Microbial Response to Potential Soil-Stabilizing Polymer Amendments for Coastal Wetland Restoration

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Microbes release extracellular polymeric substances during metabolism of organic matter, which accumulate to bind particles and increase soil aggregation. On a large scale, hydraulically dredged sediment can be amended with polymer and deposited on subsiding marshes, where the polymer stabilizes the sediment until marsh plants become established. However, adding a simple C source to the soil can potentially affect microbial activity. This study determined the effect of addition of two commercially available natural polymers (xanthan gum and guar gum) on microbial biomass and activity in three types of hydraulically dredged sediments (clay, silty clay, and sandy loam) saturated under a range of salinity regimes (1.49 and 7.46, 7.46 and 14.9, and 22.4 and 37.3 mS cm^{-1}, respectively) for four time periods (1, 8, 16, and 26 wk). The CO_2 evolved in response to added polymer suggests that microbial communities rapidly degraded the polymers. Addition of polymers provided a readily available source of C that induced a priming effect on the microbial biomass leading to increased activity. Microbial activity accelerated to a much greater rate than background (control) respiration, resulting in up to an 8.7-fold increase in loss of native soil C beyond degradation of the added polymer C. Therefore, polymer additions to stabilized sediments led to a significant increase in native soil C loss with a concomitant decrease in soil quality.

Abbreviations: CEC, cation exchange capacity; EPS, extracellular polymeric substance;

Over the last 1200 yr, the Mississippi River has delivered sediment to the Bird Foot Delta (Törnqvist et al., 1996). Levee construction and sedimentation along the lower Mississippi River have led to the extension of the delta out to the edge of the continental shelf into the Gulf of Mexico, where the depth abruptly increases to 305 m. Consequently, most of the sediment currently being carried by the River is deposited into the deeper waters of the Gulf and is not available to build land. The coastal subsidence rate of 1 to 2 cm yr^{-1} (Boesch et al., 1994) coupled with the global eustatic sea level rise of 1 to 2 mm yr^{-1} results in a very high relative rate of relative sea level rise with subsidence comprising the major portion. Current restoration efforts include diverting Mississippi River water into coastal basins to deliver water, nutrients and sediment to simulate the River's hydrology before levee building (Gardner and White, 2010; White et al., 2009a).

Coastal Louisiana, similar to other low-lying coastal areas, is vulnerable to hurricane forces, which scour sediment as a result of storm surge. In 2005, Hurricanes Katrina and Rita collectively led to more than 50,000 ha of wetland loss, resulting in open water (Barras, 2009). The state of Louisiana requires a wetland restoration solution that will protect coastal communities from increasing encroachment by the waters of the Gulf of Mexico. The U.S. Army Corps of Engineers has produced technical reports on the beneficial use of hydraulically dredged material for wetland restoration (U.S. Army Engineer Waterways Experiment Station, 1978). The use of natural polymers to improve stability of hydraulically dredged sediments is currently being studied as a possible technique to increase the stability of these saturated dredged sediment slurries for wetland restoration. This sediment then

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becomes the platform on which the marsh plants become established and accrete organic matter (DeLaune and White, 2011).

Organic matter decomposes at a significantly lower rate under anaerobic flooded conditions than in aerobic soils (DeBusk and Reddy, 2003; White and Reddy, 2001) and may aid in aggregation. The ability of soil aggregates to remain stable when flooded depends on available organic materials (Tisdall and Oades, 1982; Martens and Frankenberger, 1992). Organic matter provides a substrate for microbial production of extracellular polymeric substances (EPS). As soil organisms decompose organic materials, microbial secretions bind soil particles and small organic particles together (Tiessen and Stewart, 1988; Zhang et al., 2005). Higher microbial activity leads to greater EPS production, which leads to greater aggregation as EPS trap soil particles (Zhang et al., 2005). Even after microbial growth decelerates, microbial byproducts remain in the soil and continue to aggregate soil particles (Martens and Frankenberger, 1992). As the EPS content of intertidal soils increases, critical erosion velocity also increases, suggesting that these microbial secretions may increase sediment cohesiveness and sediment stability (Widdows et al., 2006). Polymeric substances are C compounds that exist naturally in soil, however, and undergo molecular and morphological changes over time, thus increasing the degradation rate by the microbial pool (Ratajska and Boryniec, 1998). The polymeric material unfolds, which increases the particle surface area to volume ratio available for microbial activity.

To further understand the influence of commercially available natural polymers on sediment stability, a separate component of the overall study included an engineering study that investigated the interactions between natural polymer solutions (i.e., xanthan gum and guar gum) and research-grade pure kaolinite clay (Nugent et al., 2009). At higher biopolymer concentrations in pore fluid, the liquid limit of kaolinite increased as solution viscosity increased (Nugent et al., 2009). In general, since undrained shear strength of a soil depends on moisture content, the polymers' effect of increasing the liquid limit of clays should inherently increase the undrained shear strength of an amended soil.

This study focused on the impact of natural polymers on microbial activity in hydraulically dredged sediments. Measures of microbial biomass and respiration rates provide insight about the activity of microbial communities in soils and sediments. Microbial biomass defines the microbial component of soil that contains both active and dormant microbial life stages (Sparling et al., 1985). Respiration rates represent the physiologically active component of the biomass since only microbes in the active life stage respond to substrate addition or nutrient input (Sawada et al., 2008). Physical properties such as structure and texture of soil influence size of the microbial community. Clay soils, compared with sandy soils, have a greater capacity for retaining C in the soil organic matter component because the C is protected in smaller pore spaces (Van Veen and Kuikman, 1990). In addition, soils with higher clay content have enhanced biomass retention after substrate addition for the following reasons: lower turnover rate of microbial products, increased retention of microbial biomass and organic matter, and increased nutrient adsorption (Wardle, 1992). This study determined the effect of two commercially available natural polymers (xanthan gum and guar gum) on microbial biomass and activity in three types of hydraulically dredged sediments (clay, silty clay, and sandy loam) saturated under three salinity regimes (1.49 and 7.46, 7.46 and 14.9, and 22.4 and 37.3 mS cm^{-1}) for four time periods (1, 8, 16, and 26 wk). The two polymers are potential sources of hydrolyzable C substrate that may be available for microbial degradation.

