Reducing Stormwater Runoff Volumes with Biochar Addition to Highway Soils

FINAL REPORT
December 2019

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In cooperation with

Rutgers, The State University of New Jersey
And
State of Delaware
Department of Transportation
And
U.S. Department of Transportation
Federal Highway Administration

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The Center for Advanced Infrastructure and Transportation (CAIT) is a National UTC Consortium led by Rutgers, The State University. Members of the consortium are the University of Delaware, Utah State University, Columbia University, New Jersey Institute of Technology, Princeton University, University of Texas at El Paso, Virginia Polytechnic Institute, and University of South Florida. The Center is funded by the U.S. Department of Transportation.

1. Report No.	2. Government Accession No.	3. Recipient's Catalog No.
CAIT-UTC-NC 44		

4. Title and Subtitle	5. Report Date
Reducing Stormwater Runoff Volumes with Biochar Addition to Highway Soils	December 2019
	6. Performing Organization Code CAIT/University of Delaware
7. Author(s) Paul T. Imhoff, Julia A. Maresca, Ali Nakhli, Christine Chapman	8. Performing Organization Report No. CAIT-UTC-NC 44
9. Performing Organization Name and Address University of Delaware Newark, DE 19716	10. Work Unit No.
	11. Contract or Grant No. DTRT13-G-UTC28
12. Sponsoring Agency Name and Address Center for Advanced Infrastructure and Transportation Rutgers, The State University of New Jersey 100 Brett Road Piscataway, NJ 08854	13. Type of Report and Period Covered Final Report 9/1/16 – 12/31/18
	14. Sponsoring Agency Code

15. Supplementary Notes
U.S. Department of Transportation/OST-R
1200 New Jersey Avenue, SE
Washington, DC 20590-0001

16. Abstract
Stormwater runoff from roadways is a major source of pollution, but current stormwater treatment technologies, such as bioretention cells, do not efficiently remove contaminants. New technologies are needed that can both remove more pollutants and reduce the volume of stormwater discharge. Such technologies will not only improve water quality but result in significant cost savings for state departments of transportation. Biochar, made by pyrolyzing biological material such as wood chips, may increase water infiltration when used as an amendment in stormwater bioretention cells. Here, we examined how biochar contributes to soil aggregation, which in turn improves water infiltration in soil. We show that biochar changes production of adhesive macromolecules (proteins and polysaccharides), and that particles in biochar-amended soil are, on average, larger in diameter than those in un-amended soil. These macroaggregates likely form on time scales of months to years, suggesting that biochar may be an amendment that increases in effectiveness over time.

17. Key Words	18. Distribution Statement
Stormwater, biochar, infiltration, soil aggregation	

19. Security Classification (of this report)	20. Security Classification (of this page)	21. No. of Pages	22. Price
Unclassified	Unclassified	Total #18	

Acknowledgments

The authors acknowledge the research efforts of MS student Christine Chapman, PhD student Seyyed Ali Akbar Nakhli, and undergraduate student Michael Rechsteiner. Supplemental funding for field measurements reported herein was provided by the NCHRP IDEA program.

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1. DESCRIPTION OF THE PROBLEM

Stormwater runoff from roadways is a major source of pollution. Existing stormwater treatment technologies, such as bioretention cells, are not always efficient at removing nitrogen, metals and organic contaminants, requiring large footprints for treatment. New technologies are needed that can both remove pollutants more effectively and reduce the volume of stormwater discharge. Such technologies will not only improve water quality but result in significant cost savings for state departments of transportation. Biochar, made by pyrolyzing biological material such as wood chips, may increase water infiltration when used as an amendment in stormwater bioretention cells. Here, we examined how biochar contributes to soil aggregation, which in turn improves water infiltration in soil.

2. APPROACH

This research tested the hypothesis that biochar addition to highway soils increases water infiltration, thus reducing stormwater runoff volume. We further hypothesized that biochar increases water infiltration through a multistep process: (1) in the presence of biochar, microbial populations are altered and produce more extracellular macromolecules that "glue" soil particles into aggregates, and (2) soil aggregates increase preferential water flow and thus water infiltration. To test these hypotheses, we quantified soil aggregation and adhesion-promoting biological molecules in biochar-amended or un-amended field samples and soil columns.

3. METHODOLOGY

3.1 FIELD SITE, SAMPLE COLLECTION, AND SAMPLE PREPARATION

All field experiments were conducted at a site in Middletown, DE near the intersection of Rt. 896 and Bethel Church Road (GPS coordinates 39°31'48.9"N 75°44'10.7"W). The soil here is a sandy loam (Nakhli *et al.*, 2019). In November 2015, two strips 6.1 m x 1.8 m x 0.3 m (length x width x depth) in the stormwater infiltration ditch on the northeast side of Rt. 896 were modified for these experiments (Fig. 1). One strip was tilled to a depth of 30 cm. Biochar was added to the other strip to a total of 4% by weight (~ 400 kg), and the strip was tilled to a depth of 30cm to mix the biochar with the soil. The biochar was Soil Reef™ biochar produced from Southern Yellow Pine wood by pyrolysis at 550°C for 10 minutes (The Biochar Company, Berwyn, PA), and was not further treated prior to application.

