

ASSESSING SALT MARSH HEALTH: A TEST OF THE UTILITY OF FIVE POTENTIAL INDICATORS

Steven C. Pennings¹, V. Dan Wall², Darrin J. Moore³, Mala Pattanayek⁴, Tracy L. Buck, and James J. Alberts

*University of Georgia Marine Institute
Sapelo Island, Georgia, USA 31327*

¹ *Present address: Department of Biology and Biochemistry
University of Houston,
Houston, Texas, USA 77204
E-mail: spennings@uh.edu*

² *Present address: Environmental Protection Agency
999 E. 18th St., Suite 300, (8EPR-PS)
Denver, Colorado, USA 80202*

³ *Present address: Department of Forest Science
306 Richardson Hall
Oregon State University
Corvallis, Oregon, USA 97331*

⁴ *Present address: Harding ESE
90 Digital Dr.
Novato, California, USA 94949*

Abstract: We examined the utility of five measures of salt marsh function, focusing on angiosperms and microbes, as potential indicators of salt marsh health. We studied twelve salt marsh creeks around Charleston Harbor, South Carolina, USA, six of which were polluted with metals and/or organic compounds and six of which were relatively pristine. Physical variables (sediment clay-silt content, creek water salinity) did not differ between impacted and reference sites. Microtox toxicity measures (expressed as wet or dry units) did not differ between impacted and reference sites. Photosynthesis and transpiration rates of creekbank *Spartina alterniflora* did not differ between impacted and reference sites, nor did measures of peroxidase activity in *S. alterniflora*. The glutathione concentration of *S. alterniflora* was lower at impacted sites than at reference sites; however, glutathione concentration did not respond to pollution in an earlier study in Georgia, likely because glutathione responds differently to particular chemicals rather than being a generic indicator of plant stress. Overall, these measures showed little promise as rapid indicators of salt marsh health. Other methods, such as quantifying benthic invertebrate taxa, may be more reliable for assessing ecosystem health in salt marsh systems.

Key Words: ecosystem health, environmental impacts, Georgia, glutathione, indicators, Microtox, peroxidase, photosynthesis, pollution, salt marsh, *Spartina alterniflora*, transpiration

INTRODUCTION

Scientists have used two general approaches to examining environmental impacts. The first is to measure levels of known pollutants and compare chemical concentrations to standard guidelines. This approach has the advantages of accurately determining pollutant concentrations, allowing sources of pollutants to be identified, and allowing specification of unambiguous

goals for remediation efforts. A possible weakness of this approach, however, is that it may be difficult to link levels of particular pollutants to biological impacts, especially if multiple pollutants, which might interact synergistically, are present at low levels, or the degree of toxicity of a pollutant varies across an environmental gradient. A complementary approach is to measure some aspect of ecosystem health. Although this approach may not necessarily identify the causes

of variation in ecosystem health, it has the advantages of clearly identifying impacts on natural systems and of allowing ecosystem health to be examined in the absence of information on the presence or concentration of specific pollutants. The latter advantage is particularly useful if it is not known which pollutants might be present at a site or if the number of different pollutants and/or analytical difficulties makes quantifying all the possible pollutants prohibitively expensive. Thus, this "ecosystem health" approach could potentially be used by managers to survey a wide variety of sites in order to identify a limited number of "unhealthy" sites that could then receive more intensive and expensive study.

An unresolved question, however, is how to measure ecosystem health. A variety of standard toxicity assays have been developed and used in particular applications (e.g., Swartz 1989, Traunspurger and Drews 1996, Lewis *et al.* 2001). Standard toxicity assays allow comparisons between systems and may have clearly defined performance characteristics, but they are vulnerable to the criticism that they may not be ecologically relevant to particular habitats. An alternative approach is to measure *in situ* characteristics of the ecosystem (Canfield *et al.* 1994, Lerberg *et al.* 2000). Ideally, all three measures of sediment quality (chemical analyses, toxicity assays, and measures of ecosystem structure or function) would be collected (Lamberson *et al.* 1992, Long 2000), but time and financial constraints may prevent this in many cases.