MATERIALS AND METHODS

Sediment Sample Locations

Sediment was collected at three sites in southern coastal Louisiana (Fig. 1). Bayou Chevreuil represents a freshwater site containing sediment with high clay content. Leeville represents an intermediate salinity

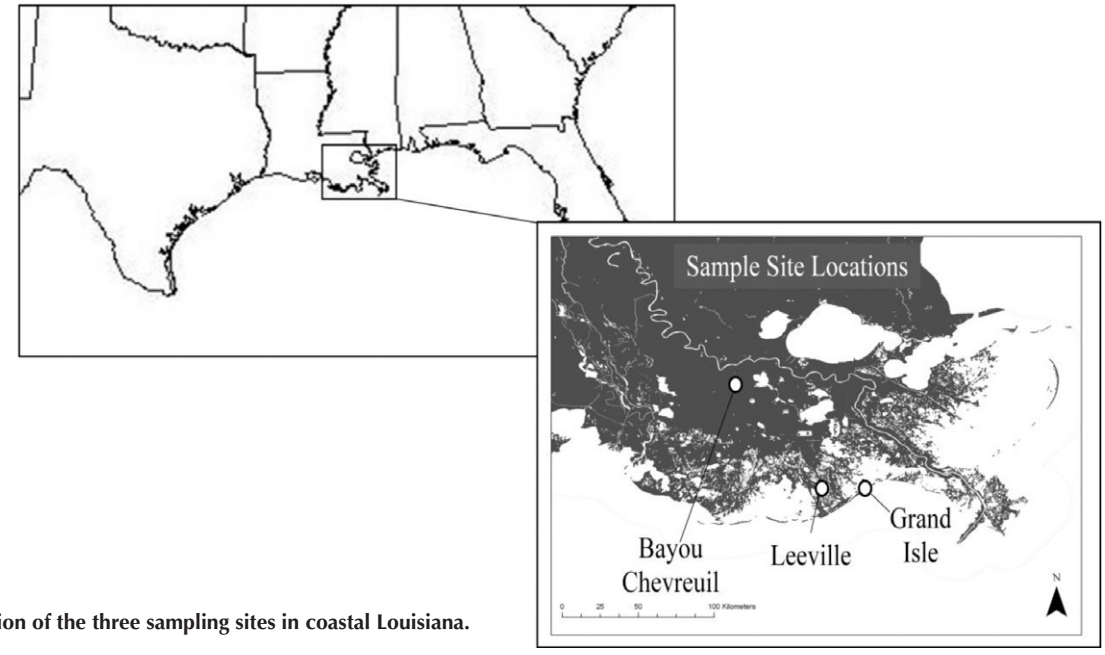

Fig. 1. Location of the three sampling sites in coastal Louisiana.

site containing sediment with moderate clay content. Grand Isle represents a marine site with predominantly sandy sediment.

Sediment Characterization

The sediment was passed through a 0.635-cm sieve to remove large plant debris. To homogenize the sediment, an electric drill attached to a paint mixer was used for 5 min in the forward direction and 5 min in the reverse direction to form a slurry similar in consistency to that produced from hydraulic dredging. The sediments were stored at 4°C for 7 mo while preliminary tests and sediment characterization were conducted. All three sediments were characterized for the following properties: moisture content, organic matter content, redox potential, pH, soil salinity, cation exchange capacity, and particle-size distribution. For moisture content, three subsamples of each sediment type were dried at 105°C until a constant weight was reached. Percentage of moisture content was calculated on a wet weight basis. Organic matter content was determined by the loss-on-ignition method (Nelson and Sommers, 1996).

For redox potential, platinum-tipped electrodes were cleaned and tested as described by Patrick et al. (1996). Platinum electrodes were inserted into sediment samples and used in conjunction with a calomel reference electrode to obtain a reading in millivolts (E_c). The values were then corrected to a standard hydrogen reference electrode (E_h). For soil pH, a calibrated combination pH electrode with a Ag/AgCl reference electrode was used (Thomas, 1996). For soil salinity, soil pore water was collected by centrifuging field moist sediment in a Fisher Scientific accuSpin 3/3R centrifuge (at 3000 × *g* for 15 min). The supernatant was then analyzed for salinity with an Accumet AB30 conductivity meter (Rhoades, 1996).

Cation-exchange capacity (CEC) was determined by the unbuffered salt extraction method according to Sumner and Miller (1996). All three sediments were saturated with 0.2 mol L^{-1} NH_4Cl, washed with deionized water, and saturated with 0.2 mol L^{-1} KNO_3 to displace the NH_4^+. The extracted supernatant was analyzed for exchangeable NH_4^+ (USEPA-103-A Rev. 4; United States Environmental Protection Agency, 1993) using a SEAL $AQ2^+$ automated discrete analyzer (Seal Analytical, Mequon, WI).

Particle-size distribution and textural class were determined by the hydrometer procedure (White et al., 2009b). Sediments were pretreated to remove carbonates and soluble salts using sodium acetate, organic matter using hydrogen peroxide, and free iron oxides using citrate-bicarbonate, sodium dithionite, and sodium chloride. Values from hydrometer readings were used to determine the proportion of sand, silt, and clay in all three sediments (Patrick, 1958).

Polymers

The polymer treatments included two natural polymers (xanthan gum and guar gum) that are commercially available (Table 1). Xanthan gum is an extracellular polysaccharide produced by the bacterium *Xanthomonas campestris* (Kim et al., 2006). Guar gum is extracted from the seed of a guar gum plant, a leguminous shrub known as *Cyamopsis tetragonoloba* (Kim et al., 2006). Polymer powder blended with water of varying salinities resulted in xanthan gum and guar gum polymer solutions made to concentration levels of 1 and 2% by weight.

Experimental Design

The experimental units were polyethylene cups with a capacity of 450 mL. The units contained 350 g of field moist sediment mixed with 175 g of 1 or 2% (w/w) polymer solution. The 2:1 sediment/polymer ratio resulted in final concentrations of polymer at 0.5 and 1% by weight. Control experimental units (i.e., sediments with no polymer amendment) received 175 mL of water of the appropriate salinity. Two different salinity solutions were applied to all experimental units for each sediment to simulate in situ salinity ranges. Salinity treatments were 1.49 and 7.46 mS cm^{-1} for the freshwater sediment, 7.46 and 14.9 mS cm^{-1} for the intermediate sediment, and 22.4 and 37.3 mS cm^{-1} for the marine sediment.

