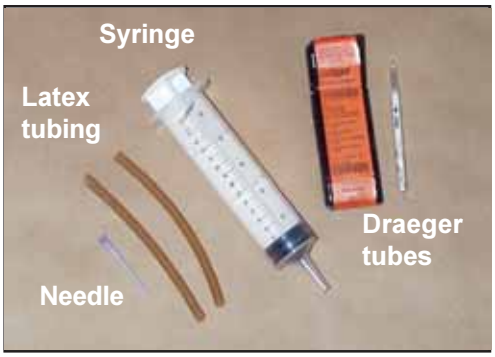

Figure 6d

Figure 7d

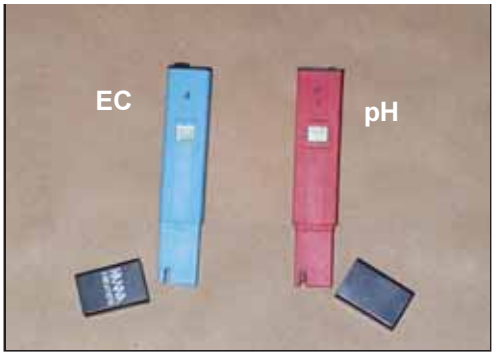

Figure 8d

Figure 5d

Figure 9d

[1]Trade names are used solely to provide specific information. Mention of a trade name does not constitute a guarantee of the product by the U.S. Department of Agriculture nor does it imply endorsement by the Department or the Natural Resources Conservation Service over comparable products that are not named.

The list on the previous pages describes all the equipment necessary for building a kit. The following are some listed items that can be easily constructed.

Construction of 2-mm sieves

1. The 2-mm-opening sieve can be made from No. 10 screens (2-mm openings) cut into discs approximately 75 mm in diameter.
2. The disc periphery is soldered with acid-core solder and mounted on the lip of 3-inch diameter PVC bushings.
3. A section of PVC (sleeve), dimensions 75 mm in diameter by 38 mm high (280 lb per inch rating), is made.
4. PVC cement is placed on the outside wall of the PVC sleeve, and on the inside wall of the PVC bushing. The PVC sleeve is pushed down firmly in to the PVC bushing, so they are cemented together (**Figure 13d)**.

Construction of 0.25-mm sieves

The 0.25-mm opening sieves can be made from No. 60 screens (hardware store) cut and mounted on the bottom of 2-inch diameter PVC joints (hardware or plumbing supply stores) using PVC cement, epoxy, or other thick glue **(Figure14d)**.

Construction of drying chamber

1. Any plastic container can be used. The container used here is a 4" x 6" plastic container with lid **(Figure 15d)**.
2. Drill two 2¼-inch holes in the bottom of the container (for insertion of 2-inch sieves).
3. Drill four ¼-inch holes in each side of the container, and a 1-inch hole on one side for insertion of the hair dryer. A 1-inch rubber grommet can be used to line the 1-inch hole to create a good seal when the hair dryer is inserted (**See Figure 15d**).

To accommodate the drying of more sieves (efficient for a large number of samples), a bigger container can be used with more 2¼-inch holes **(Figure 18d)**. The container pictured in Figure 18d is a small, trunk-style tackle box (13.5 x 8 x 6 inches). The inside tray was removed.

Figure 10d

Figure 11d

Figure 12d

Figure 13d

Construction of Soil Stability Kit

Construction of Box

1. Obtain a "parts" or "tackle" box with a lid; 18 cells, each cell at least 1¼" x 1¼" x 1½" deep **(Figure 16d)**. [sporting goods or hardware store].
2. Seal individual cells in the box with a small bead of silicone glue or caulk.

Figure 15d

Construction of Sieve Baskets (**Figure 17d**)

Materials:

- PVC, 1-inch in diameter, thin wall, about 2½ feet long.
- Aluminum window screen (1.5-mm openings), 4" x 8" piece (hardware store).
- Adhesive (epoxy or thick glue) [grocery or hardware store].

I*nstructions:*

1. Beginning at left end of the PVC pipe, make marks at 1½-inch intervals; 20 marks are needed.
2. Make a smaller mark ¼ to 3/8 inches to the *left* of all the first marks.
3. Beginning at left end, cut ¾ of the way through the tube at the first small mark using a hacksaw or bandsaw.
4. Beginning at the left end, use tin snips to make two lengthwise cuts through the tube, leaving a ¼" diameter "tab" (**See Figure 17d**).
5. Using tin snips or hacksaw, cut through the tube at the first large (1½ inch) interval mark.
6. Repeat steps 6-8 for each of the 20 sieves.
7. Cut 20 1¼" x 1¼" squares of aluminum window screen.
8. Glue a window screen square to the bottom of each sieve.
9. After allowing glue to dry, carefully trim screen to edge of sieves.

Figure 16d

Figure 17d

Figure 14d

Figure 18d

Outlet for kit items include:

Supplier[1]	Items Supplied[1]
Murray, Iowa FFA (515) 447-2517 http://www.geocities.com/murray_ffa/	Soil quality test kits (standard and deluxe kits)
Gempler's Inc. 100 Countryside Dr. P.O. Box 270 Belleville, WI 53508 (608) 424-1544 or (800) 382-8473 http://www.gemplers.com	A soil quality test kit as described in this manual. All kit items.
Fisher Scientific Pittsburgh, PA Ph. (800) 766-7000	Draeger tubes Filter paper, pH and EC meters, scales, graduated cylinders, 500-mL bottles, plastic containers, latex tubing, hypodermic needles
Scientific Industries 2207 Blue Bell Ave. Boulder, CO 80302 Ph. (303) 443-7087	Draeger tubes
Spectrum Technologies 23839 W. Andrew Rd. Plainfield, IL 60544 Ph. (800) 248-8873	AquaChek nitrate/nitrite test strips pH and EC meters scales
Markson Labsales Inc. P.O. Box 377 Wayne, New Jersey 07474 Ph. (800) 528-5114	EC standard solution (500-mL bottle, 1413 microsiemens) pH buffer capsules (pH 4, 7 and 10) Filter paper, pH and EC meters, scales graduated cylinders, 500-mL bottles, plastic containers
Walgreens	400-watt hair dryer
Forestry Suppliers, Inc. PO Box 8397 Jackson, MS 39284 Ph. (800) 647-5368	Sieves, scales
ATM Test Sieves West Allis, WI Ph. (800) 511-2096	0.25 mm screen (60 mesh) 2.0 mm screen (10 mesh)
Veterinary or medical supply	140-cc syringe
Hardware store Grocery or discount stores	2-lb hand sledges, tape measures, hand trowels, small screw drivers Plastic-wrap, 1-qt. sealable bags, 30-mL calibrated scoop

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Soil Quality Evaluation Site Description

Site Description		DATE:
Map Location	State:	County:
Geographic Location	Longitude:	Latitude:
Field or site location		
Landowner		
Soil Information		
Soil Series		
Slope %		
Erosion		
Mean Annual Temp.		
Mean Annual Precip.		
Present Management		
Cropping System (Rotations, cover crops, etc)		
Fertilizers/Pesticides (N inputs, pesticide use, etc)		
Tillage/Residue Cover (Type, depth, frequency, timing, % cover, etc)		
Irrigation (Pivot, gravity, amount and timing, etc)		
Other		
Past Management History		
Cropping System (Rotation/fallow history, etc)		
Fertilizers/Pesticides (N inputs, pesticide use, etc)		
Tillage/Residue Cover (Past tillage, frequency and type)		
Irrigation (past irrigation, how long?)		
Unusual Events (Floods, fires, land-leveling)		

Aerial view of field showing sampling sites and location of environmentally sensitive areas, such as ponds, creeks, wetlands, and other fragile sites adjacent to the field.

Scale 1 inch = _____ ft. (NA indicates sketch not to scale).

Additional Specifications and Notes:

Soil Respiration (at Initial Field Water Content)								DATE:	
	Sample site	(**H**) Ring height (cm)	Start time	End time	(**A**) Soil temp. (Celsius)	(**B**) Draeger tube %CO_2 (n=1)	Soil Respiration lbs CO_2-C/acre/day	(**B**) Draeger tube %CO_2 (n=5)	* Soil Respiration lbs CO_2-C/acre/day
1									
2									
3									
4									
Soil Respiration (at least 6 hours after irrigation or soil wetting)									
1									
2									
3									
4									

* Soil respiration = PF x ((**A** + 273)/273) x (**B** - 0.035) x 22.91 x **H** = lbs CO_2-C/acre/day

H = 5.08 cm (if not measured)

PF = Pressure Factor = 'raw' barometric pressure in inches Hg/29.9 inches.
Note: This adjustment is necessary at elevations > 3,000 ft.; otherwise PF = 1

Conversion: Degrees Celsius = 5/9 x (Degrees Fahrenheit - 32)

NOTES:

Infiltration (for 1 inch of water)								DATE:	
	Sample site	1st inch of water		**(W)** 1st Infiltration time (minutes)	* 1st Infiltration (in/hr)	2nd inch of water		**(W)** 2nd Infiltration time (minutes)	* 2nd Infiltration (in/hr)
		Start time	End time			Start time	End time		
1									
2									
3									
4									

* Conversion of infiltration time to inches per hour (in/hr); in/hr = (1/**W**) x 60

NOTES:

Bulk Density and Soil Water Status (core method)									DATE:	
	Sample site	(h) Height of ring above soil (cm)	(E) Weight of field moist soil + bag (grams)	(F) Weight of bag (grams)	Subsample for determinng soil water content				** (M) Soil H_2O content (g/g)	*** Soil bulk density (g/cm^3)
					(G) Weight of paper cup (grams)	(I) Weight of paper cup + soil (g)	(K) Dry weight of soil + cup	* (L) Dry weight of soil (grams)		
1										
2										
3										
4										
*Dry wt. of soil subsample = (K - G)				**Soil H_2O content = (I - K)/L						
***Soil bulk density = [(E - F)/(1 + M)]/[(12.7 - h) x 42.52] h = 5.08 cm (2 inches) if not measured; volume of soil = 324 cm^3										

Bulk Density and Soil Water Status for Gravelly Soils (excavation method)										
	Sample site	(n) Volume of water (cm^3)	(E) Weight of field moist soil + bag (grams)	(F) Weight of bag (grams)	Subsample for determinng soil water content				** (M) Soil H_2O content (g/g)	*** Soil bulk density (g/cm^3)
					(G) Weight of paper cup (grams)	(I) Weight of paper cup + soil (g)	(K) Dry weight of soil + cup	* (L) Dry weight of soil (grams)		
1										
2										
3										
4										
*Dry wt. of soil subsample = (K - G)				**Soil H_2O content = (I - K)/L						
***Soil bulk density = [(E - F)/(1 + M)]/(n) n = volume of soil in cm^3										