Salt marshes present unique problems for attempting to measure environmental impacts. Because salt marshes are often intertidal habitats located where rivers meet the sea, their waters and sediments may vary markedly in chemistry (e.g., salinity, oxygen availability, redox potential, sulfide concentration) across steep environmental gradients. As a result, the bioavailability of pollutants may change across estuarine gradients (De Laune and Smith 1985, Simpson and Good 1985, Gambrell 1994). In addition, the harsh physical environment of salt marshes may have selected for biota that are more resistant to chemical insults than the biota of other systems. If so, a given concentration of a pollutant might have less of an impact on ecosystem health in a salt marsh than in some other ecological system. Alternatively, the costs associated with tolerating the harsh environment may make some taxa more vulnerable than expected to novel stressors. Finally, because marshes receive pollutants from entire watersheds, marshes in developed watersheds are likely to accumulate a wide variety of pollutants, making it difficult and expensive to quantify all the pollutants and to predict or study all the possible interactions between different pollutants (Olsen

et al. 1982, Biggs *et al.* 1989). For these reasons, we explored the utility of developing indicators that would allow direct assessment of marsh health in the absence of knowledge about specific pollutant levels.

We worked in the salt marsh system surrounding Charleston harbor. Previous studies have documented pollution levels at selected sites in Charleston harbor (Sanger *et al.* 1999a,b). Comparing sites with known high and low levels of pollutants, we examined the utility of five potential indices of marsh health to detect these impacts. Our indices included Microtox® (an industry standard toxicity assay that has been widely applied), two measures of plant gas exchange (assessing physiological performance of the fundamental building block of marsh systems), and two plant stress enzymes (used widely to measure sublethal stress in other plants but poorly developed for salt marsh systems). Our goal was to determine if any of these techniques might have utility for rapidly characterizing the "health" of an estuarine site in the absence of knowledge about specific pollutant levels.

METHODS

We sampled twelve locations around Charleston Harbor, S.C. (32°50'N, 79°55'W) on July 7–10, 1998 (Figure 1). Sanger *et al.* 1999a,b studied 28 tidal creeks around Charleston Harbor in the summer of 1995 and classified them based on land use (upland versus salt marsh watersheds, degree of development of the watershed). Developed creeks had higher concentrations of trace metals, polycyclic aromatic hydrocarbons, polychlorinated biphenyls, and pesticides (DDT and metabolites) than did undeveloped creeks. Following Sanger *et al.* (1999a,b), we hereafter refer to these as impacted and un-impacted creeks. With assistance from D. M. Sanger and A. F. Holland, we selected six impacted creeks and six similar but un-impacted creeks for further study (Table 1). In selecting these creeks, we attempted to pick 12 creeks that were similar in all respects other than development history and that could be clearly classified as developed or undeveloped. We accessed creekbanks at each site by boat at moderate-to-high tides. All samples were collected concurrently within a 10-m section of each creekbank. Salinity of creek water at each site was measured with a refractometer.

Microtox Analyses

Microtox® analyses examine bacterial luminescence in the presence of an experimental sample. Rooted estuarine macrophytes may absorb pollutants through sediment pore water and/or overlying water. Sediment concentrations of pollutants are usually far greater than in