A randomized block design was implemented to evaluate how several dependent variables were affected by sediment type (i.e., sampling location), salinity, polymer type, and polymer concentration over four time periods: 1, 8, 16, and 26 wk. Response variables measured included redox potential, microbial biomass C, and microbial respiration rates. Each treatment was prepared in triplicate, which was the basis for the block design. There were 432 total experimental units (3 sediment types × 2 salinities × 2 polymers × 3 concentrations × 4 time periods × 3 replicates). Each block contained 144 experimental units, which allowed all treatment combinations to be present in three different trials.

A Barnstead Max-Q 2508 reciprocating shaker (Barnstead International, Dubuque, IA) was used to mix each sediment-polymer combination and each control unit. With a fixed circular orbit of 1.2 cm, each experimental unit shook at the maximum setting (400 rpm) for 15 min. Then, each mixture was poured into the 450-mL opaque polyethylene cups and set on the lab bench before being analyzed at predetermined time points (i.e., the end of 1, 8, 16, or 26 wk). All experimental units remained uncovered for the designated time period and were kept in the lab at a steady temperature of 20°C. The experimental units were exposed to daily light regimes with all lab lights on for 12 h per day. For the duration of each time period, decanting of any remaining supernatant fluid and reflooding with water of the appropriate salinity took place every 8 to 10 d to maintain anaerobic flooded conditions. Destructive sampling was employed. At the end of each designated time period, redox potential was measured. Then, the samples were stored for a maximum of 3 d at 4°C before further analysis.

Microbial Biomass Carbon Determination

Chloroform fumigation extraction (Vance et al., 1987), as modified by White and Reddy (2001), was conducted to determine the pool of microbial biomass C in each sample at each time point. From each experimental unit, two samples (one designated as fumigate [F]

Table 1. Properties of xanthan gum and guar gum.

Polymer	Source	Molecular formula	Molecular weight	Charge	Charge density	% Total C
Xanthan gum	microbial extracellular polymer	$(C_{35}H_{49}O_{29}$ n	0.9- 1.6 x 10^6 Da	anionic	0.25†	40.84
Guar gum	plant polysaccharide	$(C_{18–20}H_{30}O_{15})$n	1.0- 2.0 x 10^6 Da	non-ionic	0	43.25

†Charge density in mol/mol monosaccharide

and one designated as non-fumigate [NF]) were weighed out into 25-mL centrifuge tubes. For every 10 experimental units, both F and NF samples were represented in triplicate to have a set of triplicates present for each vacuum filtration. All F samples were exposed to chloroform fumes for 24 h in a vacuum desiccator. Twenty-five milliliters of K_2SO_4 extractant were added to each tube and placed on a reciprocating shaker for 30 min (Malecki-Brown and White, 2009). Samples were centrifuged in a Fisher Scientific accuSpin 3/3R centrifuge (Thermo Electron Corp., Marietta, GA) at 3450 × *g* for 10 min. After centrifugation, the supernatant was immediately filtered (Whatman 42 filter paper) by vacuum filtration. Samples were analyzed for total organic C on a Shimadzu TOC-V series carbon analyzer (Shimadzu, Kyoto, Japan). The microbial biomass C underwent a correction according to Vance et al. (1987), using a K_{EC} of 2.70 (Sparling et al., 1990).

Microbial Basal Respiration Measurements

One week after the start of the experiment, 5 g of sediment from each experimental unit with the 1 wk time treatment were weighed into 60-mL glass serum bottles (total of 108 samples). Ten milliliters of water were added, and the bottles were sealed with a rubber septa and aluminum cap. Headspace gas was removed to −68 kPa with a vacuum pump then flushed with 99.9% pure N_2 gas for 5 min. Bottles were maintained at a temperature of 25° C for the duration of the study.

After 35 d of incubation, the pressure of each serum bottle was measured using a SPER Scientific Manometer (SPER Scientific, Scottsdale, AZ). Gas samples were then withdrawn (either 50 or 100 μL) and analyzed for CO_2 on a Shimadzu gas chromatograph GC-2012 (thermal conductivity detector at 160°C; packed Poropak N column [6 ft; 80/100 mesh] with an oven temperature of 80°C). Gas samples were withdrawn and analyzed every 10 d from Day 35 to Day 125, which was determined as the period of peak CO_2 evolution. Moles of CO_2 were determined using the ideal gas law and moles of CO_2 were converted to g CO_2–C kg^{-1} dry sediment (DeBusk and Reddy, 2003).

Statistical Analyses

SAS 9.1 software (2009) and SigmaPlot 11.0 software (2008) were used to analyze and plot the data. For redox potential, microbial biomass, and microbial respiration, a simple 5-way ANOVA was conducted. After a test of Type III fixed effects with a Tukey adjustment, least squares means analysis was evaluated to look for differences between any significant effects for all dependent variables. An α value of 0.05 was used for all analyses. All presented *p* values represent results of least-squares means analysis (Tables 2, 3, and 4). Numerical results are given as the mean ± standard error. All error bars represent standard error. For all analyses, salinity and polymer type were not found to be significant; therefore, all values for different salinity levels and polymer types were combined. Organic matter, clay content, microbial biomass, and redox potential values were correlated using Pearson Product Moment correlations.

RESULTS

Sediment Characterization

The three sediments had unique characteristic properties (Table 5). In the case of the marine sediment, primarily an inorganic sediment with <2% (by weight) organic matter, we would expect a higher redox potential than the more organic-rich fresh-

Table 2. Statistical results following a five-way ANOVA, Type 3 test of fixed effects with a Tukey adjustment, and least-squares means analysis for redox potential data. A = sediment, B = salinity (sediment), C = polymer, D = concentration, E = week.