Soil Electrical Conductivity, pH, and Nitrate (NO_3^-)						DATE:	
	Sample site	(X) Weight of field moist soil (grams)	Readings for 1:1 soil:water mix.			* Estimated Soil NO_3-N (1b NO_3-N/acre)	** Exact Soil NO_3-N (1b NO_3-N/acre)
			EC (dS/m)	pH	**(Y)** Soil NO_3-N ppm (est.)		
1							
2							
3							
4							

*Estimated: lb NO_3-N/acre = **Y** x [depth of soil in cm /10] x soil bulk density x 0.89
Depth of soil = depth of soil sampled in centimeters; for kit it is 0 to 3 inches = 7.6 cm

Exact: lb NO_3-N/acre = **Y x C.F. x [depth of soil in cm /10] x soil bulk density x 0.89
C.F. = [30 mL + ((X/(1 + M)) x M)]/[X/(1 + M)] M = decimal soil water content (g/g)
Depth of soil = depth of soil sampled in centimeters; for kit it is 0 to 3 inches = 7.6 cm

Water Quality Measurements			DATE:	
	Sample site	Salinity (dS/m)	Water Nitrite (ppm)	Water Nitrate (ppm)
1				
2				
3				
4				

NOTES:

Aggregate Stability						DATE:
	Sample site	**(A)** Weight of sieve (grams)	**(B)** Weight of sieve + aggregates (grams)	**(C)** Weight of sieve + dry aggregates (grams)	**(D)** Weight of sieve + dry sand (grams)	* Percent water stable aggregates (% of soil > 0.25mm)
1						
2						
3						
4						
* % Water stable aggregates = (C - D)/(B - D) x 100						

Slake Test										DATE:
	Sample site	Individual Soil Slake Ratings								* Average Soil Slake Rating
1										
2										
3										
4										
* Soil Slake Rating = (add all of the individual ratings and divide by the total number)										

Earthworms				DATE:	NOTES:
	Sample site	Surface dwelling earthworms	Deep dwelling earthworms	Total Earthworms (no. per square foot)	
1					
2					
3					
4					

Soil Observations and Estimations	DATE:
	Description
Top soil depth (inches)	
Plant roots	
Compaction layer	
Soil texture	
Other	

Classes for Structure Index			
Structure			Class[a]
Type	Size	Grade	
Granular	Fine, Medium, Coarse	Weak	2
Granular	Fine, Medium, Coarse	Moderate	4
Granular	Fine, Medium, Coarse	Strong	5
Blocky	Very fine, Fine, Med.	Weak	1
Blocky	Very fine, Fine	Moderate	4
Blocky	Very fine, Fine, Med.	Strong	5
Blocky	Medium	Moderate	3
Plat	Thin, Medium, Thick	Very friable[b]	3
Plat	Thin, Medium, Thick	Friable[b]	2
Plat	Thin, Medium, Thick	Firm or Stronger[b]	1
Massive			1
Single Grain			1

Note: [a] Class 5 is the best. [b] Substitute horizontal moist rupture resistance.

Soil Structure						DATE:	
Depth (inches)	Type	Size	Grade	**(A)** Class	**(B)**	**(A)** x **(B)**	Structure index*
0 - 4					3		
4 - 8					2		
8 - 12					1		
*Structure index = ((Total - 6)/24) x 100				Total =			

NOTES:

Soil Quality Test Kit

SECTION II

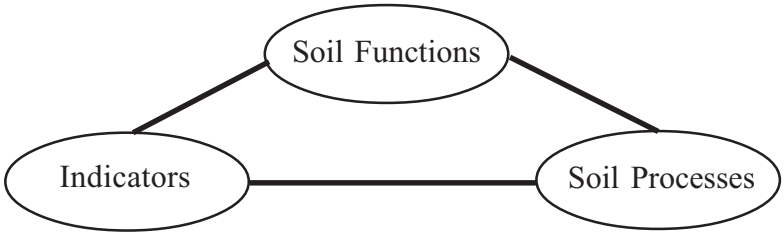
Background & Interpretive Guide for Individual Tests

Section II

INTRODUCTION

Soil quality assessment or interpretation should be considered a process through which soil resources are evaluated on the basis of soil function (what the soil does) and change in soil function in response to a specific natural or introduced stress, or management practice. Five vital soil functions have been proposed. They are: (1) sustaining biological activity, diversity, and productivity; (2) regulating and partitioning of water and solute flow; (3) filtering, buffering, degrading, immobilizing, and detoxifying organic and inorganic materials, including industrial and municipal by-products and atmospheric deposition; (4) storing and cycling of nutrients and other elements within the Earth's biosphere; and (5) providing support of socioeconomic structures and protection for archeological treasures associated with human habitation (Karlen et al., 1997).

It is also important to emphasize that soil quality evaluations must consider biological, chemical, and physical properties and processes. For interpretation, the measurements must be evaluated with respect to their long-term trends or signs of sustainability. A general sequence of how to evaluate soil quality is to (1) define the soil functions of concern, (2) identify specific soil processes associated with those functions, and (3) identify soil properties and indicators that are sensitive enough to detect changes in the functions or soil processes of concern (Carter et al., 1997).



Section II provides background and interpretive information for each test described in Section I. Each test is considered to be an indication of the level of functioning. However, indicator data is not meaningful unless a baseline or some reference condition is available for comparison or unless relative comparisons between management systems are made.

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1. Soil Respiration

Introduction

Soil respiration is the production of carbon dioxide (CO_2) as a result of biological activity in the soil by microorganisms, live roots, and macroorganisms such as earthworms, nematodes, and insects (Parkin et al., 1996). Carbon dioxide emitted from soil is a colorless and odorless gas that enters the atmosphere and annually exceeds the amount emitted by all human activities (Volk, 1994). The activity of organisms in the soil is considered to be a positive attribute for soil quality.

Soil respiration is highly variable both spatially and seasonally, and is strongly affected by moisture and temperature conditions. Because this variability can complicate interpretations, certain sampling precautions must be taken.

Knowing the history of the sampling site and characteristics of nearby soils becomes very important when evaluating respiration. Soil color may provide some assistance when interpreting respiration rates. A light colored soil with a high respiration rate may be indicative of a soil being depleted of organic matter. A relatively darker soil with the same rate could be considered healthy. The dark color indicates the presence of organic matter. Tillage or cultivation can result in loss of soil carbon (C) and increases in the amount of CO_2 released. The soil is loosened, which creates better accessibility of oxygen necessary for organic matter decomposition and respiration, resulting in CO_2 release (Reicosky and Lindstrom, 1995).

Interpretations

When comparing soil respiration rates from different sites or from the same site at different times, differences in soil temperature and soil water content must be taken into account. Soil temperature corrections can be performed using the general rule that biological activity increases by a factor of 2 with each 10°C increase in temperature (Parkin et al., 1996). The following equation can be used to standardize (to 25°C) for differences in soil temperatures that are between 15 and 35°C:

$$\text{Standardized soil respiration rate} = \text{soil respiration rate} \times 2^{[(25-T)\div 10]}$$

For soil temperatures between 0 and 15°C, the following equation is used:

$$\text{Standardized soil respiration rate} = \text{soil respiration} \times 4^{[(25-T)\div 10]}$$

For example, if you had a soil respiration rate of 15 CO_2-C lbs/a/d and soil temperature of 22°C, the first equation listed above would be used, and the standardized soil respiration rate would be calculated as follows:

1. $[(25 - 22) \div 10] = 0.3$
2. $2^{0.3} = 1.2$
3. (15 CO_2-C lbs/a/d) x 1.2 = 18 CO_2-C lb/a/d (standardized respiration rate at 25°C)

Standardization for differences in soil water content must also be taken into account. Maximum

microbial activity generally occurs when 60% of the soil pores are filled with water (Parkin et al., 1996). The amount of water in the pore space is referred to as **water-filled pore space** (WFPS), and gives an indication of how well aerated the soil is at the time of sampling.

Water-filled pore space (%) = (volumetric water content x 100) ÷ [1 - (soil bulk density ÷ 2.65)]

Soil respiration can be adjusted to equivalent values at 60% WFPS through the following equation for WFPS values between 30 and 60% (Parkin et al., 1996):

$$\text{Soil respiration}_{60} = \text{soil respiration rate} \times (60 \div \text{measured \%WFPS})$$

For WFPS values between 60 and 80%, the following equation is used:

$$\text{Soil respiration}_{60} = \text{soil respiration rate} \div [(80 - \text{\%WFPS}) \times 0.03] + 0.4$$

When the soil water content or WFPS exceeds 80%, soil respiration may be restricted by the wet conditions and should not be measured. The relationship between WFPS and soil respiration has been evaluated primarily in the laboratory and remains to be tested in the field (Parkin et al., 1996).

Table 1. General soil respiration class ratings and soil condition at optimum soil temperature and moisture conditions, primarily for agricultural land uses (Woods End Research, 1997).

Soil respiration (lbs CO_2-C/a/d)	Class	Soil condition
0	No soil activity	Soil has no biological activity and is virtually sterile.
< 9.5	Very low soil activity	Soil is very depleted of available organic matter and has little biological activity.
9.5 - 16	Moderately low soil activity	Soil is somewhat depleted of available organic matter, and biological activity is low.
16 - 32	Medium soil activity	Soil is approaching or declining from an ideal state of biological acitivity.
32 - 64	Ideal soil activity	Soil is in an ideal state of biological activity and has adequate organic matter and active populations of microorganisms.
> 64	Unusually high soil activity	Soil has a very high level of microbial activity and has high levels of available organic matter, possibly from the addition of large quantities of fresh organic matter or manure.
Conversion of Woods End Solvita respiration levels: (mg CO_2/kg/wk) x 0.039 x (1.2 g/cm^3) x (7.6 cm depth) ÷ 10 x 0.89 = (lbs CO_2-C/acre/day). It was assumed all respiration was coming from a 7.6 cm depth with an average bulk density of 1.2 g/cm^3 (Doran et al., 1997).		