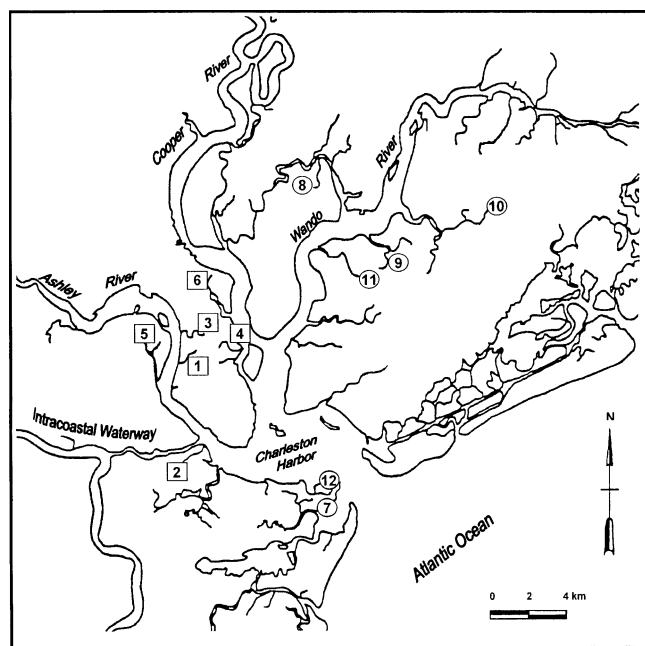


Figure 1. Charleston Harbor, showing locations of study sites (after Sanger et al. 1999a). Squares indicate impacted sites; circles indicate non-impacted sites. 1: Diesel, 2: Dill, 3: Koppers, 4: New Market, 5: Orange Grove, 6: Shipyard, 7: Battery Simkin, 8: Beresford, 9: Fosters, 10: Horlbeck, 11: Rathall, 12: Rice.

the surrounding water, suggesting a greater potential for damage from sediments, both from elevated concentrations and length of time that the macrophytes are exposed to these higher levels of contaminants. While it

has been suggested that quantifying the chemical concentrations in pore water is a better predictor of toxicity than using chemical concentrations in the bulk sediments (Carr et al. 1989, Luoma and Ho 1992), there are disadvantages in using pore water indicators of toxicity, as the sediment chemistry tends to be altered during the extraction procedure (DeValls et al. 1997). Therefore, for this study, we chose to investigate the effect of bulk solid sediment on the test organisms.

Solid Phase Test (SPT) was conducted on sediment samples following the SPT procedures according to the manufacturer's instructions (Microbics Corp. 1992, 1995). Each sample was mixed well, and 7 g of the sample was transferred to a beaker. SPT diluent (3.5% NaCl) was added to the beaker, and the suspension was stirred for 20 minutes. The sample was then serially diluted (1:2, 2 controls and 13 dilutions in duplicate) using SPT diluent by transferring 1.5 ml of the aqueous suspension into the SPT tubes, which were cooled in a water bath at 15°C. After temperature was equilibrated, a timer was set for 20 minutes, and 20 µl of reconstituted test reagent (freeze-dried bacteria mixed with ultra pure water) was pipetted into each SPT tube. The samples were then mixed well, and filter columns were inserted into each tube above the level of suspension. After 20 minutes, sediments were removed by filtration, and 0.5 ml of filtrate was pipetted into glass cuvettes placed in Microtox® wells. Salinity and pH of the samples were not adjusted. Sample light output was measured after 10 minutes using a Microtox® M500 Toxicity Analyzer. This generated a dose response curve that was used to calculate

Table 1. Land use and most abundant pollutants for each creek based on Sanger et al. (1999a,b).

Site	Land Use	Identified Pollutants ¹
Impacted		
Diesel Creek	upland developed industrial	Cr, Pb, Zn, PAH ²
Dill Creek (Plum Island)	salt marsh, highly developed area	
Koppers Creek	upland developed industrial	Cr, Cu, Pb, Zn, PAH
New Market Creek	upland developed urban/suburban	Cr, Cu, Pb, Zn, PAH, PCB ³ , DDT ⁴
Orange Grove Creek	salt marsh, highly developed area	PAH
Shipyard Creek	upland developed industrial	Cr, Cu, Pb, Zn, PAH, DDT, PCB
Not Impacted		
Battery Simkin Creek	salt marsh, undeveloped	
Beresford Creek	forested upland	
Fosters Creek	forested upland	
Horlbeck Creek	forested upland	
Rathall Creek	forested upland	
Rice Creek (Grice Cove)	salt marsh, undeveloped	

¹ Pollutants are listed if they exceeded the following arbitrary cutoff values in the "tidal creek study": Cu: >50 µg/g dry mass, Cr, Pb: >100 µg/g dry mass, Zn: >150 µg/g dry mass, PAH: >1000 ng/g dry mass, PCB: >50 ng/g dry mass, DDT: >10 ng/g dry mass.