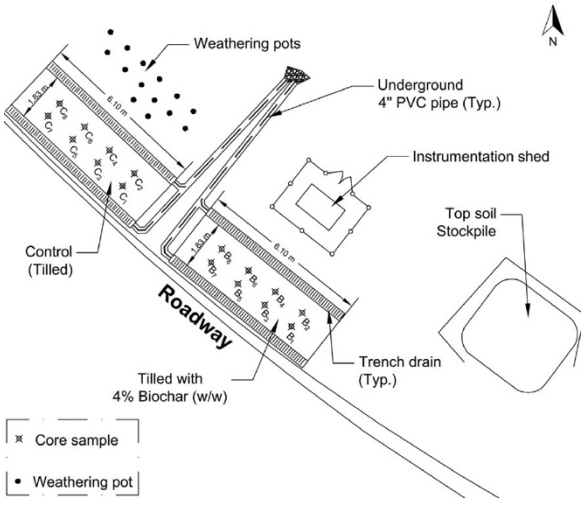

FIGURE 1. Schematic of field site. Two regions within a roadside stormwater filter were treated: one by tilling only ("control") and one by tilling with 4% biochar. Soil cores were removed where indicated.

In March 2017, eight cores (2.54 cm diameter x

30 cm deep) were taken from both the control (tilled-only) and biochar-amended areas of the Middletown field site. The cores were cut into six 5.1 cm sections which were immediately capped for storage. Core subsections from the same depth were pooled, generating six pooled samples for each area in the field site. The samples were air-dried, and invertebrates and plant material were removed using forceps.

In November 2018, cores (2.54 cm diameter) were taken at eight different locations from both the control and biochar-amended areas. At each sampling location, four cores were collected in depth increments of 0-7.5 cm, 7.5-15 cm, 15-22.5 cm, and 22.5-30 cm. Removing several short cores, rather than sectioning one long core, reduced sample compaction within the cores, which was noted during March 2017 sampling. All cores were immediately capped for storage. Cores at each depth in either the control or biochar-amended regions were pooled, and the particle size distribution and biological aggregation processes were assessed in each.

Pooled samples had an approximate mass of 250-350 g. Each pooled sample was passed through an 8-mm sieve to remove rock fragments. The remaining sample was kept in a sealed container for 2 days to allow the moisture content to equilibrate. Sub-samples (10-20 g) were removed to determine moisture content. The remaining soil in each sample was air-dried. Two 50 g subsamples from each air-dried soil mixture were removed and wet-sieved to quantity the water-stable aggregates at each sampling depth. Two 10 g subsamples of the air-dried, water-stable macroaggregates were used to quantify organo-mineral association at each sampling depth. Finally, 30-50 g subsamples were removed from each soil mixture, ground using a mortar and pestle to break apart all aggregates, and well mixed for determination of glomalin and polysaccharide content.

The soil used in all column microcosms (section 3.3) was also collected from the field site. Prior to packing the columns, soil was passed through a 2 mm diameter sieve, air dried, and gently ground using a mortar and pestle to remove existing aggregates.

3.2 Field data: Particle size assessment

3.2.1 Dry sieving

After air drying, the samples were passed through a set of nested sieves on a mechanical shaker for ten minutes. The sieves used were the following: size 4 (4.75 mm diameter), size 10 (2.0 mm diameter), size 20 (0.85 mm diameter), size 60 (0.25 mm diameter), size 140 (0.106 mm diameter), and size 200 (0.075 mm diameter). After shaking for ten minutes, the material on each sieve and in the sieve pan was weighed (Nimmo and Perkins, 2002). Material from each size fraction was then stored for future analysis.

3.2.2 Wet sieving

Samples were also wet-sieved to quantify the water stable aggregates. A modified protocol of Kemper and Rosenau 1986 was used for this project (Kemper and Rosenau, 1986). The air-dried sample (100g, March 2017 samples; 250 g, November 2018 samples) was placed on a 2 mm sieve and submerged in deionized water for 5 minutes. The sieve was then manually raised and lowered 50 times in two minutes at an amplitude of 3 cm. The material remaining on the sieve was gently backwashed into a pan and oven dried at 65°C. This process was repeated with the washthrough on 0.25mm and 0.053mm sieves. After oven drying, the material left on each sieve was weighed (Kemper and Rosenau, 1986). Biochar has a low bulk density and tends to float in water. To prevent loss of the floating particles, biochar that had floated during wet sieving was

carefully rinsed off the edges of the sieve after the sieve was removed from the water.