A high soil respiration rate, indicative of high biological activity, can be a good sign of rapid decomposition of organic residues into nutrients available for plant growth. However, decomposition of the stable organic matter is detrimental to many physical and chemical processes such as aggregation, cation exchange, and water holding capacity. Also, immediately following a tillage operation, CO_2 evolution can rise dramatically due to exposure of organic matter to organisms and oxygen. Also, soil respiration can rise dramatically after rainfall (Rochette et al., 1991). The rise in soil respiration is affected by the length of time the soil is dry before the rainfall event.

Under dry conditions, soil respiration tends to be higher in the crop row than in the interrow (Rochette et al., 1991). The higher respiration rates are attributed to the contribution from plant roots. Under wet conditions, there tends to be no difference in respiration between the row and interrow. When the soil interrow is compacted (wheel track) and the soil is wet, soil respiration tends to be lower than in the row. The lower soil porosity accounts for the lower respiration rate under compacted conditions.

Biological activity is a direct reflection of the degradation of organic matter in the soil. This degradation indicates that two processes are occurring: (1) loss of soil carbon and (2) turnover of nutrients (Parkin et al., 1996). Some optimum soil respiration rate, that balances the long-term detrimental aspects of soil carbon loss and soil nutrient turnover, must be defined .

Conversions

kg CO_2-C/ha/d = lbs CO_2-C/a/d x 1.12

g CO_2-C/m^2/d = lbs CO2-C/a/d ÷ 11.2

kg CO_2-C/ha/d = g CO_2-C/m^2/d x 10

References

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2. Infiltration

Introduction

Infiltration is the process of water entering the soil. The rate at which water enters the soil is the infiltration rate, which is dependent on the soil type; soil structure, or amount of aggregation; and the soil water content (Lowery et al., 1996). The initial soil water content at time of measurement affects the ability of the soil to pull additional water into the soil. Therefore, the infiltration rate will be higher when the soil is dry than when it is wet. This factor is important when comparing infiltration measurements of different soils. The soils should have similar moisture content when taking the measurements.

Tillage will affect the infiltration rate. Immediately after tillage, improved infiltration may occur due to the loosening of surface crusts or compacted areas. Tillage fluffs up the soil. However, tillage further disrupts aggregates and soil structure, creating the potential for compaction, surface crusting, and loss of continuous surface connected pores. Compacted soils will have less pore space, resulting in lower infiltration rates. Soils that tend to form surface crusts, which seal the soil surface, can have severely reduced infiltration rates.

Interpretations

Since infiltration is affected by the initial water content at the time of measurement, it is important that the soil water content be similar when comparing infiltration rates from different sites. The infiltration test in the soil quality kit requires two 1-inch depths of water to be applied consecutively. Application of the first inch of water is used to wet the soil, and the second inch of water determines the infiltration rate. This procedure is an attempt to standardize the soils for differences in initial water content. Infiltration rates are best determined when the soil is at or near field capacity, usually 12 to 48 hours after the soil has been thoroughly wetted (i.e., soaking rain or irrigation).

The infiltration rate is sensitive to near-surface conditions and is subject to significant change with soil use, management, and time. It is affected by the development of plant roots, earthworm burrows, soil aggregation, and by overall increases in stable organic matter (Sarrantonio et al., 1996). Infiltration is rapid into large continuous pores in the surface. Infiltration is decreased when the size

Table 2. Steady infiltration rates for general soil texture groups in very deeply wetted soil (Hillel, 1982).

Soil type	Steady infiltration rate (inches per hour)
Sands	> 0.8
Sandy and silty soils	0.4 - 0.8
Loams	0.2 - 0.4
Clayey soils	0.04 - 0.2
Sodic clayey soils	< 0.04

or amount of pore space is reduced from conditions such as structure breakdown, pore clogging by lodged particles, or slower movement of deeper water as it reaches denser subsoils (Donahue et al., 1977).

Texture, or the percentage of sand, silt, and clay will affect the infiltration rate. Usually sandy soils will have rapid infiltration rates. Some typical values for steady infiltration rates (After long continuous wetting, the rate of infiltration becomes steady.) for general soil texture groups are shown in Table 2. However, the values in Table 2 can be considerably higher in well aggregated or cracked soils and during initial stages of wetting; these values can be lower if surface crusting occurs (Hillel, 1982). Soil structure greatly influences the movement of water into the soil.

Table 3 shows the infiltration rate in minutes per inch and inches per hour and the associated infiltration class. These classes are the soil permeability classes historically used in Soil Survey. Classes are estimated from soil properties and indicate a steady infiltration rate.

Table 3. Infiltration rates and classes.

Infiltration rate (minutes per inch)	Infiltration rate (inches per hour)	Infiltration class
< 3	> 20	Very rapid
3 to 10	6 to 20	Rapid
10 to 30	2 to 6	Moderately rapid
30 to 100	0.6 to 2	Moderate
100 to 300	0.2 to 0.6	Moderately slow
300 to 1,000	0.06 to 0.2	Slow
1,000 to 40,000	0.0015 to 0.06	Very slow
> 40,000	< 0.0015	Impermeable

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3. Bulk Density

Introduction

Bulk density is defined as the ratio of oven-dried soil (mass) to its bulk volume, which includes the volume of particles and the pore space between the particles. It is dependent on the densities of the soil particles (sand, silt, clay, and organic matter) and their packing arrangement. Mineral particle densities usually range from 2.5 to 2.8 g/cm^3, while organic particles are usually less than 1.0 g/cm^3. Bulk density is a dynamic property that varies with the structural condition of the soil. This condition can be altered by cultivation; trampling by animals; agricultural machinery; and weather; i.e., raindrop impact (Arshad et al., 1996). Compacted soil layers have high bulk densities, restrict root growth, and inhibit the movement of air and water through the soil.

Interpretations

Soil bulk density can serve as an indicator of compaction and relative restrictions to root growth (See Table 4). Typical soil bulk densities range from 1.0 to 1.7 g/cm^3, and generally increase with depth in the soil profile (Arshad et al., 1996). In soils containing high amounts of swelling clays, bulk densities will vary with the water content, which should be measured at the time of sampling.

Table 4. General relationship of soil bulk density to root growth based on soil texture.

Soil texture	Ideal bulk densities (g/cm^3)	Bulk densities that may affect root growth (g/cm^3)	Bulk densities that restrict root growth (g/cm^3)
sands, loamy sands	< 1.60	1.69	> 1.80
sandy loams, loams	< 1.40	1.63	> 1.80
sandy clay loams, loams, clay loams	< 1.40	1.60	> 1.75
silts, silt loams	< 1.30	1.60	> 1.75
silt loams, silty clay loams	< 1.40	1.55	> 1.65
sandy clays, silty clays, some clay loams (35-45% clay)	< 1.10	1.49	> 1.58
clays (> 45% clay)	< 1.10	1.39	> 1.47

Comments

Bulk density values are also required for converting soil water content in percent by weight (gravimetric) to percent by volume (volumetric):

Volumetric water content (g/cm^3) = soil water content (g/g) x bulk density (g/cm^3)

and to calculate porosity, which is the amount of pore space in the soil:

soil porosity (%) = 1 - (soil bulk density ÷ 2.65).

References

Arshad, M.A., B. Lowery, and B. Grossman. 1996. Physical tests for monitoring soil quality. p.123-142. In: J.W. Doran and A.J. Jones (eds.) Methods for assessing soil quality. Soil Sci. Soc. Am. Spec. Publ. 49. SSSA, Madison, WI.

4. Electrical Conductivity

Introduction

The electrical conductivity (EC) of soil-water mixtures indicates the amount of salts present in the soil. All soils contain some salts, which are essential for plant growth. However, excess salts will hinder plant growth by affecting the soil-water balance. Soils containing excess salts occur both naturally and as a result of soil use and management. Salt-affected soils are largely found in the western arid and semiarid areas of the country, where the annual rainfall is low, allowing salts to accumulate in the soil profile. The electrical conductivity measurement detects the amount of cations or anions (salts) in solution; the greater the amount of anions or cations, the greater the electrical conductivity reading. The ions generally associated with salinity are Ca^{2+}, Mg^{2+}, K^{+}, Na^{+}, H^{+} (cations), or NO_3^{-}, SO_4^{-}, Cl^{-}, HCO_3^{-}, OH^{-} (anions).

Interpretations

In general, $EC_{1:1}$ values between 0 and 0.8 dS/m are acceptable for general crop growth. Site specific interpretations for soil quality will depend on specific land use and crop tolerance. Table 5 shows the soil salinity class and general crop and microbial responses for each class.

Table 5. Electrical conductivity measurement and salinity classes for a 1:1 soil:water suspension.

Electrical Conductivity (dS m^{-1} at 25 C)	Salinity class	Crop response	Microbial response
0 - 0.98	Non saline	Almost negligible effects	Few organisms affected
0.98 - 1.71	Very slightly saline	Yields of very sensitive crops restricted	Selected microbial processes altered (nitrification/denitrification)
1.71 - 3.16	Slightly saline	Yields of most crops restricted	Major microbial processes influenced (respiration/ammonification)
3.16 - 6.07	Moderately saline	Only tolerant crops yield satifactorily	Salt tolerant microorganisms predominate (fungi, actinomycetes, some bacteria)
> 6.07	Strongly saline	Only very tolerant crops yield satisfactorily	A select few halophilic organisms are active

Adapted from Soil Survey Staff (1993), Janzen (1993), and Smith and Doran (1996). Conversions from the saturation paste extract to the 1:1 soil:water suspensions were perfomed using the regression equation (y = 2.75 x -0.69) developed by Hogg and Henry (1984).

Table 6 provides general salt tolerance ratings for selected crops. These ratings apply to soils in which chloride (Cl^-) is the predominant anion. The EC of soils containing gypsum will tolerate 1 dS/m higher than those listed in this table (Tanji, 1990). Consult a local Soil Survey to determine if gypsum is present in the soil of interest.

Table 6. Salt tolerance of selected crops (Tanji, 1990).