² Polycyclic aromatic hydrocarbons.

³ Polychlorinated biphenyls.

⁴ DDT and metabolites.

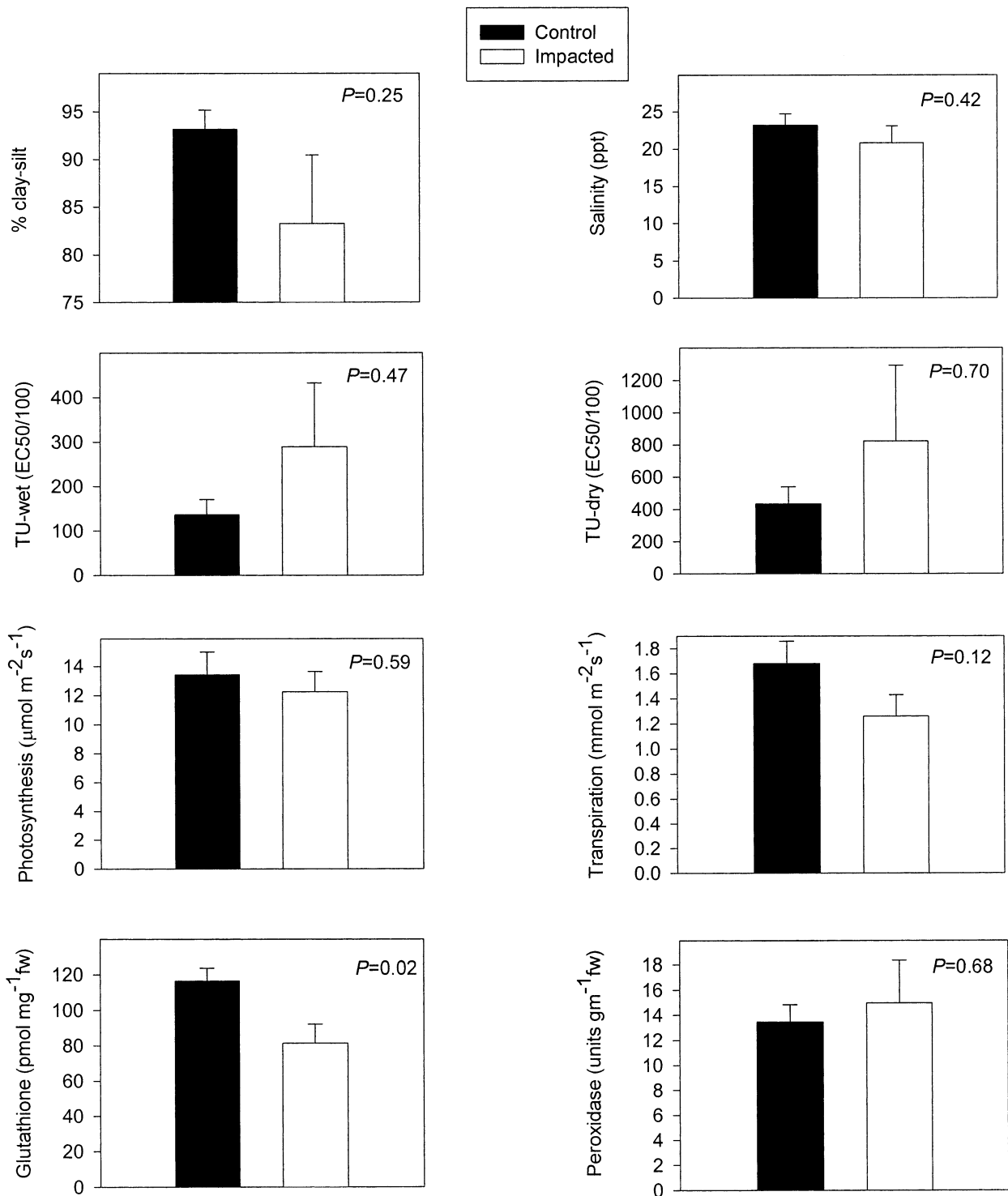


Figure 2. Physical and biological variables measured at impacted and non-impacted sites ($n=6$ each). Data are means \pm 1 SE.