Effect	Num DF	Den DF	F Value	Pr > F
A	2	287	101.59	< 0.0001
B	3	287	1.37	0.2522
C	1	287	1.17	0.2806
A×C	2	287	0.87	0.421
B×C	3	287	0.02	0.9969
D	2	287	66.67	< 0.0001
A×D	4	287	11.82	< 0.0001
B×C	6	287	1.44	0.2005
C×D	2	287	0.91	0.404
A×C×D	4	287	5.2	0.0005
B×C×D	6	287	0.59	0.7395
E	3	287	19	< 0.0001
A×E	6	287	3.47	0.0025
B×E	9	287	1.15	0.3255
C×E	3	287	0.35	0.7882
A×C×E	6	287	1.04	0.4008
B×C×E	9	287	0.24	0.9877
D×E	6	287	2.14	0.0495
A×D×E	12	287	0.93	0.5122
B×D×E	18	287	0.49	0.9616
C×D×E	6	287	0.65	0.6926
A×C×D×E	12	287	2.28	0.0087
B×C×D×E	18	287	0.61	0.8904

Table 3. Statistical results following a five-way ANOVA, Type 3 test of fixed effects with a Tukey adjustment, and least-squares means analysis for microbial biomass data. A = sediment, B = salinity(sediment), C = polymer, D = concentration, E = week.

Effect	Num DF	Den DF	F Value	Pr > F
A	2	288	215.45	< 0.0001
B	3	288	1.79	0.1491
C	1	288	0.4	0.5267
A×C	2	288	2.24	0.1088
B×C	3	288	0.73	0.5357
D	2	288	0.77	0.465
A×D	4	288	0.43	0.785
B´C	6	288	0.4	0.8806
C×D	2	288	0.03	0.968
A×C×D	4	288	0.35	0.8453
B×C×D	6	288	0.54	0.778
E	3	288	21.56	< 0.0001
A×E	6	288	8.31	< 0.0001
B×E	9	288	0.41	0.9319
C×E	3	288	0.72	0.5402
A×C×E	6	288	0.51	0.8013
B×C×E	9	288	0.28	0.9791
D×E	6	288	0.38	0.8914
A×D×E	12	288	0.28	0.9925
B×D×E	18	288	0.33	0.996
C×D×E	6	288	0.05	0.9993
A×C×D×E	12	288	0.15	0.9996
B×C×D×E	18	288	0.26	0.9991

water and intermediate sediments. However, we suggest that hurricane effects may have played a role. The marine sediment had a visible layer of 1- to 2-cm fresh organic material from the neighboring wetlands deposited at the surface from recent hurricane storm surge. This available organic material likely led to the lower than expected measured redox potential values. This sediment also exhibited a noticeable hydrogen sulfide odor indicating strongly reducing conditions consistent with our readings.

Redox Potential

The presence of a polymer did have a significant effect on redox potential for the intermediate and marine sediments (Fig. 2). For the control samples, the redox potential values for all three sediments across time periods were not significantly different, with values of 16, −22, and −12 mV for the freshwater, intermediate, and marine sediments, respectively. In general, the redox potential for the freshwater sediment was significantly higher (i.e., more positive) than the redox potential for the intermediate and marine sites for Weeks 1, 8, and 16 ($P < 0.0001$, Fig. 2). The redox potential values for the intermediate and marine sediments were not significantly different from each other at any time period. Redox potential of the freshwater sediment amended with a polymer was not significantly different from redox potential of the control samples. The redox potential decreased in the first 8 wk of the experiment, which was followed by a significant increase by Week 16 ($P = 0.0075$).

The presence of a polymer did significantly affect redox potential values for both the intermediate and marine sediments compared with control samples ($P < 0.0001$, Fig. 2). The redox potential values for the two concentration levels were not significantly different from each other. For the intermediate sediment, the redox potential values for samples with polymer decreased to −180 ± 15.9 mV by Week 8, followed by a significant increase by Week 26 ($P = 0.0020$). These values were significantly lower than the average redox potential value of −22 ± 12.7 mV for the intermediate control sediment ($P < 0.0001$). For the marine sediment, the redox potential values for samples with polymer decreased to −176 ± 10.3 mV by Week 8, followed by a significant increase by Week 26 ($P < 0.0001$). These values were significantly lower than the average redox potential value of −11.5 ± 13.4 mV for the marine control sediment ($P < 0.0001$).

Table 4. Statistical results following a five-way ANOVA, Type 3 test of fixed effects with a Tukey adjustment, and least-squares means analysis for basal respiration data. A = sediment, B = salinity(sediment), C = polymer, D = concentration, E = day.

Effect	Num DF	Den DF	F Value	Pr > F
A	2	69.9	58.67	< 0.0001
B	3	69.8	0.86	0.4683
C	1	71.1	2.27	0.1364
A×C	2	69.9	1.56	0.2168
B×C	3	69.8	3.42	0.0219
D	2	71	130.45	< 0.0001
A×D	4	69.6	5.96	0.0003
B×C	6	69.8	1.04	0.4094
C×D	2	71	3.61	0.0322
A×C×D	4	69.6	1.77	0.1454
B×C×D	6	69.8	0.91	0.4915
E	9	413	4.21	< 0.0001
A×E	17	412	3.19	< 0.0001
B×E	26	412	0.63	0.9193
C×E	9	413	0.88	0.5434
A×C×E	17	412	0.6	0.8897
B×C×E	26	412	0.79	0.7566
D×E	18	413	2.31	0.0019
A×D×E	34	412	1.3	0.1278
B×D×E	52	412	1.05	0.3828
C×D×E	18	413	0.5	0.9599
A×C×D×E	34	412	0.57	0.9774
B×C×D×E	52	412	0.62	0.9835

Table 5. Characteristic properties of the freshwater, intermediate, and marine sediments.