Crop	Rating	Crop	Rating	Crop	Rating
Alfalfa	MS	Clover, iadino	MS	Loquat	S
Alkali grass, Nuttall	T	Clover, red	MS	Love grass	MS
Alkali sacaton	T	Clover, strawberry	MS	Mango	S
Almond	S	Clover, sweet	MT	Milkvetch, Cicer	MS
Apple	S	Clover, white Dutch	MS	Millet, foxtail	MS
Apricot	S	Corn	MS	Muskmelon	MS
Artichoke	MT	Corn (forage)	MS	Oat grass, tall	MS
Asparagus	T	Corn, sweet	MS	Oats (forage)	MS
Avocado	S	Cotton	T	Okra	S
Barley	T	Cowpea	MT	Olive	MT
Barley (forage)	MT	Cowpea (forage)	MS	Onion	S
Bean	S	Cucumber	MS	Orange	S
Beet, red	MT	Currant	T	Orchard grass	MS
Bentgrass	MS	Dallis grass	MS	Panic grass, blue	MT
Bermuda grass	T	Date palm	T	Papaya	MT
Blackberry	S	Eggplant	MS	Rape	MT
Bluestem, Angleton	MS	Fescue, tall	MT	Parsnip	S
Boysenberry	S	Fescue, meadow	MT	Passion fruit	S
Broad bean	MS	Fig	MT	Pea	S
Broccoli	MS	Flax	MS	Peach	S
Brome, mountain	MT	Foxtail, meadow	MS	Pear	S
Brome, smooth	MS	Gooseberry	S	Pepper	MS
Brussels sprouts	MS	Grama, blue	MS	Persimmon	S
Buffelgrass	MS	Grape	MS	Pineapple	MT
Burnet	MS	Grapefruit	S	Plume, prune	S
Cabbage	MS	Guar	T	Pomegranate	MT
Canary grass, reed	MT	Guayule	T	Potato	MS
Carrot	S	Harding grass	MT	Pummelo	S
Castorbean	MS	Jojoba	T	Pumpkin	MS
Cauliflower	MS	Jujube	MT	Radish	MS
Celery	MS	Kale	MS	Rescue grass	MT
Cherimoya	S	Kaller grass	T	Raspberry	S
Cherry, sweet	S	Kenaf	MT	Rhodes grass	MT
Cherry, sand	S	Kohlrabi	MS	Rice, paddy	S
Clover, alsike	MS	Lemon	S	Rose apple	S
Clover, berseem	MS	Lettuce	MS	Rye	T
Clover, hubam	MT	Lime	S	Rye (forage)	MS

Table 6. Continued.

Crop	Rating	Crop	Rating	Crop	Rating
Ryegrass, perennial	MT	Sudan grass	MT	Wheat, semidwarf	T
Safflower	MT	Sugar beet	T	Wheat, durum	T
Salt grass, desert	T	Sugarcane	MS	Wheat, durum (forage)	MT
Sapote, white	S	Sunflower	MS	Wheat (forage)	MT
Sesame	S	Sweet potato	MS	Wheat grass, standard	MT
Sesbania	MS	Tangerine	S	Wheat grass, fairway	T
Sirato	MS	Timothy	MS	Wheat grass, interm.	MT
Sorgham	MT	Tomato	MS	Wheat grass, slender	MT
Soybean	MT	Trefoil, narrowleaf	MT	Wheat grass, tall	T
Sphaerophysa	MS	Triticale	T	Wheat grass, western	MT
Spinach	MS	Turnip	MS	Wild rye, Altai	T
Squash, scallop	MS	Vetch, common	MS	Wild rye, beardless	MT
Squash, zucchini	MT	Watermelon	MS	Wild rye, Canadian	MT
Strawberry	S	Wheat	MT	Wild rye, Russian	T

Rating	EC range for 1:1 soil:water suspension for which yield reductions occur
S = Sensitive	> 0.90 dS/m
MS = Moderately sensitive	> 1.40 dS/m
MT = Moderately tolerant	> 2.50 dS/m
T = Tolerant	> 4.00 dS/m

Excess salts affect plant growth by (1) direct toxicities; e.g., boron; (2) disrupting the ionic balance in the plant; (3) interfering with nutrient uptake; e.g., blossom-end rot of tomatoes due to high salt interference with calcium uptake; and (4) reducing the availability of water by lowering the osmotic potential (Fitter and Hay, 1987). Excess sodium (Na^+), often expressed as *exchangeable sodium percentage* (ESP), can deteriorate soil structure by dispersing soil clays.

Considerations

The electrical conductivity of a solution is affected by temperature. Generally the electrical conductivity of a solution increases with temperature at a rate of approximately 1.9% per 1°C increase (Rhoades, 1993). The conductivities in Table 5 are standardized at 25°C. Most EC meters adjust for deviations from 25°C within a specific temperature range. Therefore, conductivity measurements must be taken within this temperature range (Refer to instructions packaged with the meter.) to avoid under- or overestimating the electrical conductivity.

Generally, the effects of soil moisture on the EC measurement will be negligible when soil water content is at or below field capacity. If water content is above field capacity, adjustments should be made to maintain a 1:1 ratio of soil to water. Another approach would be to air-dry the soil if it is too wet.

When distilled water is not available, tap or rain water can be used. Measure the conductivity of the water source, and subtract the water source EC value from the sample EC value.

The relationship between electrical conductivity and salt concentration is only approximate. General relationships that have been established are (Rhoades, 1996):

1) Total cation (or anion) concentration: meq/L ≅ 10 x EC (dS/m).
2) Total dissolved solids: mg/L ≅ 640 x EC (dS/m).
3) Osmotic pressure: kPa (at 25°C) ≅ 36 x EC (dS/m).

Where NO_3^- is the predominant ion in the soil solution, a very useful relationship has been established between the EC (in 1:1 soil to water mixture) readings and soil nitrate (NO_3^-) concentrations (Smith and Doran, 1996).

EC (dS/m) x 140 ≅ mg NO_3^--N/kg of soil

This relationship assumes the complete extractability of NO_3^- in water and that NO_3^- is the major anion in the soil solution.

Conversions

1 dS/m (decisiemens per meter) = 1 mmhos/cm (millimhos per centimeter)
1 dS/m (decisiemens per meter) = 1000 μS/cm (microsiemens per centimeter)
1000 μS/cm (microsiemens per centimeter) = mS/cm (millisiemens per centimeter)

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5. Soil pH

Introduction

Soil pH is a measure of the acidity or alkalinity of a soil, which affects the availability of plant nutrients, activity of microorganisms, and the solubility of soil minerals. Major factors affecting soil pH are temperature and rainfall, which control the intensity of leaching and soil mineral weathering. Acidity is generally associated with leached soils; alkalinity generally occurs in drier regions. However, agricultural practices, such as liming or addition of ammonium fertilizers, can alter soil pH. The pH measurement is actually measuring the hydrogen ion activity [H^+] in the soil solution.

Interpretations

In general, pH values between 6 and 7.5 are optimum for general crop growth. Site specific interpretations for soil quality will depend on specific land use and crop tolerance.

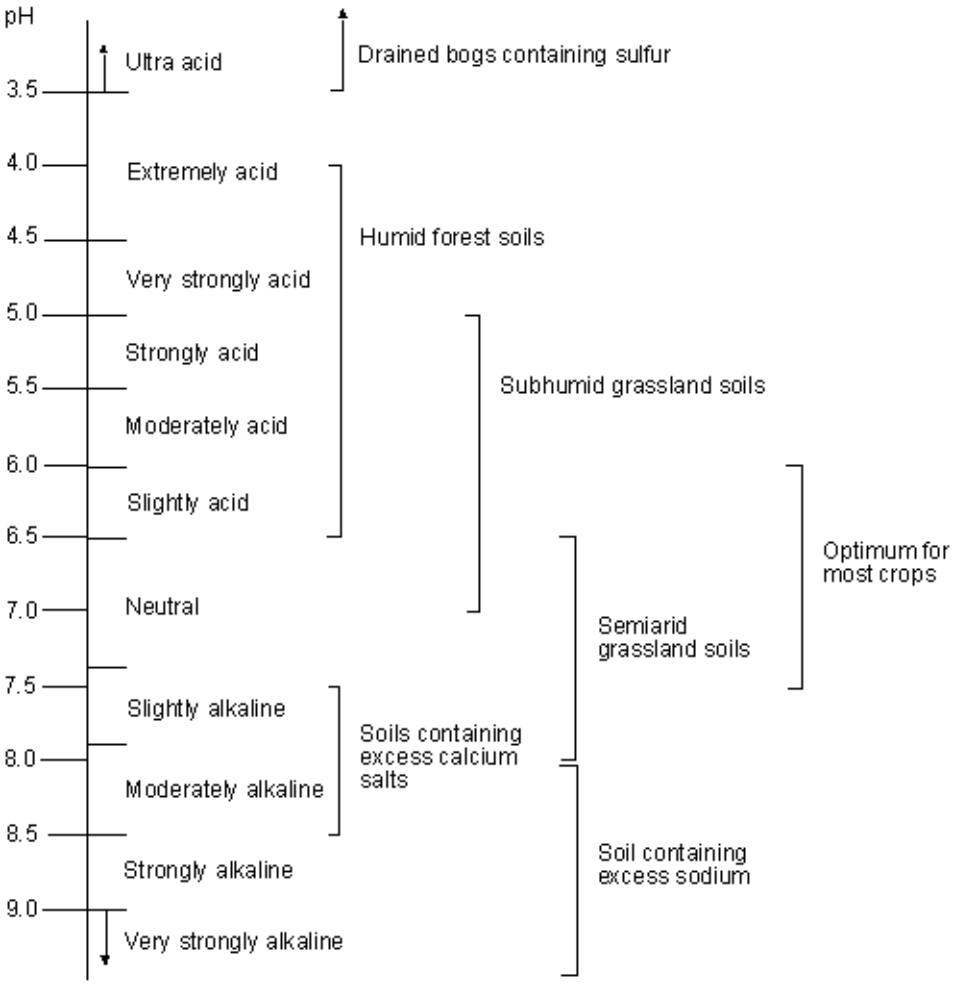


Figure 1. Soil pH, ranges for pH classes, and associated soil conditions. Adapted from the National Soil Survey Manual (1993) and Troeh and Thompson (1993).

Table 7. Suitable soil pH ranges for selected crops (Whittaker et al., 1959).

