Basic Soil Health Test Kit Manual

Rodale Institute

Research Department

2016

Contents

Basic Soil Health Kit Contents:

1. Testing manual
2. Four plastic mini-beakers
3. Four perforated sample cups with green lids
4. Two divided petri dishes
5. One 15 ml test tube
6. One 50 ml test tube
7. Seven pieces of filter paper
8. Seven pH test strips
9. Seven nitrate test strips
10. Seven phosphate test strips
11. Seven sulfate test strips
12. One mini-sieve/strainer
13. One spray bottle
14. One wooden stir stick

Basic Soil Health Kit Tests and Information:

This kit contains enough supplies to conduct seven basic soil tests (measuring pH, nitrates, phosphates, sulfates, water infiltration, water holding capacity, and aggregate stability). The pH, nitrate, phosphate, and sulfate tests may be done seven times, because each test strip and filter paper may be used only once, but the rest of the supplies may be washed in mild soap, rinsed thoroughly, and reused. The supplies used for the water infiltration and water holding capacity tests may be reused multiple times, but if damaged, additional supplies will be available in packs. These and more test strips will be available at the Rodale Institute online store. An optional test to observe soil microbial communities using your own supplies is also available.

To receive periodic updates, including additional test procedures, please email Kristine.nichols@rodaleinstitute.org with Soil Test Kit in the subject line.

Disclaimer

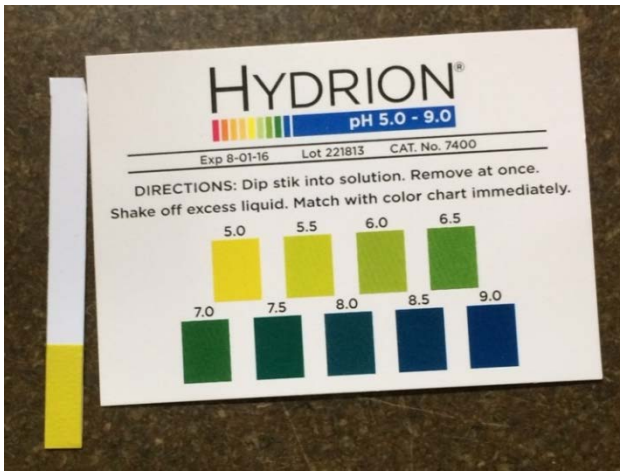
Measuring Soil pH, Nitrates, and Phosphates

Materials:

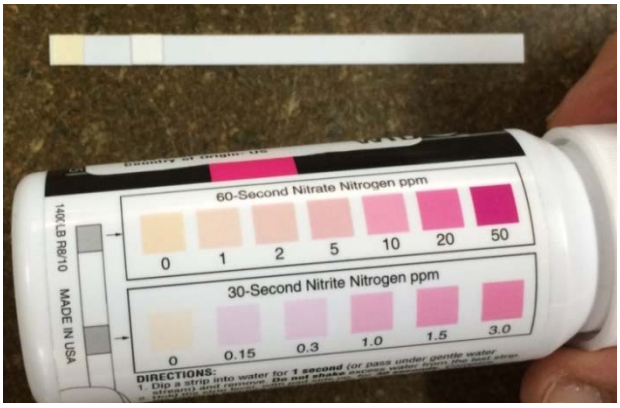
50 ml test tube
Sample cup
Wooden stir stick
Filter paper
pH test strips
Nitrate test strips
Phosphate test strips
Sulfate test strips
Timer/stopwatch (not included)

Method:

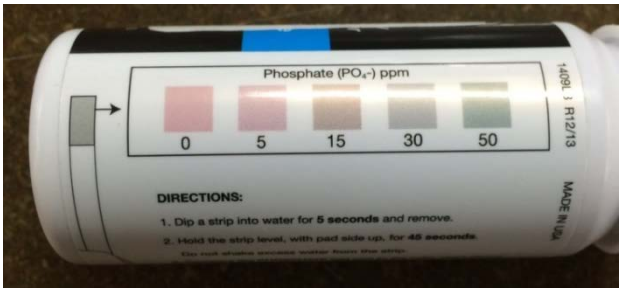
1. Using the 50 ml test tube, collect a 30 ml soil sample and transfer into the sample cup. Add 30 ml of tap water to the sample cup. (Note: If using compost or soil with high organic matter content, you will need to use twice as much water as soil – 30 ml of soil and 60 ml of water.)
2. Stir with wooden stir stick until thoroughly mixed.
3. Fold filter paper into quarters, then open it to look like a cone, and place it in sample cup. Water will move out of the soil into the filter paper cone so a water sample without soil debris may be tested as below.
4. Dip a pH test strip (strip has single yellow pad) in the water in the filter paper cone and remove immediately, shake off excess liquid, and compare to the color chart on the following page. If the pad is not yellow, then it has expired and do not use.
5. Dip nitrate test strip (strip has two pads both either white or pinkish) in the water in the filter paper cone for 1 second, shake off excess liquid, and after 1 minute compare with color chart on the following page. If pads have developed a color that is not zero, then they have expired and do not use.
6. Dip phosphate test strip (strip has single pinkish pad) in the water in the filter paper cone for 5 seconds, shake off excess liquid, and after 45 seconds with color chart on the following page. If pad has developed a color that is not zero, then they have expired and do not use.
7. Dip sulfate test strip (strip has four pads) in the water in the filter paper cone for 1 second, shake off excess liquid, and after 2 minutes compare with color chart on the following page. If pads have developed a color that is not zero, then they have expired and do not use.
8. Test strips and filter paper cannot be reused, but sample cup, stir stick and test tube may be washed with mild soap, rinsed thoroughly with water and reused.



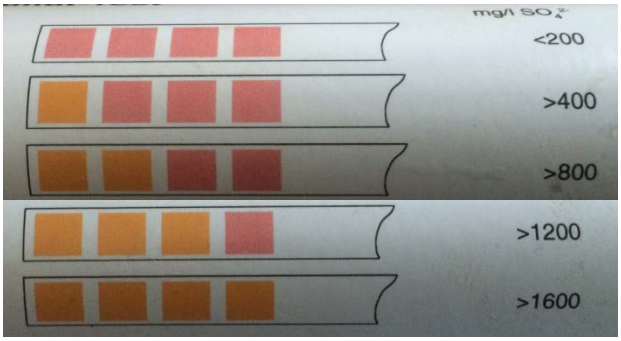
pH Test Strip Color Chart – After dipping the strip in the filtered water, compare the color of the pad to the color on the chart immediately.



Nitrate Test Strip Color Chart – After dipping the strip in the filtered water for 1 second, shaking off the excess liquid and letting it rest for 1 minute, compare the color of the pad to the color on the chart. Only look at color on bottom pad for nitrate comparison. Soils will not have high enough nitrite levels to be measured.



Phosphate Test Strip Color Chart – After dipping the strip in the filtered water for 1 second, shaking off excess liquid and letting it rest for 1 minute, compare the color of the pad to the color on the chart.



Sulfate Test Strip Color Chart – After dipping the strip in the filtered water for 1 second, shaking off excess liquid and letting it rest for 2 minutes, compare the color of the pad to the color on the chart.

Estimating Water Infiltration, Water Holding Capacity, and Soil Nitrate, Phosphate, and Sulfate Content

Materials:

50 ml test tube
15 ml test tube
Mini-beaker
Perforated sample cup
pH test strips
Nitrate test strips
Phosphate test strips
Sulfate test strips
Timer/stopwatch (not included)

Method:

1. Place 50 ml of soil in perforated sample cup, gently break up any large clods and shake to distribute evenly. Nest perforated sample cup inside the mini-beaker.
2. Pour 50 ml of tap water over the soil in the perforated cup. This is equal to about 2.54 cm or 1 inch rainfall.
3. Using a timer/stopwatch, start timing how long it takes for the water from when you have finished pouring it to completely infiltrate into the soil. Stop and record when complete (i.e. when the surface of the soil is just glistening).
4. Collect and examine for debris the water in the mini-beaker, and transfer to 15 or 50 ml, as appropriate, test tube to measure volume.
5. Dip a pH test strip (strip has single yellow pad) the water in the test tube and remove immediately, shake off excess liquid, and compare to the color chart on the above page. If the pad is not yellow, then it has expired and do not use.
6. Dip nitrate test strip (strip has two pads both either white or pinkish) in the water in the test tube for 1 second, shake off excess liquid, and after 1 minute compare with color chart on the above page. If pads have developed a color that is not zero, then they have expired and do not use.
7. Dip phosphate test strip (strip has single pinkish pad) in water in the test tube for 5 seconds, shake off excess liquid, and after 45 seconds compare with color chart on the above page. If pad has developed a color that is not zero, then they have expired and do not use.
8. Dip sulfate test strip (strip has four pads) in water in the test tube for 1 second, shake off excess liquid, and after 2 minutes compare with color chart on the above page. If pads have developed a color that is not zero, then they have expired and do not use.
9. The nitrate, phosphate, and sulfate tests will give you an indication of how much nitrate, phosphate, and sulfate may runoff.
10. Test strips cannot be reused, but test tubes, mini beaker, and perforated sample cup may be washed with mild soap, rinsed thoroughly with water and reused.

Observing Aggregate Stability

Materials:
Mini-beaker
Petri dish, divided in half
Mini-sieve/strainer
Spray bottle
Wooden stir stick

Method:
1. Collect a 100 ml soil sample (a sample may be collected from around the plant roots if present) with the mini-beaker.
2. Place the soil on a newspaper or other paper to air-dry and leave at room temperature for 3-5 days depending on how wet the soil sample was and ambient humidity. (After drying, the sample can be stored in a plastic bag at room temperature until ready to use.)
3. While the sample is air-drying, gently massage the sample by hand to break up large clods.
4. Pass the soil through the mini-sieve by adding enough air-dried soil to form a thin (0.25 inch) layer on the bottom of the mini-sieve.
5. Shake the mini-sieve 40 times manually to pass soil particles smaller than the openings on the mini-sieve through the sieve.
6. Pour the aggregates on top of the screen in the mini-sieve onto a piece of newspaper or paper.
7. Repeat steps 4-6 until all the soil has been either collected on the screen or passed through the screen.
8. Using the wooden stir stick or a ½ teaspoon, transfer a small amount of aggregates that were collected on top of the screen into one side of the Petri dish.
9. Repeat with another sample if you wish and place in the other half of the Petri dish.
10. Place tap water into the spray bottle.
11. Spray water onto the inside edge of the Petri dish and allow it to soak into the aggregates.
12. Observe the amount of aggregates that remain intact.
13. All materials may be washed with mild soap, rinsed thoroughly with water and reused.

Optional Quantitative Method for Slaked Aggregate Stability

After step 7,
1. Weigh and record the weight of the mini-sieve using a kitchen scale.
2. Prepare a small bowl with enough tap water to submerge the sieve to the point where the water is near the top, but does not go over the side.
3. Transfer about a ½ teaspoon of the aggregates from the newspaper or paper back onto sieve and weigh and record the weight using a kitchen scale. Subtract the weight from step 2 from this weight to have the weight of the aggregates.
4. Gently place the sieve with the soil from step 2 into the bowl with water prepared in step 1.
5. Gently move the sieve up and down manually in the column of water – making sure not to move it up so high that the bottom of the sieve breaks the water surface. Repeat moving the sieve up and down 40 times in a two minute period.
6. Place the mini-sieve and soil on a weighed oven safe pan, such as a pie tin, and dry at 250^{o}F for 2 hours or until dry and weigh the sieve, soil, and pan. Subtract the weight of the sieve and pan (i.e. dry weight of the aggregates). Divide this weight by the weight from step 3 and multiple by 100 to get the percentage of water stable aggregates by slaking.