Soil properties	Freshwater	Intermediate	Marine
Moisture Content % (wet weight)	74.8	67.1	36.3
OM Content %	13.8%	8.34%	1.56%
Redox Potential (mV)	−26	−18	−207
Soil pH	6.50	6.90	6.90
CEC (cmol charge kg $^{-1}$ dry sediment)	125.0	84.6	27.7
Porewater Salinity (ppt)	0.50	4.60	15.5
% Sand	9.17	19.2	70.8
% Silt	20.8	38.3	13.3
% Clay	70.0	42.5	15.8
Textural Class	Clay	Silty Clay	Sandy Loam

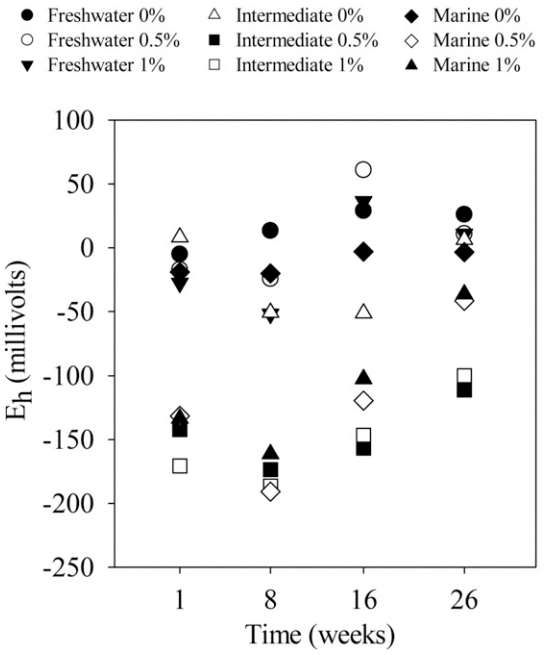

Fig. 2. Mean final redox potential values (mV) for the freshwater, intermediate, and marine sediments for Weeks 1, 8, 16, and 26 at polymer concentrations 0, 0.5, and 1% by weight. Values for polymer types and salinity levels for each sediment have been averaged due to no significant differences ($n = 48$).

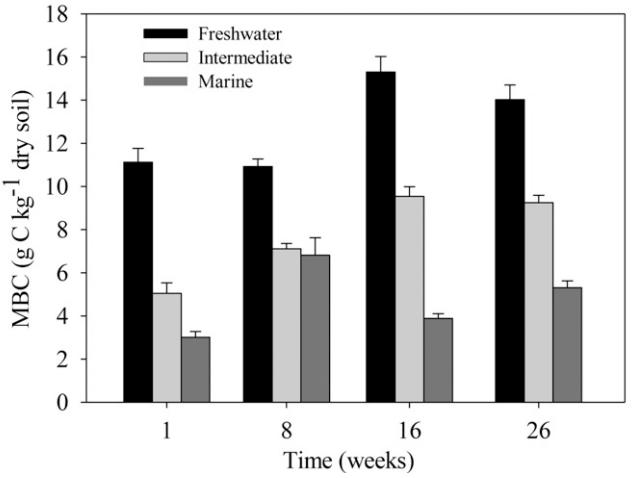

Fig. 3. Microbial biomass C (g C kg^{-1} dry soil) for the freshwater, intermediate, and marine sediments at Weeks 1, 8, 16, and 26. Values for polymer type, concentration level, and salinity have been averaged due to no significant differences. (*n* = 36)

Microbial Biomass

The presence of a polymer had no significant effect on the pool of microbial biomass C (MBC). The freshwater sediment had the highest amount of MBC with an average value of 12.8 ± 0.345 g C kg^{-1} dry sediment (Fig. 3). The intermediate sediment had an intermediate amount of MBC averaging 7.74 ± 0.247 g C kg^{-1} dry sediment. The marine sediment had the lowest amount of MBC with an average value of 4.76 ± 0.114 g C kg^{-1} dry sediment. The values are within the range seen in other studies (White and Reddy, 2000; Malecki-Brown et al., 2007; Corstanje and Reddy, 2004).

For the freshwater and intermediate sediments, the MBC significantly increased by Week 16 ($P < 0.0001$). For the marine sediment, MBC peaked in Week 8; the increase by Week 26 was not significant. The MBC at Week 8 was significantly higher than the MBC at Week 1 ($P = 0.0001$) and Week 16 ($P = 0.0133$).

Microbial Basal Respiration

The 1% (w/w) polymer treatment did have a significant effect on basal respiration rates. In general, the freshwater sediment had the highest respiration rates, followed by the intermediate and the marine sediments. The total cumulative amounts of respiration for the control samples were 25.8, 12, and 4.33 g CO_2–C kg^{-1} dry sediment for the freshwater, intermediate, and marine sediments, respectively.

For all three sediments, on any given day of measurement, the microbial respiration with 1% polymer was significantly greater than the control samples ($P < 0.0001$, Fig. 4). In general, for each sediment type, the respiration rate from the 0.5% (w/w) polymer was not significantly different than the control samples. For the freshwater and intermediate sediments, the samples with 1% polymer had a significantly greater respiration rate than the samples with 0.5% polymer ($P < 0.0001$).

Generally, respiration of 50% of C input indicates complete degradation of the material while the remaining C is assimilated into biomass (Shen and Bartha, 1996). At the 0.5% concentration level, the microbial respiration in the freshwater sediment was significantly greater than for the intermediate ($P = 0.0037$) and marine sediments ($P < 0.0001$). At the 1% concentration level, the respiration for all three sediments was significantly different from each other ($P < 0.0045$).

Results suggest that the microbial consortia in all three sediments completely degraded the added polymer (Fig. 5). At the 0.5% polymer concentration level, microbes increased respiration to 6.6-, 4.6-, and 8.0-fold more C than added as polymer for the freshwater, intermediate, and marine sediments, respectively (Table 6). At the 1% polymer concentration level, microbes increased respiration to 8.4-, 7.4-, and 8.7-fold more C than added as polymer for the freshwater, intermediate, and marine sediments, respectively. These results suggest a priming effect in which the polymer stimulates microbial activity thereby increasing the catabolism of the native soil organic C pool. The term priming effect describes accelerated decomposition of soil organic matter after addition of some other organic material (Fontaine et al., 2003).