the toxicity units (TU-wet) for the sample, defined as $100/\text{EC}_{50}$. The TU values based on sediment dry weight (TU-dry) were also calculated. Samples were weighed after drying in aliquots (in triplicate) in a vented oven at 100°C for 24 hours.

Percent Clay-Silt

Microtox[®] SPT can be affected by the silt and clay content of sediments, as high silt-clay contents can preferentially adsorb the test bacteria, causing reduction in light emission not necessarily due to toxicity

(Ringwood et al. 1997). Therefore, the clay-silt content of the samples was determined by the standard pipet method of soil particle size analyses (Klemm et al. 1993, Strobel et al. 1995).

Gas Exchange Measurements

Plant gas exchange (photosynthesis, transpiration) rates were used as rapid indicators of production rates. Instantaneous gas exchange measurements were taken using an ADC Corporation, Model LCA4 (Dynamax Inc., Houston, Texas) open gas exchange system and a portable light source ($1.5 \text{ mmol m}^{-2}\text{s}^{-1}$). Readings were taken between 10:00 and 15:00 h on the second fully expanded leaf from the top of 3 separate plants at each site.

Peroxidase Activity (POD) and Glutathione Analysis (tGSH)

Peroxidase isozymes have a wide variety of functions, including chlorophyll degradation, hormonal development and regulation, lignin synthesis, and oxygen radical detoxification (Kato and Shimizu 1985, Siegel 1993). To assess the effects of contamination on peroxidase activity in *S. alterniflora* (Loisel.), POD analysis was conducted via the method of Byl et al. (1994) with minor modifications as described in Wall et al. (2001). The top (youngest) leaf of 3 individual *S. alterniflora* plants was collected and analyzed as a single composite sample/site.

An important route of detoxification of heavy metals in plants is conjugation with reduced glutathione (Mehra et al. 1996, Zenk 1996). These glutathione-bound heavy metals are thought to be transferred to metal chelating polypeptides (Mehra et al. 1996) and transported to vacuoles (Zenk 1996). Total glutathione concentrations (tGSH) in *S. alterniflora* were determined using a modification of the glutathione reductase enzymatic recycling method of Griffith (1980) as described in Wall et al. (2001). As above, a single composite sample of three plants was analyzed from each site.

Statistical Analyses

Subsamples were averaged within sites to yield a single value per site for statistical analyses. Proportional data (% clay-silt) were angular-transformed, and toxicity data were log-transformed before analysis to improve normality. Impacted and reference sites were compared using 2-sample t-tests. To determine if variables covaried among sites, site means for all variables were correlated with each other using Pearson's correlations. Previous studies of a larger group of

creeks (Sanger et al. 1999a,b, Lerberg et al. 2000) found differences between creeks that drained only salt marsh versus upland habitats. Given our smaller set of creeks, including "creek type" as a factor in the analysis (using 2-way ANOVA) did not lead to any increase in explanatory power. For clarity, we therefore present simpler t-test analyses with the two types of creeks grouped together.

RESULTS

Neither of the physical variables that we measured (salinity and % clay-silt) varied significantly between impacted and reference sites (Figure 2). However, the three lowest % clay-silt values coupled with the three lowest salinity values all occurred at impacted sites (Table 2). Thus, it is possible that a more detailed study might have identified significant physical differences between impacted and reference sites. Salinity and % clay-silt were positively correlated across sites (Figure 3, Table 3).