Optional Activity - Winogradsky Column

Introduction

A Winogradsky column is used to illustrate how microorganisms develop in certain habitats in response to environmental conditions and how populations exhibit succession i.e., how one group of organisms succeeds another over time as conditions change. These columns are complete, self-contained recycling systems, driven only by energy from sunlight! Invented by Sergei Winogradsky, this method uses a deep, clear cylindrical vessel with an air-tight lid, samples (soil, roots, leaves, or other organic matter), and water. The vessel incubates under natural light or a growth lamp for several months with observations being made on a weekly basis.

All the organisms are present initially in low numbers, but when the tubes are incubated for 2 to 3 months, different types of microorganisms proliferate and occupy distinct zones where environmental conditions favor their specific activities. The first organisms are photosynthetic algae and bacteria and populations using plant materials, carbohydrates, lignins, etc., for growth under aerobic conditions and in so doing, creates an anaerobic environment especially at the bottom. Under these anaerobic conditions, sulfate-reducing organisms multiply using organic carbon compounds and making sulfuric acid (i.e. H_2S). Such an environment now allows the development of photosynthetic bacteria requiring reduced sulfur compounds, anaerobiosis and light. Consequently, one finds purple sulfur bacteria and green sulfur bacteria growing deep in the vessel while at the same time it is possible to find blue-green algae growing in the surface water since they are not strict anaerobes. The photosynthetic organisms (i.e. algae, bacteria and cyanobacteria) tend to cluster at different levels since they are motile and may swim to where concentrations of nutrients and degree of illumination are most appropriate for their growth - an example of both chemotactic and phototactic response.

The column provides numerous gradients, depending on additive nutrients – types and amounts of organic matter or addition of synthetic nutrients – from which the variety of organisms may grow. The aerobic water phase and anaerobic soil phase are one such distinction. Due to low oxygen solubility in water, the water quickly becomes anoxic towards the interface of soil and water. Anaerobic phototrophs are still present to a large extent in the soil phase, and there is still capacity for biofilm creation and colony expansion. Algae and other aerobic phototrophs are present along the surface and water of the upper half of the columns. Green growth is often attributed to these organisms.

Method

1. Fill a clear glass or plastic cylinder or a bottle about 2/3 full with soil from different sites or adding differing amounts of residue.
2. The sample may be supplemented with ~0.25% w/w calcium carbonate and ~0.50% w/w calcium sulfate or sodium sulfate, shredded newspaper or hay (for cellulose), ground egg-shell and/or egg yolk.
3. Rain or tap water is added until the soil is saturated and about half the remaining volume is filled.

4. Cap or seal the container and let it sit for several months in natural light or under a growth lamp.
5. Observe growth of organisms and formation of biofilms. See Figs. 1 and 2.
6. Record the width the layers and changes in color periodically (i.e. every 1-2 weeks).
7. Take a sample from different layers using the transfer pipet and place on microscope slides, cover with cover slip, and observe under the microscope. Record microscopic observations – organism type, shape, size, number, or other distinguishing characteristics – on ten fields of view.

Figure 1. Winogradsky columns made in either clear cylinders (A) or bottles (B) exhibit changes in color as different organisms grow due to changes in the oxygen concentration or carbon sources.