A sand correction was also performed on the wet sieved core samples, to distinguish between true soil aggregates and large sand or biochar particles (Denef *et al.*, 2001). Five grams of each oven dried, wet sieved size fraction was suspended in 0.5% sodium hexametaphosphate in 100 ml beakers on a shaker at 100 rpm for 18 hours to disperse all particles. After dispersion, the wet sieving process was repeated. The sand corrected values for the field samples were obtained using the following equation (Denef *et al.*, 2001):

$$sand\ corrected\ values = aggregate\ weight - \left(\left[\frac{sand\ weight}{5\ g}\right] * aggregate\ weight\right)$$

Equation (1)

The aggregate weight is the weight left on each sieve before the sand correction step. The sand weight is the weight left on each sieve after the sand correction step. A single sample was used to determine the water stable aggregates at each depth for March 2017, while two samples were used for November 2018.

3.3 Laboratory Soil Microcosms

A series of soil microcosms was prepared to examine the effects of time, biochar amendment, and water chemistry on soil aggregation. Polyvinyl chloride pipe with an interior diameter of 5.08 cm was cut to a height of 11.43 cm, producing tubes with an internal volume of 232 cm^3. The bottom of each tube was covered with cotton fabric to allow drainage. The tops were covered with perforated plastic cling wrap to reduce evaporative water loss. The top and bottom coverings were secured with rubber bands.

Water (10% v/v) was added to air-dried, gently ground soil. This slurry was then packed into columns (untreated microcosms). Biochar was added to this slurry to 2% or 4% (w/w) and mixed, then biochar-amended columns were packed. The biochar used was the same biochar used in the field site, and was not rinsed or sieved prior to adding it to the microcosms. Soil only (no biochar) that was autoclaved three times at 121°C for twenty minutes each time was used as an abiotic control.

Water, either deionized (DI) or artificial stormwater solution (ASW, composed of 860 ppm calcium sulfate, 2 ppm sodium nitrate, and 0.6 ppm disodium phosphate) was added to each soil column to establish a water content corresponding to approximately 70% saturation. Soil microcosms were incubated in the dark at 25°C for 16 weeks. They were weighed two to three times a week, the mass loss was recorded, and water (either DI or ASW) was added to maintain 70% of saturation. The microcosms were sampled destructively at 0, 1, 2, 4, 8, 12, and 16 weeks. There were two replicates of each condition.

The values for 70% of saturation were calculated by determining the air-filled porosity from the bulk and skeletal density of the soil and biochar (assuming a particle density of 2.65 g/cm^3 for the soil and a skeletal density of 1.051 g/cm^3 for the biochar (Nakhli *et al.*, 2019)), and then taking 70% of that value. First, the combined particle density is determined for each biochar and soil mixture using Equation (2):

$$particle\ density\ of\ mixture = \frac{100}{\left(\frac{percent\ soil}{soil\ particle\ density}\right) + \left(\frac{percent\ biochar}{biochar\ particle\ density}\right)}$$

Equation (2)

Then, the combined particle density of each biochar and soil mixture is used to determine the air-filled porosity of that mixture, which is complementary to the volume of water require to completely saturate the mixture. Equation (3) gives the volumetric water content needed to completely saturate the sample.

$$total\ porosity = 1 - \frac{bulk\ density\ of\ mix}{particle\ density\ of\ mix}$$

Equation (3)

At the designated timepoints, the microcosms were sacrificed, air dried, and dry sieved and stored according to the protocol used for the field samples. Samples of soil only and 4% biochar amendment at zero and 16 weeks were wet sieved according to the protocol used for the field samples.

3.4 Assessment of Processes Affecting Macroaggregation

Water-stable macroaggregates form in soil when organic matter and soil minerals are bound together (organo-mineral association); when glomalin, a fungal protein, is present; and/or if extracellular polysaccharides are produced by bacteria. To determine if one or more of these processes is enhanced with biochar amendment at the field site, an analysis for organo-mineral association was conducted on water-stable aggregates collected in November 2018. Glomalin and polysaccharides were also extracted and measured for the soil samples.

To assess organo-mineral association in macroaggregates, aggregate-density fractionation was used to separate free light particulate organic matter (POM), occluded light and mineral-associated POM, and minerals using a procedure developed from the literature (Six *et al.*, 1998; Rasmussen *et al.*, 2005; Brodowski *et al.*, 2006; Pronk *et al.*, 2012). To determine the free light POM fraction of the macroaggregates, a 10 g subsample of air-dried, water-stable macroaggregates (diameter > 250 µm) was suspended in 30 mL solution of 1.6 g/cm^3 sodium polytungstate in a 50-mL graduated conical centrifuge tube. This sample was reciprocally shaken gently by hand to suspend the particles (minimum 10 strokes). The material remaining on the lid and sides of the tube were washed into suspension with an additional 10 mL sodium polytungstate solution. After 20 minutes the sample was centrifuged at 3074 $\times$ *g* for 60 min. The supernatant, including floating material (free light POM), was removed, and the free light POM was collected on a 20 µm nylon filter by vacuum filtration, heavily rinsed in DI water to remove sodium polytungstate, air-dried and weighed. To quantify mineral-associated POM in the same subsample, the pellet was then resuspended in a 2.4 g/cm^3 sodium polytungstate solution, equilibrated, and centrifuged, and the material in the supernatant was quantified after filtration as above. Last, the remaining sediment in the tube was washed with DI water, air dried and weighted to determine the heavy mineral component within the macroaggregates. Two 10-g macroaggregate samples were analyzed at each measurement depth for the control and biochar-amended regions.