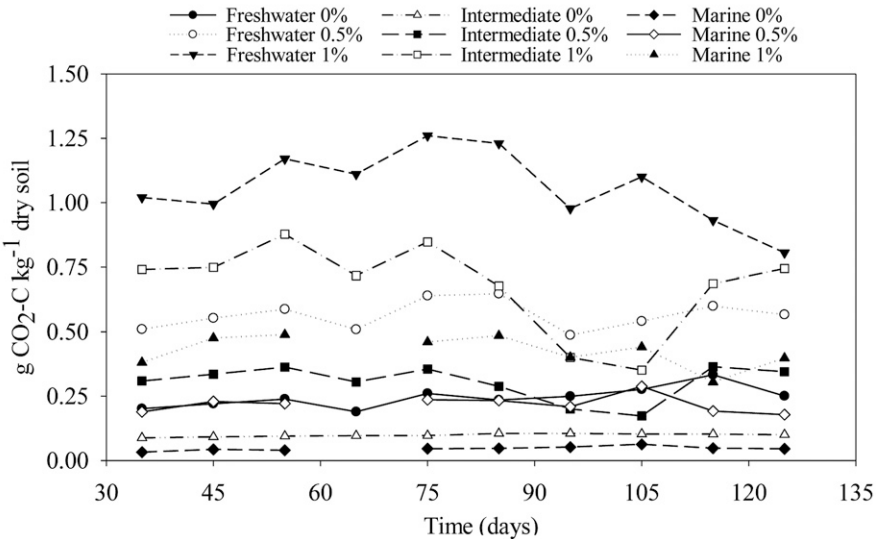

Fig. 4. Basal respiration curves (g CO_2–C kg^{-1} dry soil) for the freshwater, intermediate, and marine sediments at the 0, 0.5, and 1% (by weight) concentration levels. Values for polymer types and salinity levels have been averaged due to no significant differences. (*n* = 12)

DISCUSSION

For all sediments, the polymer immediately increased microbial activity as evidenced by lower redox potential values and higher respiration rates compared with controls. Furthermore, redox potential values continued to decrease in the first 8 wk. Even though the polymer was metabolized, soil microbes continued to maintain an increased C mineralization rate.

The polymer additions did not significantly affect MBC, but the MBC pool size increased over time regardless of soil type. The MBC significantly correlated with organic matter content for each sediment type as seen

in other studies (White and Reddy, 1999; D'Angelo and Reddy, 1999) with a Pearson Product-Moment coefficient value of 0.68 ($P < 0.0001$). For example, the freshwater sediment had the highest MBC and the highest organic matter content whereas the marine sediment had the lowest MBC and the lowest organic matter content. The microbial community metabolized additional C and assimilated it into the biomass pool. For both the freshwater and intermediate sediments, microbial biomass increase was sustained over 16 wk, possibly because those sediments had higher soil organic matter content. In the marine sediment, the biomass reached a peak by Week 8 but then decreased for the rest of the study, likely due to the lower organic matter content of the sediment.

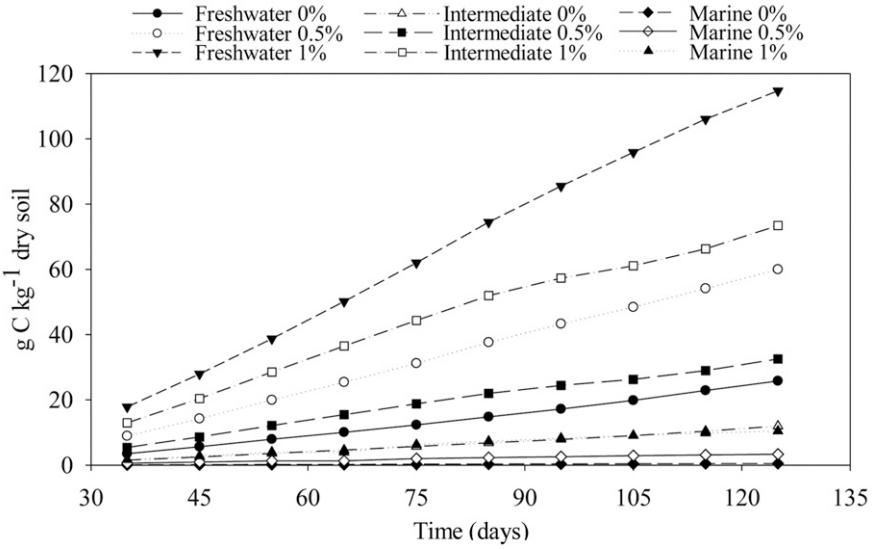

Fig. 5. Cumulative amount of C respired (g C kg^{-1} dry soil) over 125 d for the Freshwater, Intermediate, and Marine sediments at polymer concentrations 0, 0.5, and 1 % (by weight). Values for polymer types and salinity levels have been averaged due to no significant differences. (n = 12)

The cumulative amount of respiration indicates that more C was metabolized than was added from the polymer. After removing the polymer C, microbial communities continued to respire at an accelerated rate. The percentage of respiration over the control samples indicates that additional C substrates from the soil organic matter component were metabolized after the polymer was degraded (Table 6 and Fig. 5). For the freshwater sediment with the 0.5% polymer, an additional 114% respiration above the control and the added polymer led to the loss of 29.5 g C kg^{-1} dry soil. For the freshwater sediment with the 1% polymer, the number increases to 309% respiration above the control and the polymer, resulting in the loss of 79.7 g C kg^{-1} dry soil. For the intermediate salinity sediment, the samples with 0.5 and 1% polymer had 141 and 451% greater respiration above the control and the added polymer. For the marine sediment, the samples with 0.5 and 1% polymer had 347 and 763% greater respiration above the control and the added polymer. The additional percentage of respiration on samples with polymer increases from the freshwater to the intermediate to the marine sediment. Even though the marine sediment had the greatest percentage of respiration above the control, the freshwater sediment still had the greatest loss of soil C in g C kg^{-1} dry soil.

While this manuscript focuses on the effects of polymer additions on microbial processes, effects of the three sediment types, salinity levels, polymer types and addition rates on soil aggregation were also evaluated (Land, 2010). None of the independent variables had a significant effect on aggregation. After considering the conclusions from the lab-based engineering study done as a separate component of the overall study (Nugent et al., 2009), several potential reasons explain why the polymer solutions were not effective at increasing stability of actual dredged sediment from the field. In Nugent's study, polymer impacts on soil stability were determined by the use of pure kaolinite clay with 89% clay content. At a field site with naturally heterogeneous sediments, environmental variables such as high moisture, highly variable clay content, mixtures of clay minerals, presence of organic matter, and microbial communities promote abiotic and biotic variability. Results suggest that the polymer was metabolized very quickly at the beginning of the experimental

Table 6. Amount of C added from the polymer and the cumulative amount of C respired after 125 d for the freshwater, intermediate, and marine sediments at the 0, 0.5, and 1% (by weight) polymer concentration levels. Values for polymer types and salinities have been averaged due to no significant differences.