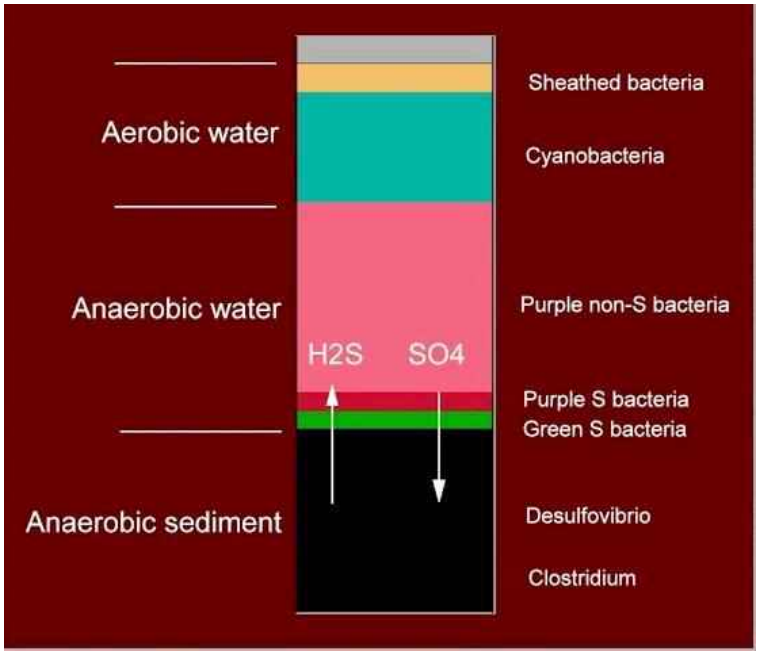


Figure 2. In a Winogradsky column, microbial habitats change as oxygen and carbon sources change. These changes are exhibited in the formation of different colored layers as illustrated above.

Websites

1. Solvita® Soil Respiration Test - https://solvita.com/soil
2. Soil Quality Test Kit Guide - http://soils.usda.gov/sqi/assessment/test_kit.html
3. Sponges for Soil Properties – http://www.apsru.gov.au/apsru/Projects/wfs/ActionLearning/Tools_Book/SpongSoilProp1.pdf
4. Soil Science Education Home Page - http://soil.gsfc.nasa.gov/index.html
5. Soil Education - http://soils.usda.gov/education/index.html
6. Agriculture is a Science - http://www.ars.usda.gov/is/kids/fair/story.htm
7. Building Better Soil – Taste the Difference - http://www.conservationinformation.org/publications/BetterSoilRecipe.pdf
8. Hold That Soil - http://www.arlingtonecho.net/pdf_files/hld_soilgr2.PDF
9. Sponge and Bucket Demonstration - http://animalrangeextension.montana.edu/LoL/Module-2c/2-Demonstration3.htm
10. Soil Erosion Demonstration - http://soils.usda.gov/education/resources/k_12/lessons/experiments/erosion/
11. How Soil Acts as a Water and Sponge - http://waterquality.montana.edu/docs/education/4h_manual_ch9a.shtml
12. Surface Area vs. Size and Shape - http://courses.soil.ncsu.edu/resources/physics/texture/soilgeo.swf
13. Understanding Science – http://undsci.berkeley.edu/

Recorded Presentations

1. Soil Health Demonstration at COP21, https://www.youtube.com/watch?v=PHGfryub2SQ
2. Climate Change and Organic Agriculture, http://www.conservationwebinars.net/webinars/climate-change-and-organic-agriculture/?searchterm=None
3. Soil Biology and Plants as Weed Management Tools, Delta Western Slope Soil Health Conference, https://www.youtube.com/watch?v=mcdeZcA8ctI
4. Soil is the Heart of the System, https://www.youtube.com/watch?v=14fDrB8n08E
5. Sustainable Intensification by Managing Microbial Communities and Processes in Agroecosystems. *Monday, November 3, 2014: 12:05 PM, Long Beach, CA* https://scisoc.confex.com/scisoc/2014am/webprogram/Paper86997.html
6. Cover Cropping Impacts on Arbuscular Mycorrhizal Fungi and Soil Aggregation. *Tuesday, November 4, 2014: 8:00 AM* https://scisoc.confex.com/scisoc/2014am/webprogram/Paper86852.html
7. Soil Health and Organic Agriculture, http://www.conservationwebinars.net/webinars/organic-farming-and-soil-health
8. White paper press conference at Rodale Inc. https://www.youtube.com/watch?v=e2ptCPcPJyI