Glomalin and polysaccharide contents were measured for representative masses from the entire soil samples instead of selecting samples that consisted only of macrroaggregates, since during wet sieving these soil proteins may have been rinsed from the samples. Glomalin-related soil protein (GRSP) content was measured for a 1 g dry subsample from the 30-50 g of ground, well-

mixed sample at each measurement depth following previously developed procedures (Bedini *et al.*, 2009; Singh *et al.*, 2016; Kumar *et al.*, 2018). First, 8 mL of a 20 mM citrate solution (pH = 7.0) was added to the soil sample and the suspension autoclaved at 121 °C for 30 min. This suspension was centrifuged at 5000 g for 20 min and the supernatant stored at 4 °C until analysis. These steps were repeated four more times on the 1 g sample with 50 mM citrate (pH = 8.0) and autoclaving for 60 min after each extraction until the resulting supernatant had a straw-like color. The supernatant from the first extraction was used to determine easily extractable GRSP, while the supernatants from all extractions were mixed at appropriate volume ratios to obtain total GRSP. The protein content in the first extraction and in the solution produced from mixing all extractions was determined by Bradford assay (Thermo Fisher Scientific Co.) with bovine serum albumin as the standard. Two 1 g subsamples were analyzed in this manner at each measurement depth for the control and biochar-amended regions.

Total and labile soil polysaccharide content was also measured (Carter and Gregorich, 2007) . For total polysaccharide content, a 1 g dry subsample of mixed soil was extracted using 4.0 mL of 12 M H_2SO_4. After 2 h, the acid suspension was diluted to 0.5 M by adding DI water and then autoclaved for 1 h. The autoclaved sample was centrifuged to separate liquid and solid phases. Then, 1 mL of supernatant, 1 mL of 5% w/v phenol, and 5 mL of concentrated H_2SO_4 were added to a sample cuvette. After 10 min, the cuvette was placed in a water bath (25-30°C) for 25 min. Afterward, the absorbance at 490 nm was measured using a spectrophotometer. Calibration curves were created according to the same procedure using a known concentration of glucose and these used to determine the total soil polysaccharide content (glucose equivalent) of samples. Labile polysaccharide content was measured following steps similar to those outlined above for total polysaccharide content using a 1g dry subsample of the mixed soil, using 100 mL of 0.5 M H_2SO_4 rather than 4.0 mL of 12 M H_2SO_4. Two 1 g subsamples were analyzed at each measurement depth for the control and biochar-amended regions for measurement of total and labile soil polysaccharide content.

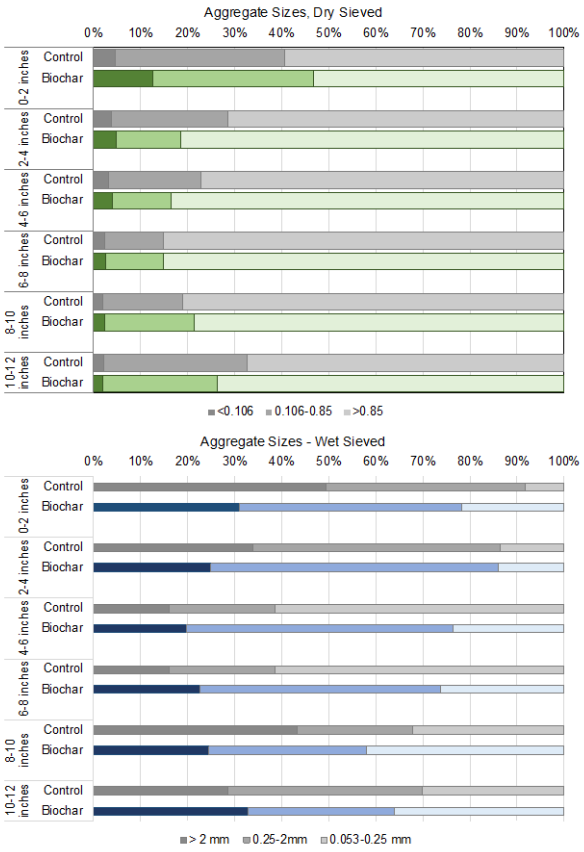

FIGURE 2. Aggregate sizes in the field samples collected in March 2018. A. Dry sieving. Samples were air-dried, then sieved, and the material retained on each sieve was weighed. In the surface sample, the biochar-amended samples had more fine particles, but at most other depths, these samples had more large particles than the control samples. **B. Wet sieving.** The biochar-amended samples have more water-stable aggregates in the two largest size classes than the control samples.