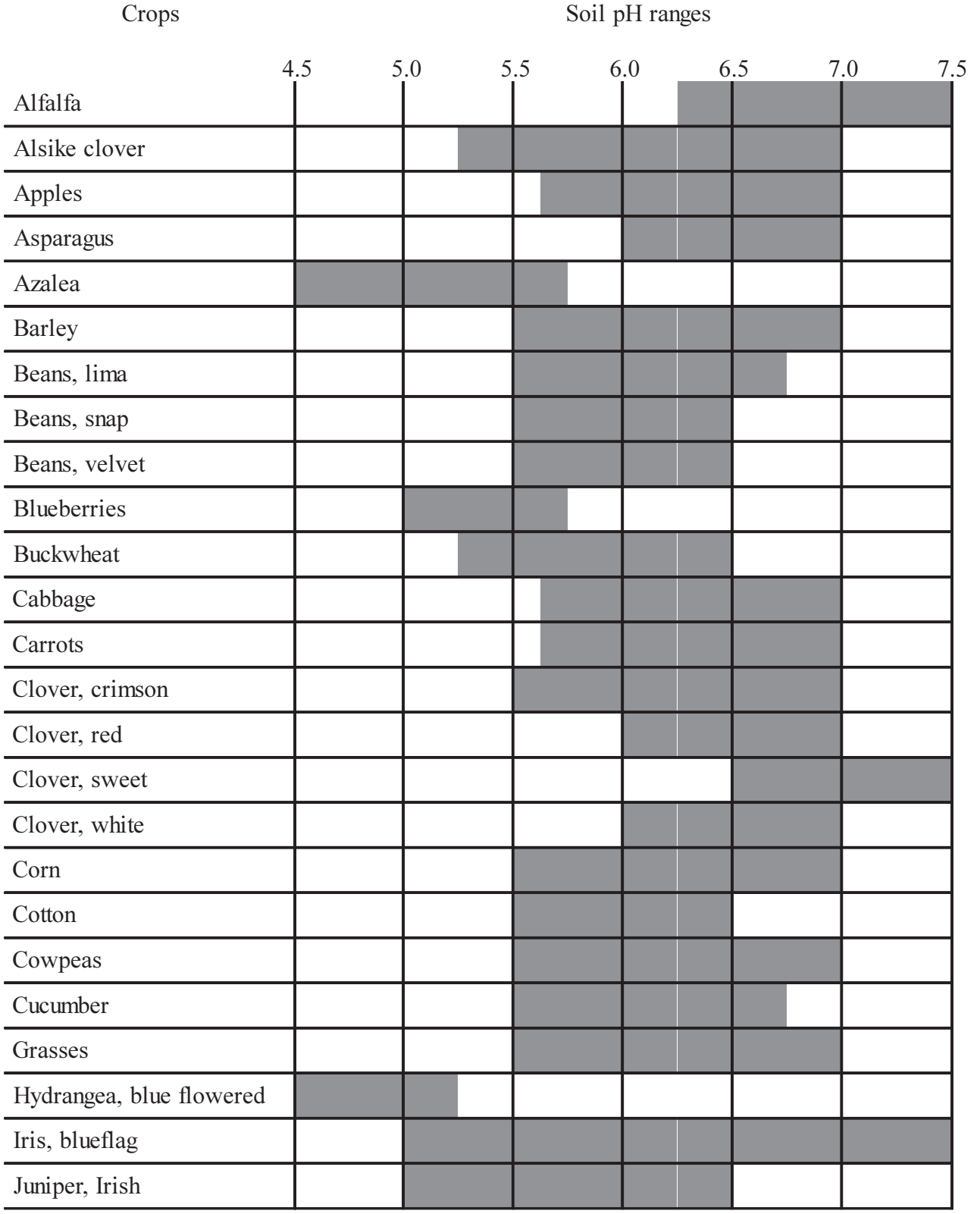
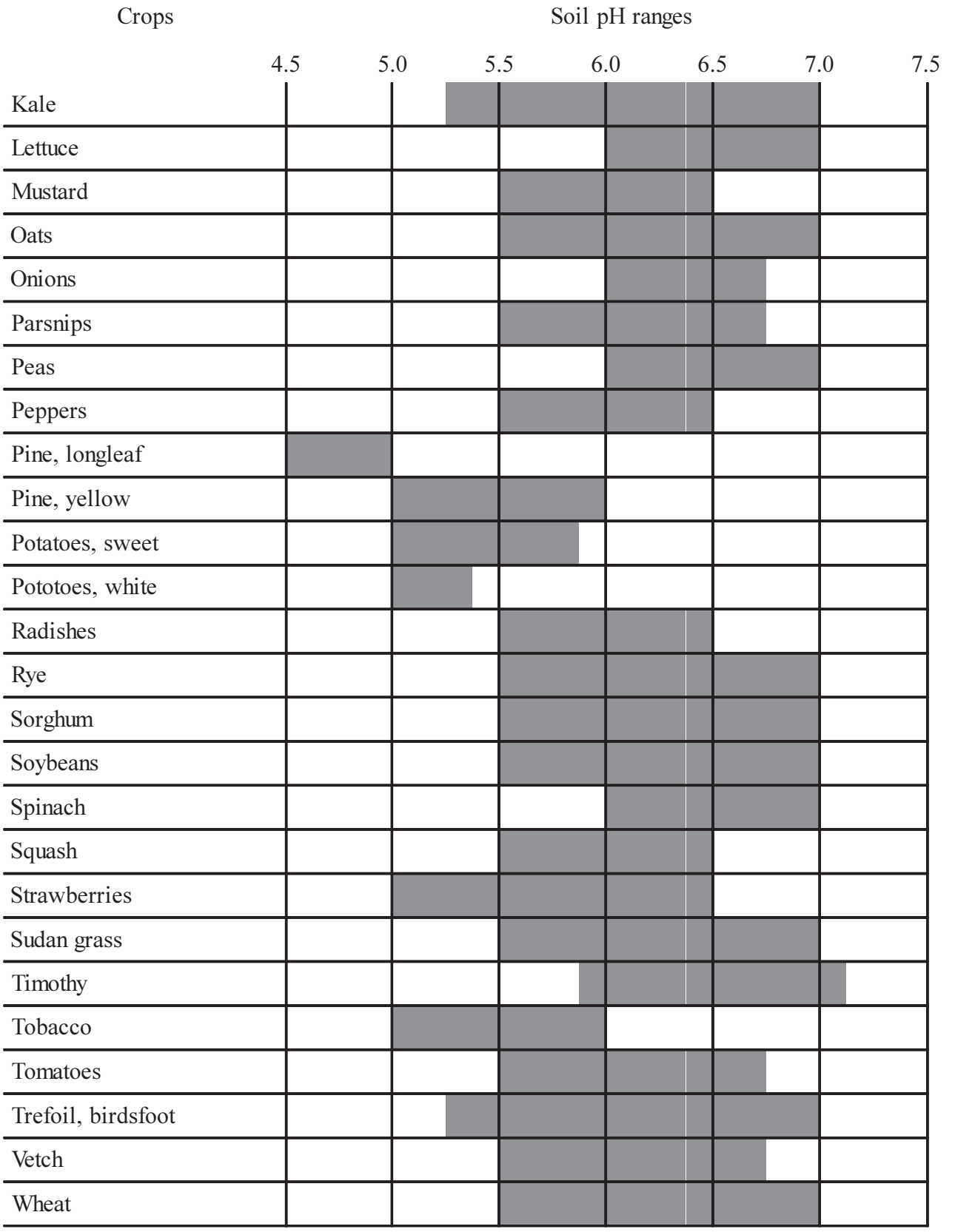


Table 7. Continued.



Nutrient Availability

Soil pH affects the availability of nutrients to plants or crops (Figure 2). Nutrient availability is affected by changes in the solubility of soil minerals. Most minerals are more soluble in acid soils than in neutral or slightly basic soils. The greatest availability for most nutrients is between pH 6 and 7 (Figure 2). Where nutrients are shown interlocking in Figure 2, those nutrients at that pH combine to form insoluble compounds, reducing their availability. Soil pH also affects the activity of beneficial microorganisms, which affects nutrient availability. In general, fungi function at a wide pH range, but bacteria and actinomycetes function better at intermediate and higher pH.

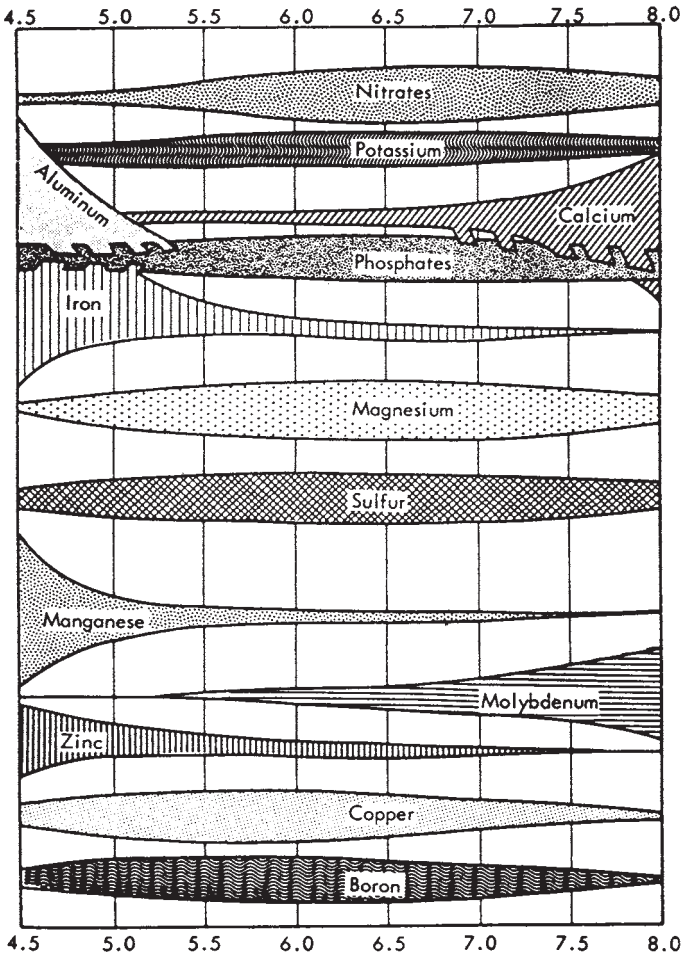


Figure 2. Nutrient availability based on pH of mineral soils (Soils Handbook, Kentucky Agr. Exp. Stn. Misc. 383, 1970, p.28).

Comments

The presence of salts affects soil pH by decreasing the reading by 0.2 to 0.3 pH units (Thomas, 1996). To mask the effects of salts, a 0.01 M $CaCl_2$ solution has been commonly used instead of distilled water.

Declining pH is a sign of inefficient N use where ammonia based fertilizers are used (see Smith and Doran, 1996).

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6. Soil Nitrate

Introduction

Soil nitrate (NO_3^-) is a form of inorganic nitrogen (N) that is available for use by plants. It forms from the mineralization (by microorganisms) of organic forms of N (i.e., soil organic matter, crop residue, and manure) in the soil. The rate of N mineralization is dependent on the amount of soil organic N, water content, temperature, pH, and aeration. Crop needs are met by soil-derived mineral-N and by fertilizer-N. Efficient management of soil N requires knowledge of crop needs for N and the amount of soil-derived N. Nitrate is mobile in soil, so it can be leached with percolating water below the root zone. All soils lose a small amount of nitrate to groundwater, including soils under natural vegetation. When amounts leach that are greater than what occurs naturally, we need to be concerned. Nitrate is not a contaminant until it leaches below the root zone or is transported off-site in surface runoff. When leached to groundwater, there is a human and animal health risk. In surface water systems, nitrate can contribute to euthrophication.