Sediment	Concentration of polymer	Added C from polymer †	50% Respiration rule	Cumulative area under the curve	Respiration above control and polymer	Respiration above control and polymer
	%	g C kg^{-1} dry soil	g C kg^{-1} dry soil	g C kg^{-1} dry soil	g C kg^{-1} dry soil	%
Freshwater	0	—	—	25.8	—	—
	0.5	9.49	4.75	60	29.5	114
	1	19	9.5	115	79.7	309
Intermediate	0	—	—	12	—	—
	0.5	7.28	3.64	32.5	16.9	141
	1	14.6	7.30	73.4	54.1	451
Marine	0	—	—	4.33	—	—
	0.5	3.76	1.88	21.2	15	347
	1	7.51	3.76	41.1	33	763

†Due to differences in soil moisture content, each sediment type received a different amount of C from the polymer addition.

timeline. Reasons for such a quick metabolism of the polymers relate to increased microbial activity in the sediments and perhaps because both xanthan gum and guar gum are water-soluble, which increases their availability to microbes when mixed with saturated sediments.

Relatively high levels of polymer addition caused an increase in the active component of the microbial biomass. Two possible sources for the increase in respired CO_2 include mineralization of soil organic C or mineralization of secreted microbial by-products and microbial biomass turnover. In either case, metabolism of these materials removes organic material from the sediment that might otherwise lead to aggregation of soil particles and improved soil quality for plant growth. The use of natural polymers to stabilize hydraulically dredged sediments stimulates growth of the microbial community and induces a priming effect, which leads to increased mineralization of natural organic matter and higher concentrations of released CO_2. While the addition of natural polymers did not negatively affect microbial activities in sediments, the polymers may have been decomposed so quickly that they did not contribute to the processes of soil aggregation under the conditions imposed in this study (Land, 2010).

CONCLUSIONS

The addition of two natural polymers (xanthan gum and guar gum) provided an additional source of microbially available organic C that induced a priming effect on the microbial biomass and consequently did not contribute to increased sediment aggregation. As much as a sevenfold increase in microbial activity over the control resulted in a loss of up to 8.7 times more native soil C than the control. Consequently, the addition of these polymers will lead to increased wetland loss as the soil C is converted to CO_2, at least over the short-term. To increase stability of hydraulically dredged sediments, an amendment that is not water-soluble and that resists microbial decomposition might provide a more effective substrate than the two natural, water-soluble polymers studied here. Additionally, an amendment that does not stimulate microbial activity would be preferred to avoid increased loss of soil C and to prevent contributing to CO_2 concentrations in the atmosphere. Further research is needed to investigate the interactions between synthetic polymers and fully saturated sediments.

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REFERENCES

Barras, J.A. 2009. Land area change and overview of major hurricane impacts in coastal Louisiana, 2004–08. U.S. Geological Survey Scientific Investigations Map 3080:1–6.

Black, C.A. (Ed.). 1965. Methods of soil analysis. Part 1. Agron. Monogr. 9. ASA and SSSA, Madison, WI.

Boesch, D.F., M.N. Josselyn, A.J. Mehta, J.T. Morris, W.K. Nuttle, C.A. Simestad, and D.J.P. Swift. 1994. Scientific assessment of coastal wetland loss, restoration, and management in Louisiana. J. Coastal Res. Special Issue No. 20:1–13.

Corstanje, R.D., and K.R. Reddy. 2004. Response of biogeochemical indicators to a drawdown and subsequent reflood. J. Environ. Qual. 33:2357–2366. doi:10.2134/jeq2004.2357

D'Angelo, E.M., and K.R. Reddy. 1999. Regulators of heterotrophic microbial potentials in wetland soils. Soil Biol. Biochem. 31:815–830. doi:10.1016/S0038-0717(98)00181-3

DeBusk, W.F., and K.R. Reddy. 2003. Nutrient and hydrology effects on soil respiration in a northern Everglades marsh. J. Environ. Qual. 32:702–710. doi:10.2134/jeq2003.0702

DeLaune, R.D., and J.R. White. 2011. Will coastal wetlands continue to sequester carbon in response to increase in global sea level? A case study of the rapidly subsiding Mississippi River deltaic plain. Clim. Change 10.1007/s10584–011–0089–6.

Fontaine, S., A. Mariotti, and L. Abbadie. 2003. The priming effect of organic matter: A question of microbial competition? Soil Biol. Biochem. 35:837–843. doi:10.1016/S0038-0717(03)00123-8

Gardner, L.M., and J.R. White. 2010. Denitrification enzyme activity as a potential spatial indicator of nitrate loading in a Mississippi River diversion wetland soil, Louisiana, USA. Soil Sci. Soc. Am. J. 74:1037–1047. doi:10.2136/sssaj2008.0354

Kim, D., H.T. Lai, G.V. Chilingar, and T.F. Yen. 2006. Geopolymer formation and its unique properties. Environ. Geol. 51:103–111. doi:10.1007/s00254-006-0308-z

Land, L. 2010. Physical and microbial responses of dredged sediment to two soil-stabilizing amendments, xanthan gum and guar gum, for use in coastal wetland restoration. Master's Thesis. Louisiana State University.

Malecki-Brown, L.M., and J.R. White. 2009. Effect of aluminum-containing amendments on phosphorus sequestration of wastewater treatment wetland soil. Soil Sci. Soc. Am. J. 73:852–861. doi:10.2136/sssaj2007.0115

Malecki-Brown, L.M., J.R. White, and K.R. Reddy. 2007. Soil biogeochemical characteristics influenced by alum application in a municipal wastewater treatment wetland. J. Environ. Qual. 36:1904–1913. doi:10.2134/jeq2007.0159

Martens, D.A., and W.T. Frankenberger. 1992. Decomposition of bacterial polymers in soil and their influence on soil structure. Biol. Fertil. Soils 13:65–73. doi:10.1007/BF00337337

Nelson, D.W., and L.E. Sommers. 1996. Total carbon, organic carbon, and organic matter. p. 961–1010. *In* D.L. Sparks (ed.) Methods of soil analysis. Part 3. SSSA Book Ser. 5. SSSA, Madison, WI.