4. FINDINGS

4.1 PARTICLE SIZE ASSESSMENT

4.1.1 MARCH 2017 FIELD CORES

After collection of cores from the field site (Fig. 1), cores were sectioned and soil was air-died, then dry sieved to determine the size distribution of particles in each sample. At most depths, the biochar-amended section had a larger percentage of the sample in aggregates with diameter > 0.85 mm (Fig. 2A). The untreated control section has a larger percentage of the sample in microaggregates (diameter 0.106 mm to 0.85 mm). However, the difference between the untreated and biochar-treated samples was small.

Wet sieving was then used to determine the mass and size distribution of water-stable aggregates in the same samples (Fig. 2B). A correction was performed to correct for large particles such as gravel and large sand grains. The percent of mass in macroaggregates for the top 20 cm in the control section ranges from 13% to 42% (with a maximum at 4 to 8 inches), and 32% to 38% (with a maximum at 8 to 12 inches) for the biochar amended section. As depth increases, the mass of water-stable aggregates in the control samples decreases, while the mass of water-stable aggregates in the samples with biochar remains more constant. The mass of water-stable aggregates is similar for both samples at the shallowest and deepest depths, and is larger in the biochar-amended section than the control section at the intermediate depths. The differences between the dry and wet sieving suggest that wet sieving is more informative about aggregation in the field, where regular wetting and drying cycles occur.

4.1.2 NOVEMBER 2018 FIELD CORES

Soil aggregation is a dynamic process, and macroaggregation is expected to change seasonally and as soils age. For this reason, a second set of cores were collected in November 2018 for particle size analysis. Because the water-stable macroaggregates likely have the most influence on the presence of soil macropores and increased rates of stormwater infiltration, only water stable aggregates were reported for these samples.

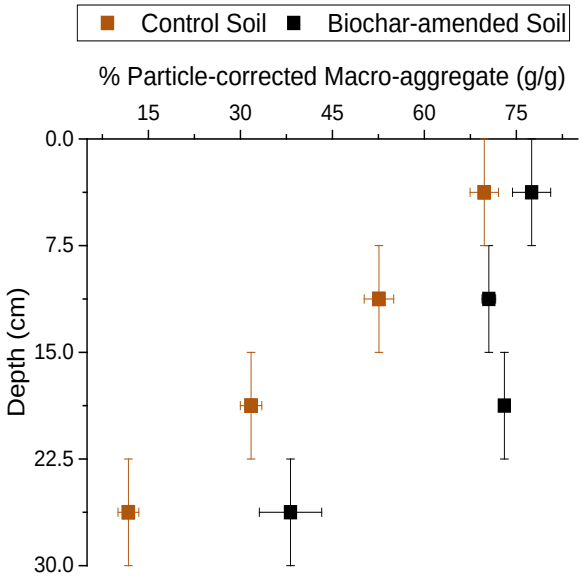

FIGURE 3. Water-stable macroaggregates (> 250 µm) in core samples collected in November 2018. Vertical error bars indicate measurement depth, while horizontal error bars are +/- one standard error.

Water stable aggregates are plotted versus depth (Fig. 3). At all depths the biochar-amended samples had a greater mass of water-stable macroaggregates than the control section. While data trends observed in November 2018 are similar to those for March 2017, at all depths there are more water-stable macroaggregates in 2018 than 2017. This illustrates the time-dependent nature of soil aggregation: macroaggregates form and break apart because of changing soil conditions. While at the

shallowest depth there were a greater number of water-stable macroaggregates in the control versus the biochar-amended section in 2017, in 2018 at all depths there were more macroaggregates in biochar-amended versus control regions.

These results suggest that biochar stabilizes soil aggregates under wet conditions. To investigate processes that might contribute to aggregate formation, we then used soil from this field site in a series of laboratory incubations to monitor formation and stability of aggregates in the presence and absence of biochar.