Interpretations

The amount of residual nitrate-N in the soil at any one time is a function of the rate at which microorganisms decompose soil organic matter (Figure 3). This rate is dependent on temperature, moisture, aeration, type of organic residues, pH, and other factors (Dahnke and Johnson, 1990). Also, once soil nitrate has formed, it is subject to leaching, fixation, denitrification, and plant uptake (Figure 3). Therefore, it is difficult to interpret the nitrate-N content in terms of how much and when N will be available to meet crop needs. However, residual nitrate-N tests can be useful in determining fertilizer-N needs of crops in certain regions during specific times of the year and at specific crop growth stages (Dahnke and Johnson, 1996). **For interpretations of residual nitrate-N tests for crop needs, consult local or regional calibrations.**

Any amount of nitrate in the soil that is not used by the crop may potentially be leached from the root zone and become an environmental liability. Nitrate is not adsorbed on to soil particles unless they have a positive charge. Therefore, nitrate can readily move with percolating water out of the root zone and into groundwater or into surface waters through subsurface flow (Figure 3). Acidic soils of the humid tropics contain a significant amount of positively charged soil particles which can hold nitrate and keep it from leaching.

Nitrogen Cycling

In general, soil nitrate levels will change significantly during the course of the year and from week to week. Soil nitrogen is continuously cycling, moving from one form to another (Figure 3). It is derived primarily from atmospheric nitrogen gas (N_2). Soil microorganisms fix N_2 to produce organic nitrogen, which becomes part of the soil organic matter. The decomposition of organic matter converts some organic nitrogen into mineral nitrogen (mineralization). Ammonium (NH_4^+) produced by mineralization (an intermediate step) can be converted to nitrate by specific microorganisms (nitrification). The nitrate formed is then available for uptake by plants or microorganisms and is converted to organic forms of nitrogen (immobilization). Under water logged or anaerobic conditions, nitrate may be substituted for oxygen and ultimately released to the atmo

sphere as elemental nitrogen or nitrous oxide gas (N_2 or N_2O) [denitrification]. Each N transformation depends on the activity and abundance of a specific population of microorganisms that require different sets of optimal environmental conditions.

Primary *sources* of nitrates:

- addition of fertilizers containing nitrate,
- microbial conversion of ammonium fertilizers to nitrate-N,
- microbial conversion of organic N (i.e., soil organic matter and manures) to nitrate-N.

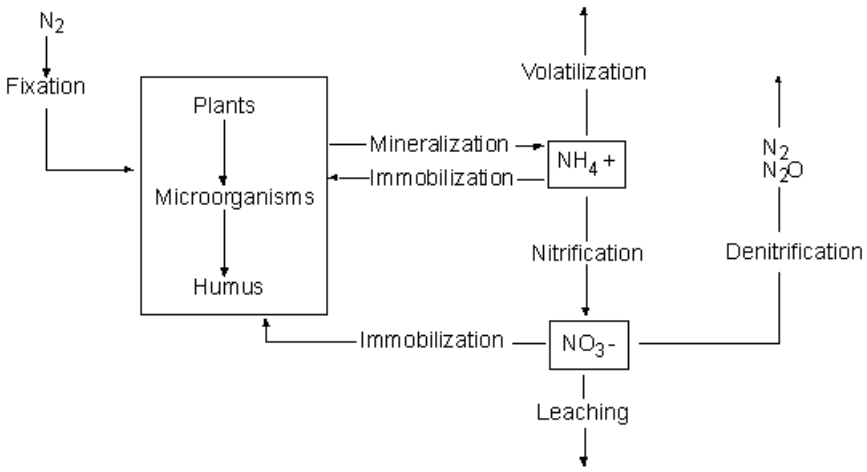

Figure 3. Generalized soil nitrogen cycle.

Primary *fates* of nitrates:

- utilization by microorganisms or plant roots (immobilization)
- leached below the root zone
- moved off-site in surface runoff
- microbial conversion of nitrate-N to nitrogen gas

Comments

The nitrate/nitrite test strips can determine both nitrate and nitrite concentrations (two test pads on each test strip). Nitrite levels in soils are usually not detectable (in a transition state); therefore, its measurement is not warranted. The nitrate test pad on the test strip measures the sum of both nitrate-N and nitrite-N present in the sample. If nitrite is detected in the sample, the amount can be subtracted from the nitrate reading to get the actual amount of nitrate-N in the sample.

Spring soil nitrate-N tests can be used to assess the effectiveness of soil and cropping management practices in providing sufficient N for optimal crop yields. For example, for corn in the Midwest, values of 20-25 ppm nitrate-N in top foot (30 cm) of soil are needed (14-16 ppm is the threshold for soils receiving manure or having alfalfa or soybeans as the previous crop) [Allan et al., 1996; page 196].

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7. Aggregate Stability

Introduction

Aggregate stability is a measure of the vulnerability of soil aggregates to external destructive forces (Hillel, 1982). An aggregate consists of several soil particles bound together. The destructive force in this test is flowing water. Aggregates that stand up to the forces of water are called water stable aggregates (WSA). In general, the greater the percentage of stable aggregates, the less erodible the soil will be. Soil aggregates are a product of the soil microbial community, the soil organic and mineral components, the nature of the above-ground plant community, and ecosystem history. They are important in the movement and storage of soil water and in soil aeration, erosion, root development, and microbial community activity (Tate, 1995). Breakdown of aggregates is the first step to crust development and surface sealing, which impedes water infiltration and increases erosion. Soil aggregation can change over a period of time, such as in a season or year. Aggregates can form, disintegrate, and reform periodically (Hillel, 1982).

Interpretations

The percentage of water stable aggregates indicates the amount resistant to disturbance by flowing water. **In general, greater amounts of stable aggregates are better for soil quality**.

Aggregates improve soil quality by:

- protecting soil organic matter entrapped in the aggregates from exposure to air and microbial decomposition,
- decreasing soil erodibility,
- improving water and air movement (Aggregates increase the amount of large pore spaces.),
- improving the physical environment for root growth,
- improving soil organism habitat.

Aggregate stability is affected by the amount and type of the following soil constituents (Kemper, 1966):

Soil Organic Matter content:

Aggregate stability generally increases with organic matter content (Table 1). The effect is more pronounced in soils containing small amounts of clay. Generally, increases in organic matter above 2% do not increase aggregate stability appreciably.

Soil Clay content:

Aggregate stability is affected by the amount and type of clay in the soil and generally increases with clay content (Table 1). This effect decreases at higher clay contents (Table 1). In general, high surface-area clays (i.e., montmorillonite) tend to cause greater aggregation than low surface-area clays (i.e., kaolinite).

Aluminum and Iron Oxide content:

Aggregate stability generally increases with free iron oxide content. In general, free aluminum oxides do not appreciably increase aggregate stability.

Calcium Carbonate content:

The calcium carbonate content generally does not appreciably affect aggregate stability.

Exchangeable Sodium content:

Aggregate stability decreases with increasing amounts of exchangeable sodium. In general, water stable aggregates are nonexistent in soils with greater than 20% exchangeable Na^+.

Table 8 contains suitable values for aggregate stability based on soil organic matter and clay content. A suitable range of values could be developed for a soil using the aggregate stability values for the organic matter content and clay content as end members to the range. For example, for a soil with 2% organic matter and 10% clay, the suitable aggregate stability range (taken from Table 8) would be 65 to 75% water stable aggregates.

Table 8. Suitable values for % water stable aggregates based on clay and organic matter content (Kemper, 1966). Water stable aggregates for % clay should be read independently of % organic matter in this table.

Organic Matter (%)	Water Stable Aggregates (%)		Clay (%)	Water Stable Aggregates (%)
0.4	53		5	60
0.8	66		10	65
1.2	70		20	70
2	75		30	74
4	77		40	78
8	81		60	82
12	85		80	86

Aggregate stability values are based on 519 soil samples from the arid, semiarid, and subhumid regions of the United States and Canada. The majority of the samples were from cultivated areas, but a large number were taken from virgin or replanted grasslands (Kemper, 1966).

Soil aggregates are divided into two general groups based on aggregate size (diameter):

- **Microaggregates** (less than 250 μm) consist of primary soil particles and smaller microaggregates bound together. Binding agents include:
  - humified organic matter (organic polymers)
  - polyvalent metals or cations
  - roots and fungal hyphae
  - polysaccharides
  - plant and microbial debris (encrusted)
  - iron and aluminum amorphous oxides
- **Macroaggregates** (> 250 μm) consist of microaggregates bound together. Major binding agents are:
  - fungal hyphae
  - fibrous roots
  - polysaccharides

iron and aluminum oxides (soils that contain more than 10% iron and aluminum oxides)

The size of the water stable aggregates measured in the soil quality kit are macroaggregates.

Macroaggregates form readily under the following conditins:

- under pasture or forage grasses (dense, fibrous root mass),
- where organic residues have been added,
- where large amounts of microaggregates (< 250 μm diameter) are present.

Differences between micro- and macroaggregates include the following:

- Macroaggregates are more sensitive to changes in management than microaggregates and thus, are considered a better indicator of changes in soil quality. Macroaggregate stability depends on management because of the transient nature of the binding agents.
- Macroaggregates form more rapidly than microaggregates.
- Carbon is more stable in microaggregates than in macroaggregates.
- Microaggregates are more water stable than macroaggregates.
- When the proportion of macro- to microaggregates increases, soil quality increases.

Considerations and Comments

The temperature of the water used to sieve the soils should be maintained within the range of 22 to 25°C (71.5 to 77°F). At higher water temperatures, aggregate stability tends to decrease.

To make observational estimations of aggregate stability or relative comparisons, weighing and drying of the aggregates are not necessary.

When dry aggregates are wetted up too quickly at atmospheric pressure, disintegration and slaking can result. Upon rapid wetting, capillary water entering the pores causes air entrapped inside the aggregate pores to increase in pressure causing them to rupture (Kemper and Rosenau, 1986).