Nugent, R.A., G.P. Zhang, and R.P. Gambrell. 2009. Effect of exopolymers an the liquid limit of clays and its engineering implications. Transp. Res. Rec. 2101:34–43. doi:10.3141/2101-05

Patrick, W.H. 1958. Modification of method of particle size analysis. Soil Sci. Soc. Am. Proc. 22:366–367. doi:10.2136/sssaj1958.03615995002200040027x

Patrick, W.H., R.P. Gambrell, and S.P. Faulkner. 1996. Redox measurements of soils. p. 1255–1273. *In* D.L. Sparks (ed.) Methods of soil analysis. Part 3. SSSA Book Ser. 5. SSSA, Madison, WI.

Ratajska, M., and S. Boryniec. 1998. Physical and chemical aspects of biodegradation of natural polymers. React. Funct. Polym. 38:35–49. doi:10.1016/S1381-5148(98)00031-5

Rhoades, J.D. 1996. Salinity: Electrical conductivity and total dissolved solids. p. 417–435. *In* D.L. Sparks (ed.) Methods of soil analysis. Part 3. SSSA Book Ser. 5. SSSA, Madison, WI.

Sawada, K., S. Funakawa, and T. Kosaki. 2008. Soil microorganisms have a threshold concentration of glucose to increase the ratio of respiration to assimilation. J. Plant Nutr. Soil Sci. 54:216–223. doi:10.1111/j.1747-0765.2007.00235.x

Shen, J., and R. Bartha. 1996. Metabolic efficiency and turnover of soil microbial communities in biodegradation tests. Appl. Environ. Microbiol. 62:2411–2415.

Sparling, G.P., C.W. Feltham, J. Reynolds, A.W. West, and P. Singleton. 1990. Estimation of soil microbial C by a fumigation extraction method– use on soils of high organic-matter content, and a reassessment of the K_{EC} factor. Soil Biol. Biochem. 22:301–307. doi:10.1016/0038-0717(90)90104-8

Sparling, G.P., A.W. West, and K.N. Whale. 1985. Interference from plant-roots in the estimation of soil microbial ATP, C, N and P. Soil Biol. Biochem. 17:275–278. doi:10.1016/0038-0717(85)90060-4

Sumner, M.E., and W.P. Miller. 1996. Cation exchange capacity and exchange coefficients. p. 1201–1229. *In* D.L. Sparks (ed.) Methods of soil analysis. Part 3. SSSA Book Ser. 5. SSSA, Madison, WI.

Thomas, G.W. 1996. Soil pH and soil acidity. p. 475–490. *In* D.L. Sparks (ed.) Methods of soil analysis. Part 3. SSSA Book Ser. 5. SSSA, Madison, WI.

Tiessen, H., and J.W.B. Stewart. 1988. Light and electron-microscopy of stained microaggregates—The role of organic-matter and microbes in soil aggregation. Biogeochemistry 5:312–322. doi:10.1007/BF02180070

Tisdall, J.M., and J.M. Oades. 1982. Organic-matter and water-stable aggregates in soils. J. Soil Sci. 33:141–163. doi:10.1111/j.1365-2389.1982.tb01755.x

Tornqvist, T.E., T.R. Kidder, W.J. Autin, K. van der Borg, A.F.M. de Jong, C.J.W. Klerks, E.M.A. Snijders, J.E.A. Storms, R.L. van Dam, and M.C. Wiemann. 1996. A revised chronology for Mississippi river subdeltas. Science 273:1693–1696. doi:10.1126/science.273.5282.1693

U.S. Army Engineer Waterways Experiment Station. 1978. Wetland habitat development with dredged material: Engineering and plant propagation. Tech. Rep. DS-78–16, NTIS No. AD A073 493. U.S. Army Corp. of Engineers, Vicksburg, MS.

Vance, E.D., P.C. Brookes, and D.S. Jenkinson. 1987. An extraction method for measuring soil microbial biomass-C. Soil Biol. Biochem. 19:703–707. doi:10.1016/0038-0717(87)90052-6

Van Veen, J.A., and P.J. Kuikman. 1990. Soil structural aspects of decomposition of organic-matter by microorganisms. Biogeochemistry 11:213–233. doi:10.1007/BF00004497

Wardle, D.A. 1992. A comparative-assessment of factors which influence microbial biomass carbon and nitrogen levels in soil. Biol. Rev. Camb. Philos. Soc. 67:321–358. doi:10.1111/j.1469-185X.1992.tb00728.x

White, J.R., R.W. Fulweiler, C.Y. Li, S. Bargu, N.D. Walker, R.R. Twilley, and S.E. Green. 2009a. The Mississippi River Flood of 2008—Characteristics of a large freshwater diversion on physical, chemical and biological characteristics of a shallow, estuarine lake. Environ. Sci. Technol. 43:5599–5604. doi:10.1021/es900318t

White, J.R., R.D. DeLaune, C.Y. Li, and S. Bentley. 2009b. Distribution of methyl and total mercury in Louisiana and Georgia shelf sediments. Anal. Lett. 2:1219–1231. doi:10.1080/00032710902901947

White, J.R., and K.R. Reddy. 1999. The influence of nitrate and phosphorus loading on denitrifying enzyme activity in Everglades wetland soils. Soil Sci. Soc. Am. J. 63:1945–1954. doi:10.2136/sssaj1999.6361945x

White, J.R., and K.R. Reddy. 2000. The effects of phosphorus loading on organic nitrogen mineralization of soils and detritus along a nutrient gradient in the northern Everglades, Florida. Soil Sci. Soc. Am. J. 64:1525–1534. doi:10.2136/sssaj2000.6441525x

White, J.R., and K.R. Reddy. 2001. Influence of selected inorganic electron acceptors on organic nitrogen mineralization in Everglades soils. Soil Sci. Soc. Am. J. 65:941–948. doi:10.2136/sssaj2001.653941x

Widdows, J., M.D. Brinsley, N.D. Poper, F.J. Staff, S.G. Bolam, and P.J. Somerfield. 2006. Changes in biota and sediment erodability following the placement of fine dredged material on upper intertidal shores of estuaries. Mar. Ecol. Prog. Ser. 319:27–41. doi:10.3354/meps319027

Zhang, B., R. Horn, and P.D. Hallett. 2005. Mechanical resilience of degraded soil amended with organic matter. Soil Sci. Soc. Am. J. 69:864–871. doi:10.2136/sssaj2003.0256