Journal Articles

1. Brinton, W., R. Haney, E. Evans (2007) Simplified Approach to Measuring Soil CO2 Respiration: Comparison of chemical titration, CO2 IRGA analysis and the Solvita Gels. Proceedings ASA-SSSA-CSA Annual Meetings, New Orleans.
2. Bruns, M.A. and L.B. Byrne. 2004. Scale model of a soil aggregate and associated organisms: A teaching tool for soil ecology. J. Nat. Resour. Life Sci. Educ. 33: 85-91.
3. Franzluebbers, A.J., R. Haney, C. Honeycutt, H. Schonberg and F. Hons (2000) Flush of Carbon Dioxide Following Rewetting of Dried Soil Relates to Active Organic Pools. Soil Sci. Soc. Am. J. 64:613–623 - Haney, R. et al (2001) A rapid procedure for prediction of N mineralization. Biol. Fertil. Soils, 33: 100-104.
4. Haney, R., W. Brinton (2008) Soil CO2 respiration: Comparison of chemical titration, CO2 IRGA analysis and the Solvita Gel System. Renewable Agriculture & Food Systems, 23:171-176.
5. Haney, R., W. Brinton (2008) Estimating Soil C, N, and P Mineralization from short-term CO2 respiration. Communications in Soil Science and Plant Analysis, 39: 2706-2720.
6. Haney, R. et al (2010) Simple and Rapid Laboratory Method for Rewetting Dry Soil for Incubations. Communications in Soil Science and Plant Analysis, 41:1493-1501.
7. Kemper, W.D. and W.S. Chepil. 1965. Size distribution of aggregates. In: Methods of soil analysis, Part I. Ed: C.A. Black. Agronomy No. 9. American Society of Agronomy 499-509.
8. Nichols, K.A., T. Caesar, and J. Halvorson. 2013. Roles of biology, chemistry, and physics in soil macroaggregate formation and stabilization. Open Agric. J. 7: 107-117. http://dx.doi.org/10.2174/1874331520131011003
9. Nichols, K.A., and S. Samson-Liebig. 2011. An inexpensive and simple method to demonstrate soil water and nutrient flow. J. Nat. Res. Life Sci. Ed. 40: 51-57. http://dx.doi.org/10.4195/jnrlse.2009.0033
10. Nichols, K.A. and M. Toro. 2011. A whole soil aggregate stability index (WSSI) for evaluating soil aggregation. Soil Till. Res. 111: 99-104. http://dx.doi.org/10.1016/j.still.2010.08.014
11. Nichols, K.A. and S.F. Wright. 2004. Contributions of soil fungi to organic matter in agricultural soils. p. 179-198. In: Functions and Management of Soil Organic Matter in Agroecosystems. F. Magdoff and R. Weil (Eds.). CRC Press.
12. Six, J., H. Bossuyt, S. Degryze, and K. Denef. 2004. A history of research on the link between (micro)aggregates, soil biota, and soil organic matter dynamics. Soil and Tillage Research 79: 7-31.
13. Six, J., E.T. Elliott, and K. Paustian. 2000. Soil structure and soil organic matter. II. A normalized stability index and the effect of mineralogy. Soil Science Society of America Journal 64(3): 1042-1049.
14. Wright, S.F. 2000. A fluorescent antibody assay for hyphae and glomalin from arbuscular mycorrhizal fungi. Plant and Soil 226: 171-177.
15. Wright, S.F., M. Franke-Snyder, J.B. Morton, and A. Upadhyaya. 1996. Time-course study and partial characterization of a protein on hyphae of arbuscular mycorrhizal fungi during active colonization of roots. Plant and Soil 181: 193-203.
16. Wright, S.F., J.L. Starr, and I.C. Paltineanu. 1999. Changes in aggregate stability and concentration of glomalin during tillage management transition. Soil Science Society of America Journal 63: 1825-1829.

17. Wright, S.F. and A. Upadhyaya. 1996. Extraction of an abundant and unusual protein from soil and comparison with hyphal protein of arbuscular mycorrhizal fungi. Soil Science 161: 575-585.
18. Wright, S.F., and A. Upadhyaya. 1998. A survey of soils for aggregate stability and glomalin, a glycoproteins produced by hyphae of arbuscular mycorrhizal fungi. Plant and Soil 198: 97-107.
19. Wright, S.F., and A. Upadhyaya. 1999. Quantification of arbuscular mycorrhizal fungi activity by the glomalin concentration on hyphal traps. Mycorrhiza 8: 283-285.