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8. Soil Slaking

Introduction

Slaking is the process of fragmentation that occurs when aggregates are suddenly immersed in water (Chan and Mullins, 1994). Slaking occurs because the aggregates are not strong enough to withstand the stresses of rapid water uptake. At fast rates of wetting, internal stresses arise from differential swelling and air entrapment in the soil aggregate (Kay, 1998). These stresses may be released through the creation of an increasingly extensive network of failure zones in the soil fragments or aggregates. The differences between tests of aggregate stability and slaking are the type of stress applied and the size of aggregates or soil fragments used. The slake test is a qualitative and simpler test to perform. The two tests may not necessarily yield the same results.

Interpretations

The slake test in the kit yields a stability rating of 0 to 6 (Herrick, 1998). Soil fragments or aggregates which fall into classes 0 to 3 are relatively unstable. Class 4 indicates some stability, but very little strength. Classes 5 and 6 represent relatively stable soil fragments or aggregates. Soil strength relates to the ability of the soil to resist loss of its structure.

Stability ratings of soil surface crust fragments are interpreted differently. Soil crust formation in agricultural systems reduces the capacity of the soil to function (i.e., soil crusts can reduce air and water movement into the soil and can inhibit seedling germination). In general, weakly formed or unstable crusts are better than very strong or stable crusts, which have a greater potential to lower soil quality. The subsurface fragments or aggregates directly beneath the crust are tested to provide an indication of the potential for future slaking and crusting of the soil (potential of crust formation).

Slaking is affected by:

- the soil water content,
- rate of wetting,
- texture,
- clay mineralogy, and
- organic matter content.

Slaking is more severe when the soil is initially dry than when it is moist. For loamy soils, the pressure of entrapped air has been shown to be more important. For clayey soils, differential swelling was shown as the more important process (Chan and Mullins, 1994). In general, organic matter can influence both the rate of wetting and the resistance to stress generated during wetting (Kay, 1998). The stability of aggregates is strongly dependant on the rate of wetting; therefore, aggregate stability declines as the rate of wetting increases.

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9. Earthworms

Introduction

Earthworm populations may vary with site characteristics (food availability and soil conditions), season, and species. Populations are highly variable in space and time, which can range from less than 10 to greater than 10,000 individuals per square meter (Curry, 1998). However, not all areas or soils support earthworms. Either they were not introduced, or environmental conditions are not favorable. Earthworms generally increase soil microbial activity and soil chemical fertility and enhance soil physical properties.

Interpretations

About 10 earthworms per square foot of soil (100 worms/m^2) is generally considered a good population in agricultural systems. Populations generally do not exceed 20 per square foot of soil (200 worms/m^2) in cultivated systems (Edwards, 1983). In grassland systems, populations can generally range up to about 50 per square foot of soil (500 worms/m^2) [Edwards, 1983]. The hand digging method does not capture certain deep-burrowing or fast moving earthworm species. However, hand digging is one of the best methods available.

Earthworms improve soil quality by:

- increasing the availability of nutrients. (Available plant nutrients (N, P, & K) tend to be higher in fresh earthworm casts than in the bulk soil.) [Edwards et al., 1995];
- accelerating the decomposition of organic matter by incorporating litter into the soil and activating both mineralization and humification processes;
- improving soil physical properties, such as aggregation and soil porosity;
- suppressing certain pests or disease organisms; and
- enhancing beneficial microorganisms.

Earthworms and soil aggregation processes:

- Fresh earthworm casts are often highly dispersed, nearly saturated masses of soil, which are unstable and susceptible to erosion (Edwards et al., 1995). As earthworm casts age, they can become more stable. The organic matter content, wet-dry cycles, and fungal hyphae and other microbial products help to stabilize casts over time and improve the aggregation of soil.
- In general, the more sensitive the soil is to physical disturbance, the more effective casting is for stable aggregation but less effective for tensile strength (Schrader and Zhang, 1997).

Factors affecting earthworm populations include the following (Curry, 1998):

Tillage

- Tillage generally kills about 25% of the earthworm population. The indirect effects of tillage affects the remaining population. These indirect effects include increases in surface temperature, decreased soil moisture regimes, reduced litter input, and more rapid oxidation (decomposition) of crop residues.
- Earthworm populations are often greater under no-till than under conventional tillage. Large populations of both surface-dwelling and deep-burrowing earthworms are often

associated with improved soil physical conditions. Higher infiltration rates often occur in no-till than in conventional tillage systems due to (in part) the large number of macropores from earthworm activity.

Temperature

The optimum temperature range for earthworms is between 10 and 20°C. The upper lethal range is 25 to 35°C. Few species can tolerate temperatures below 0°C. Many species have behavioral and/or physiological adaptations that enable them to survive unfavorable conditions.

Soil Properties

- Medium textured soils are more favorable for earthworms than sandy or clayey soils.
- Depth of aeration in soils affects the deep-burrowing species.
- Soil pH affects earthworm populations. Earthworms are usually absent in soils with pH less than 3.5 and are scarce in soils with pH between 3.5 and 4.5. The majority of the earthworms live in soils with pH between 5.0 and 7.4.
- Quality and amount of food (organic matter) affect earthworm distribution and abundance.

Food Source

Litter, or organic, residue on the soil surface is the primary food source for earthworms in most ecosystems. However, dead roots and root exudates can also be important food sources. If the physical and chemical environments are not limiting, the quality and quantity of litter input frequently determines earthworm abundance.

Soil Disturbance

- Earthworm populations are generally higher in undisturbed soil systems.
- Population size depends on the severity and frequency of soil disturbance.
- If the soil disturbance is not repeated, earthworm populations can recover fairly rapidly (within a few years).

Soil Moisture

Soil moisture restrictions generally determine earthworm distributions and their activity.

Agrochemicals

- Pesticides, especially insecticides, can affect earthworm populations. The majority of triazine herbicides (i.e., atrazine, simazine, and cyanizine) are slightly toxic. Carbamate-based fungicides (i.e., carbendazim, benomyl, and thiophanate-methyl) are very toxic. Organophosphates (i.e., phorate, isozophos, chlorpyrifos, and ethoprophos) and most of the carbamate-based insecticides (i.e., carbaryl, carbofuran, methomy, and methiocarb) are toxic. Most of the nematicides (i.e., D-D, metham-sodium, and methyl bromide) have been reported to be toxic to earthworms (Edwards et al., 1995).
- Regular use of ammonium sulfate and anhydrous ammonia and sulfate coated urea has been shown to decrease earthworm populations (Edwards et al., 1995).

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10. Soil Physical Observations and Estimations

Topsoil Depth

Topsoil depth is important for water storage and nutrient supply for plant growth. Generally, removal of the topsoil will result in loss of soil fertility, water-holding capacity, soil organic carbon content, and productivity. Measurements of topsoil depth over time provide a good estimate of soil loss (erosion).

Interpretations

Change in topsoil thickness is usually a result of wind erosion, water erosion, deposition of material, or land leveling. Eroded soils will commonly have a reduced Ap horizon (plow layer) or topsoil thickness. Natural erosion occurs in the absence of human disturbances. However, it is the accelerated erosion caused by plowing, burning, overgrazing, and other management practices that remove the protective vegetative cover and results in loss of soil quality.

Root Growth

Depth of soil to a layer that would restrict root growth strongly affects crop production. Factors that influence rooting depth include high salt content and depth to bedrock, stone layer, hard pan, frozen layer, and water table (Arshad et al., 1996).

When continuous pores are present in the soil, roots will grow through these pores as a result of the low mechanical impedance. The distribution of roots in the soil profile is a function of soil depth, thickness, and mechanical resistance of the root-impeding soil layers (Bennie, 1996).

Interpretations

Roots growing through restrictive soil layers undergo morphological changes, particularly root stunting and thickening (Bennie, 1996). Impeded roots are generally shorter, thicker, and more irregularly shaped. The shorter root system will exploit a smaller soil volume for plant nutrients and water, causing the plant to maintain a higher than normal uptake rate of nutrients and water per unit of root length. Also, more photosynthetic energy is needed to sustain root length increases that do occur. All of these factors can result in plant stress, which may eventually result in reduced crop growth and productivity.

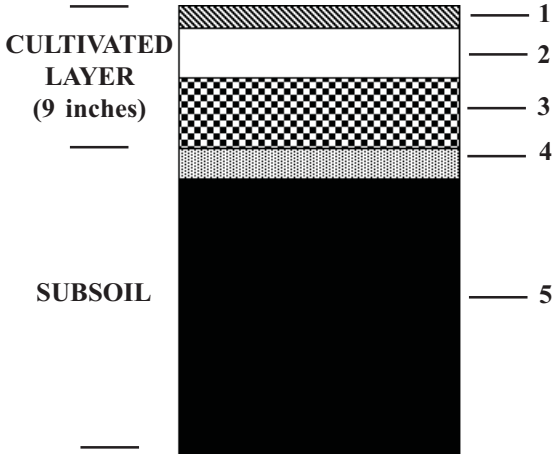
Penetration Resistance

Penetration resistance is a measure of the ease with which an object can be pushed into the soil (Bradford, 1986). It gives an indication of root-impeding layers in the soil and can be used in comparing relative strengths among similar soil types. It can also be used for determining hardpans, zones of compaction, or dense soil layers.

Interpretations

Soil compaction that results in severely restricted root growth is caused mainly by trampling of animals, use of farm and tillage equipment, and vehicular traffic. The type of root system will

determine the ability of a root to penetrate the soil. Figure 4 shows typical locations of compaction zones in cultivated soils.



Zone 1: Surface crusting, which may impede seedling emergence and water infiltration.
Zone 2: Low impedance zone for roots; loosened by tillage.
Zone 3: Plowed or deeply loosened cultivated soil that has been recompacted by vehicular traffic.
Zone 4: Subsoil compaction by wheel traffic and tillage implement-soil interactions during tillage.
Zone 5: May contain high mechanical impedance due to inherent factors, such as duripans, fragipans, ortstein layers, petrocalcic layers, etc., which may occur near the surface if topsoil is not present.

Figure 4. General position of soil compaction zones in cultivated systems (Bennie, 1996)

Penetration resistance depends strongly on the soil water content: the dryer the soil, the greater the resistance to penetration. Therefore, the water content of the soil should be noted when taking a measurement. Penetration resistance is best determined when the soil is at field capacity, which is a uniform condition that can be reproduced from season to season.

Soil Structure

Soil structure is the arrangement and organization of particles in the soil. It is strongly affected by changes in climate, biological activity, and soil management practices. Soil structure affects the retention and transmission of water and air in the soil as well as the mechanical properties of the soil. Observing and describing soil structure in the field is subjective and qualitative.