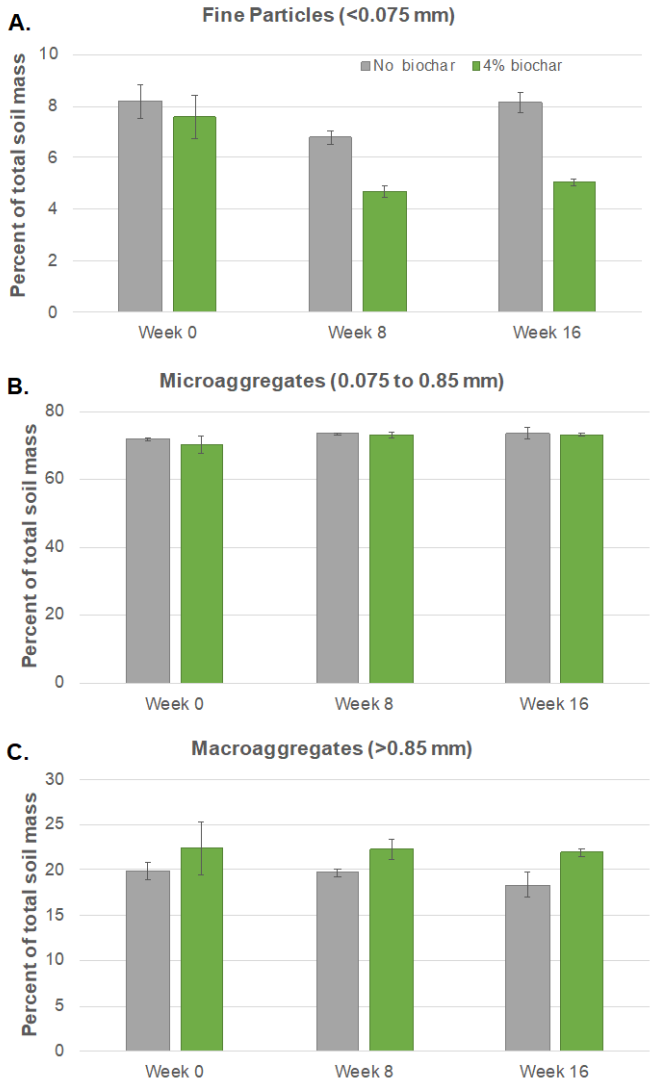

FIGURE 4. Particle size distribution in soil microcosms. Small pots of soil were maintained at a constant water content for 16 weeks. Samples were collected each week and size distribution analyzed by dry sieving. Bars indicate average percent of total soil mass in the specified size fraction and are averages of 4 samples; error bars indicate 1 standard deviation. (A) Untreated soil has more fine particles (<0.025 ⍰m) than soil amended with 4% biochar. The mass of the fine particles in biochar-amended soil decreases within 8 weeks. (B) Untreated soil has more microaggregates (particles between 0.025 and 0.85 ⍰m), but mass in microaggregates does not change significantly in either soil type over the course of 16 weeks. (C) Biochar-amended soil consistently has slightly more mass in macroaggregates than untreated soil.

4.2 Soil Microcosms

4.2.1 Average Particle Size and Macroaggregate Mass in Microcosms

Soil microcosms with no biochar, 2% biochar, and 4% biochar were prepared and incubated for 0-16 weeks to determine the amount and size distribution of soil aggregates in soil amended with different amounts of biochar and either DI water or artificial stormwater (ASW). Particle size distribution was measured using dry sieving, as described above. The particle size distributions in both un-amended and biochar-amended microcosms treated with DI water and ASW were indistinguishable, so these microcosms were analyzed as replicates.

The 4% biochar amended samples have more mass in the macroaggregate fraction than the other samples, ranging from 22% to 23% (Fig. 4A). The 0% biochar amended samples have the most mass in the fine fraction (Fig. 4C), likely because the soil particles are smaller than the biochar particles at the beginning of the experiment. The samples without biochar and the samples with a 2% biochar amendment showed similar trends in mass in macroaggregates, with no significant change in percent of mass in macroaggregates over time, and both have a lower percent of mass in macroaggregates than the samples amended with 4% biochar. The dry sieving data from the first set of microcosm experiments suggests that although particle size distribution varies until the soil moisture content reaches equilibrium,

there was no significant change in soil aggregation over a 16-week period.

Data from the particle size analysis of the field soil and biochar were used to predict the particle size distribution of the soil and 4% biochar-amended soil if there were no aggregation. In this case, 10% of soil without biochar amendment would be in macroaggregates (diameter > 0.25 mm), while ~11% of soil amended with 4% biochar would be in this category. However, the measured particle size distribution in both untreated and biochar-amended soils is larger than would be expected if aggregation were not occurring, indicating that some process holds soil or soil and biochar particles together. In the untreated soil, there is no difference between week 0 and week 16 (Fig. 5A), meaning that aggregation does not change over a 16-week time frame. Similarly, in the biochar-treated soil, the particles are larger than would be predicted if there were no aggregation. In this soil, there are more macroaggregates after 16 weeks than at 0 weeks, but the difference is minimal (Fig. 5B).