Interpretations

For plant growth it is desirable to have a physical condition in which the soil is an optimally loose, friable, and porous assemblage of aggregates permitting free movement of water and air, easy cultivation and planting, and unobstructed germination and root growth (Hillel, 1982). The soil structure index is a general quality placement that indicates the closeness to the condition described above. In general, the higher the index value the better the soil's capacity to transmit water and air and to promote root growth and development.

Soil processes involved in the development of soil structure are as follows (Rowell, 1994):

- drying and wetting, which cause shrinking and swelling, creating cracks and channels;
- freezing and thawing, which creates spaces as ice is formed;
- the action of roots (removal of water, release of exudates (organic materials), and formation of root channels);
- the action of soil animals (moving soil material around, creating burrows, and bringing soil

mineral and organic materials into close association); and

- the action of microorganisms (breaking down plant and animal residues and creating soil organic matter and humus as a binding material).

Soil Texture

Soil texture refers to the distribution of sand, silt, and clay sized mineral particles in the soil. Texture is one of the most stable attributes of the soil, being modified only slightly by cultivation and other practices that cause mixing of the different soil layers.

Interpretations

This test is routinely used by soil scientists and provides reliable estimates of soil texture. The textural class places the soil in one area of the triangular diagram based on the distribution of sand, silt and clay in the soil (Figure 5). Texture is an important characteristic, because it influences fertility and helps determine water intake rates, water storage in the soil, ease of tillage, and amounts of aeration. For example, clay soils will retain more water and nutrients than a sandy soil.

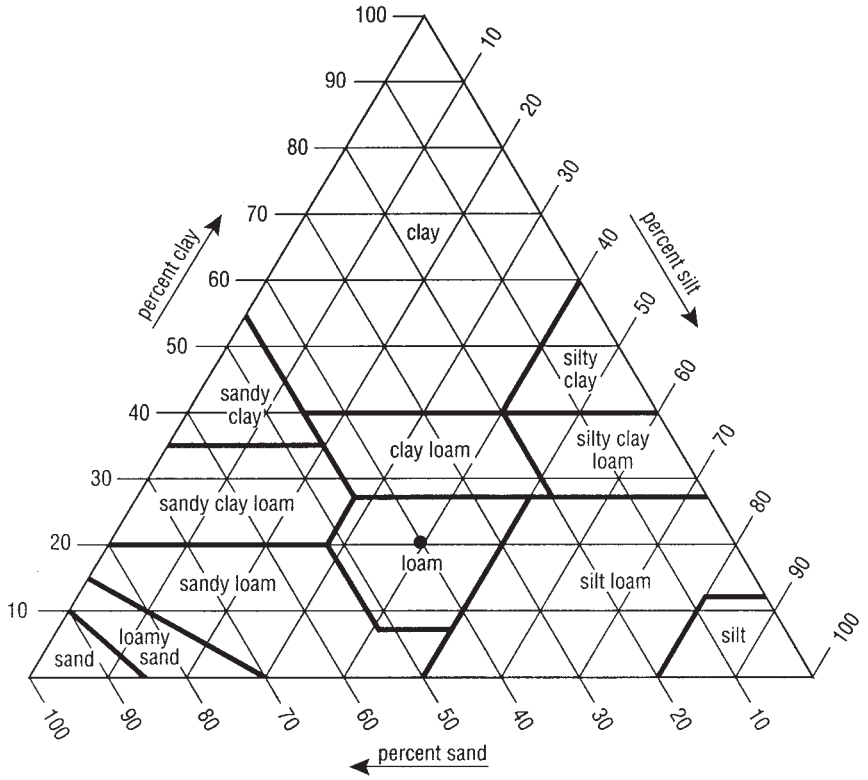


Figure 5. Soil textural triangle showing the percentages of clay, silt, and sand in the textural classes.

Mineral Particles Soil is composed of mineral particles that vary in size. There are three general classifications (or soil separates) of mineral particles:

- *sand* particles - 2.0 mm (very coarse) to .05 mm (very fine);
- *silt* particles - .05 mm to .002 mm;
- *clay* particles - smaller than .002 mm.

Twelve Soil Textural Classes. Definitions of the 12 textural classes are based on the relative proportion, or weight, of these three particle classifications. Sandy soil, for example, has a greater proportion of sand particles than silt or clay. In reading the textural triangle (Figure 5), any two particle size percentages will locate the textural class. For example, a soil containing 20% clay and 40% sand is located in the *loam* textural class (Figure 5).

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11. Water Quality

The quality of water is relative to the purpose for which the water is used; therefore, specific use conditions will determine the suitability of a water body (James et al., 1982).

Water Electrical Conductivity

Introduction

Water salinity levels, as measured by electrical conductivity, can be used to assess irrigation water quality. Other water quality concerns about saline waters include possible physiological effects on humans and animals and mineral taste. Also, high concentrations of certain mineral salts can cause corrosion damage in water systems. The measurement of electrical conductivity is an indicator of the total dissolved solids (TDS) in water. The relationship of EC to TDS will vary depending upon the distribution of major constituent elements present in the water.

Interpretations

Tables 9 and 10 contain safe salinity limits for human and livestock drinking water. Table 11 contains general guidelines for salinity in irrigation waters. For aquatic plant growth, salinity levels should be kept as close to natural conditions as possible (US EPA, 1973).

Table 9. Safe limits for drinking water (US EPA) and average salinity levels for river waters of the world (James et al., 1982)

Water	EC[2] (dS/m 25°C)	Total Dissolved Solids (mg/L)
Drinking water		
SMCL[1]	0.78	500
Livestock and poultry		
US EPA recommendation	4.7	3000
Average salinity levels in river waters		
North America	0.23	146
Europe	0.28	182
Australia	0.09	59
World	0.19	120

[1] SMCL = secondary maximum contaminant levels are unenforceable Federal guidelines regarding taste, odor, color, and other non-aesthetic effects in drinking water.
[2] EC estimated from total dissolved solids (TDS); EC = TDS/640

Table 10. Use of saline waters for livestock and poultry (US EPA, 1973)		
Comment	EC[1] (dS/m 25°C)	Total Dissolved Solids (mg/L)
Relatively low level of salinity. Excellent for all classes of livestock and poultry.	< 1.6	< 1,000
Very satisfactory for all classes of livestock and poultry. May cause temporary and mild diarrhea in livestock not accustomed to them or watery droppings in poultry.	1.6 - 4.7	1,000 - 3,000
Satisfactory for livestock, but may cause temporary diarrhea or be refused first by animals not accustomed to them. Poor waters for poultry, often causing watery feces, increased mortality, and decreased growth, especially in turkeys.	4.7 - 7.8	3,000 - 5,000
Can be used with reasonable safety for dairy and beef cattle, for sheep, swine, and horses. Avoid use for pregnant or lactating animals. Unfit for poultry and probably for swine.	7.8 - 10.9	5,000 - 7,000
Considerable risk in using for pregnant or lactating cows, horses, or sheep, or the young of these animals. In general, use should be avoided although older ruminants, horses, poultry, and swine may subsist on them under certain conditions.	10.9 - 15.6	7,000 - 10,000
Risks are too great and are not recommended for use under any conditions.	> 15.6	> 10,000

[1] EC estimated from total dissolved solids (TDS); EC = TDS/640

Table 11. General purpose guidelines for salinity in irrigation water for arid and semi-arid regions (US EPA, 1973).		
Classification	EC dS/m	TDS mg/L
Water for which no effects are usually noticed	0.75	500
Water that can have detrimental effects on sensitive crops	0.75-1.50	500-1,000
Water that can have adverse effects on many crops: requires careful management	1.50-3.00	1,000-2,000
Water that can be used for tolerant plants on permeable soils with careful management	3.0-7.50	2,000-5,000

Water Nitrate and Nitrite Levels

Introduction

Nitrate in water is of concern in regards to human and animal health and to environmental quality of ground and surface waters. Nitrate in drinking water can cause methemoglobinemia ("blue baby syndrome") in infants under six months of age and can have toxic effects in livestock and poultry. The toxicity occurs with the conversion of nitrate to nitrite after it has been consumed. Nitrite has a more rapid and pronounced toxicity effect than nitrate in drinking water. Fortunately, nitrite concentrations in water sources are usually very low. Nitrate in surface waters can cause accelerated growth of algae and aquatic plants, causing depletion of dissolved oxygen and general degradation of the water body (eutrophication). Eutrophication jeopardizes the use of water for recreation, sport and commercial fishing, agriculture, industry, and municipal supply. Also, nitrates can adversely impact aquatic ecosystems. Nitrate entering surface and groundwater is from non-point sources; both urban and agricultural runoff and leachate are recognized as contributors.

Interpretations

The levels of N required to induce eutrophication will vary depending on the nitrogen to phosphorous ratio. Excesses of either or both N and P can lead to eutrophication. Excessive growth of algae has been shown to occur when total phosphorus (mostly phosphate) levels exceed 0.10 ppm. Table 12 shows commonly used values for total nitrogen (mostly nitrate or ammonia). Eutrophication is defined as an increase in the nutrient status of natural waters that causes accelerated growth of algae or water plants, depletion of dissolved oxygen, increased turbidity, and general degradation of water quality (Pierzynski et al., 1994).

Table 12. Human, animal, and environmental limits of nitrate and nitrite in water (Pierzynski et al., 1994; US EPA, 1973).

Description	Limit or threshold
US EPA maximum contaminant level for nitrate-N in public drinking water	10 mg NO_3-N L^{-1}
US EPA maximum contaminant level for nitrite-N in public drinking water	1 mg NO_2-N L^{-1}
Recommended safe level for livestock and poultry drinking water	40 mg NO_3-N + NO_2-N L^{-1}
Recommended safe level for Nitrite-N alone in livestock and poultry drinking water	10 mg NO_2-N L^{-1}
Threshold for eutrophication in fresh water environments	0.5-1.0 mg N L^{-1}
Threshold for eutrophication in marine environments	> 0.6 mg N L^{-1}

Comments

The nitrate/nitrite test strips can determine both nitrate and nitrite concentrations (two test pads on each test strip). The nitrate test pad on the test strip measures the sum of both nitrate-N and nitrite-N present in the sample. If nitrite is detected in the sample, the amount can be subtracted from the nitrate reading to get the actual amount of nitrate-N in the sample. However, nitrite is rarely found in drinking waters at levels above 0.1 mg L^{-1} (Manahan, 1993).

1 ppm (parts per million) = 1 mg L^{-1} (milligram per liter)

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