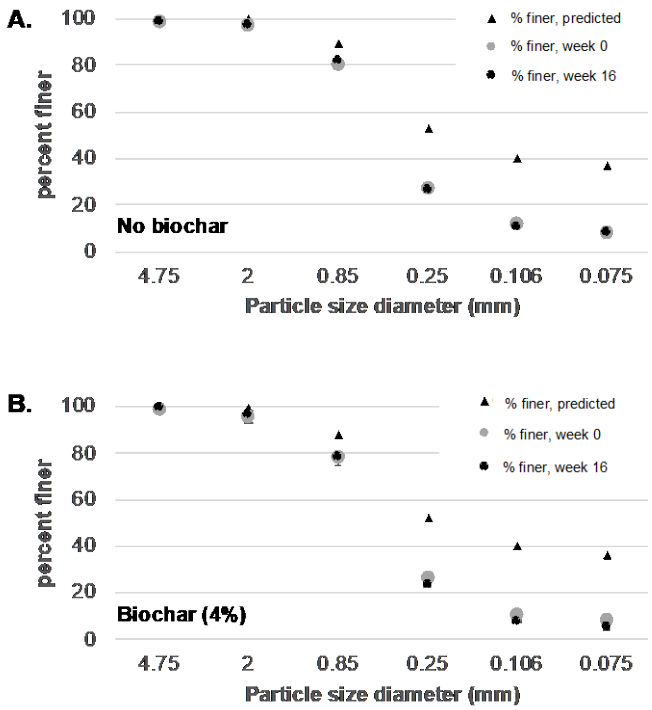

FIGURE 5. Predicted particle size distributions in soil alone and biochar-amended soil. A. Particle sizes of aggregates from untreated soil, plotted as "percent finer," or percent of soil mass that passes through a sieve of the stated size. B. Particle sizes of aggregates from biochar-amended soil (4% biochar by mass), plotted as "percent finer," or percent of soil mass that passes through a sieve of the stated size. In both cases, observed particle sizes were consistently larger than predicted.

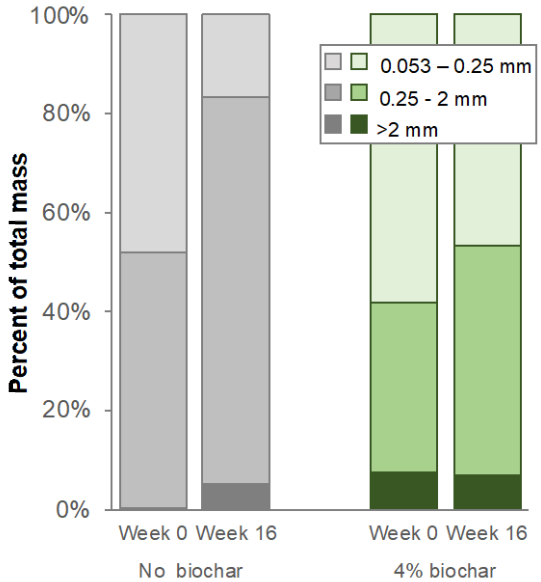

FIGURE 6. Water-stable aggregates in microcosms. After dry sieving, samples were wet-sieved and masses of particles of each size class were determined. Correction for sand content was calculated as described in section 3.2.2. The mass in the microaggregate fraction increased in both biochar-amended and untreated soils over 16 weeks.

4.2.2 Water-stable aggregates

Samples from Weeks 0 and 16 were wet sieved to quantify changes in water-stable aggregates over time. The percent of water-stable macroaggregates larger than 2 mm in diameter in the unamended soil increases over the course of 16 weeks (Fig. 6). Approximately 7% of the mass in the biochar-amended soil in macroaggregates > 2mm, and this fraction did not change over the duration of the experiment. In both treatments, the percentage of mass in the 0.25 mm to 2 mm size fraction increases. The percentage of water-stable microaggregates (0.053 mm to 0.25 mm) decreases over this time period. Broadly speaking, aggregation is occurring in both soils, but macroaggregate formation, especially in biochar-treated soil, may require more time.

4.3 Assessment of Processes Affecting Macroaggregation

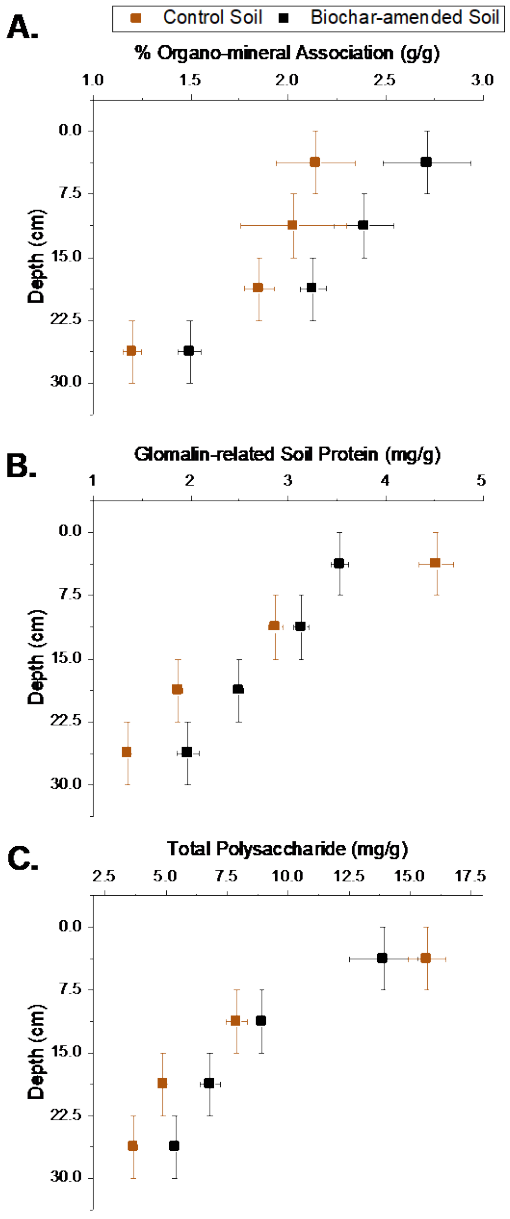

FIGURE 7. Aggregation mechanisms in soil. (A) Organo-mineral associations. Biochar-treated soil has a greater percentage of the mass in organo-mineral associations at all depths. (B) There is more glomalin-related soil protein in biochar-amended soil than in unamended soil at all depths except the shallowest. (C) Total polysaccharide concentration is higher in biochar-amended soil than in unamended soil at all depths except the shallowest.

Macroaggregates from the November 2018 field samples were analyzed to determine if organo-mineral association, glomalin, or polysaccharides could explain the difference in macroaggregation between biochar-amended and control soils. Results of density fraction measurements indicate that at all depths biochar amendment resulted in greater organo-mineral association (Fig. 7A). Thus, the addition of biochar increased the association between light organic matter, which includes biochar particles, with the denser mineral fraction.

Glomalin is a protein produced by soil fungi and has been shown to glue microaggregates into macroaggregates in many soils where fungal activity is important. Glomalin was measured in soil samples and except for the shallowest depth was greater in biochar-amended than the control soil (Fig. 7B). The difference in glomalin was most significant, though, at the shallowest depth where it was approximately 30% greater in the control versus biochar-amended soil. Thus, while biochar amendment was correlated with greater glomalin at depths exceeding 7.5 cm, at the top surface biochar did not enhance this soil protein. Because macroaggregation was more significant in biochar-amended soil at all depths (Fig. 3), the role of glomalin in enhancing macroaggregation in biochar-amended media is unclear.

Soil bacteria produce polysaccharides that may also cement microaggregates into macroaggregates. Total polysaccharide content was measured in soil samples and except for the shallowest depth was greater in biochar-amended soil than in the control soil (Fig. 7C). This result is similar to that reported above for glomalin: total polysaccharide content is consistent with greater degree of macroaggregation in biochar-amended versus control soil except for 0-7.5 cm.

Bacterial and fungal activity, as measured by extracellular polysaccharides and extracellular proteins, respectively, is different in un-amended and biochar-amended soils. This different biological activity may contribute to different aggregation rates and particle size distributions, which in turn leads to altered water infiltration rates.

5 CONCLUSIONS

- Biochar stabilizes soil aggregates under wet conditions, but chemical constituents in the water do not affect aggregation processes.
- The measured particle size distribution in both field samples and laboratory microcosms has a larger average diameter and greater mass fraction in larger size fractions than would be expected if aggregation did not occur.
- Formation of aggregates >2 mm diameter requires more than 16 weeks of incubation, or incubations that mimic the wetting/drying cycles that occur in nature.
- Biological activity contributes to aggregate formation, specifically via proteins (glomalin) and polysaccharides that have adhesive properties.

6 RECOMMENDATIONS

6.1 AMENDMENT OF ROADWAY STORMWATER FACILITIES

Amendment of a sandy loam roadway soil with 4% wood biochar effectively increases water infiltration rates. While field tests demonstrated this process for this soil/biochar combination, it is unclear if increases in infiltration will be as significant for other soil/biochar mixtures. Recent studies investigating biochar application to agricultural soils indicate that biochar amendment does not always cause increased soil aggregation and infiltration. Similar limitations may occur for application to roadway soils.

The laboratory portion of this study, using the same sandy loam soil and same biochar as in the field test, was intended to provide a better understanding of the soil aggregation process. Over the 16-week period of testing, significant soil aggregation was not observed. This may be due to the need for longer incubation time, or it may be because of the experimental methodology, where stormwater was added periodically to maintain constant soil wetness. While the procedures followed are standard in the soil science community for assessing soil aggregation, they do not mimic the natural wetting/drying cycles that occur for roadway soils subjected to periodic stormwater events.

Because biochar amendment to soils does not always result in increased soil aggregation, it is important that laboratory and/or small pilot-scale field tests be conducted for each biochar/soil combination to verify that the mechanism will occur before large-scale application of biochar.

6.2 FUTURE RESEARCH

Future research investigating mechanisms of soil aggregate formation should extend the time of incubation of soil microcosms, investigate the dynamic changes in microbial and fungal populations, and assess how changes in water infiltration rates affect rates of contaminant removal. In addition, laboratory testing should mimic the natural wetting/drying cycles in the field.

While this investigation documented changes in microbial and fungal communities that were the likely causes of increased soil aggregation and thus stormwater infiltration, it is unclear what properties of the biochar favored these changes. Future research should assess the properties of

biochar that are critical for enhancing soil microbial communities that promote soil aggregation and thus increased stormwater infiltration.

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