Results of the Microtox[®] assays did not differ significantly between impacted and reference sites (Figure 2, Table 2). TUwet and TUDry values were highly correlated (Table 3). An apparent trend towards higher TU values at impacted sites was largely driven by a single site (Orange Grove) with very high TU values (Table 2).

Plant gas exchange parameters (photosynthesis and transpiration) did not differ significantly between impacted and reference sites (Figure 2, Table 2). Photosynthesis and transpiration did not correlate with any other measured parameters (Table 3). Peroxidase values did not differ significantly between impacted and reference sites (Figure 2, Table 2), but they were negatively correlated with % clay-silt values (Table 3). This correlation, however, was strongly influenced by a single point and was not significant ($r = -0.36$, $P = 0.28$) when this point was omitted. In contrast, glutathione levels were significantly lower at impacted sites (Figure 2, Table 2). Glutathione levels were negatively correlated with microtox toxicity units (wet and dry) (Figure 3, Table 3) suggesting that glutathione and microtox may have been responding to common environmental stressors. These correlations, however, were strongly driven by a single site (Orange Grove) with very high TU levels and very low glutathione levels, and they were not significant (TUwet: $r = -0.46$, $P = 0.16$; TUDry: $r = -0.39$, $P = 0.24$) when this point was omitted.

DISCUSSION

We found few differences between impacted and reference creeks in any of the parameters that we mea-

Table 2. Physical and biological variables (mean \pm 1 SE) broken down by site.

Site	Clay-Silt % n = 1 ¹	Salinity ppt n = 1 ¹	TUwet 100/EC50 n = 3	TUdry 100/EC50 n = 3	Photosynthesis $\mu\text{mol m}^{-2}\text{s}^{-1}$ n = 3	Transpiration $\text{mmol m}^{-2}\text{s}^{-1}$ n = 3	Glutathione pmol mg^{-1} n = 1 ¹	Peroxidase units gm^{-1} n = 1 ¹
Impacted								
Diesel	95.67	28	95.94 \pm 15.45	274.56 \pm 40.58	7.45 \pm 1.04	1.51 \pm 0.25	85.86	14.36
Dill	95.60	25	225.56 \pm 5.49	637.05 \pm 14	14.28 \pm 1.41	0.78 \pm 0.14	86.26	9.04
Koppers	51.90	18	209.56 \pm 98.98	392.59 \pm 168.44	8.89 \pm 2.24	0.79 \pm 0.17	70.72	29.28
New Market	81.95	15	181.66 \pm 188.03	393.94 \pm 255.17	15.01 \pm 1.54	1.53 \pm 0.14	96.64	7.98
Orange Grove	97.97	24	990.58 \pm 171.68	3141.72 \pm 595.09	12.52 \pm 3.42	1.18 \pm 0.18	36.04	9.64
Shipyard	76.60	15	32.52 \pm 5.80	97.91 \pm 14.03	15.41 \pm 2.31	1.76 \pm 0.09	112.98	19.54
Not Impacted								
Battery Simkin	97.73	30	150.44 \pm 10.15	374.33 \pm 22.61	9.97 \pm 3.21	1.49 \pm 0.11	127.74	12.70
Beresford	83.94	20	285.99 \pm 182.45	868.71 \pm 561.83	9.77 \pm 0.60	1.26 \pm 0.07	117.76	16.80
Fosters	93.86	22	60.63 \pm 11.78	181.57 \pm 32.36	16.36 \pm 3.65	1.24 \pm 0.04	130.52	9.80
Horibeck	94.38	20	133.62 \pm 32.33	509.90 \pm 157.68	19.53 \pm 2.69	1.67 \pm 0.13	105.80	9.34
Rathall	92.44	22	134.65 \pm 33.34	504.30 \pm 79.70	11.76 \pm 1.81	2.13 \pm 0.25	85.86	15.12
Rice	96.59	25	52.40 \pm 4.82	154.51 \pm 12.24	13.14 \pm 2.91	2.28 \pm 0.25	130.52	16.96

¹ Composite of 3 pooled samples.

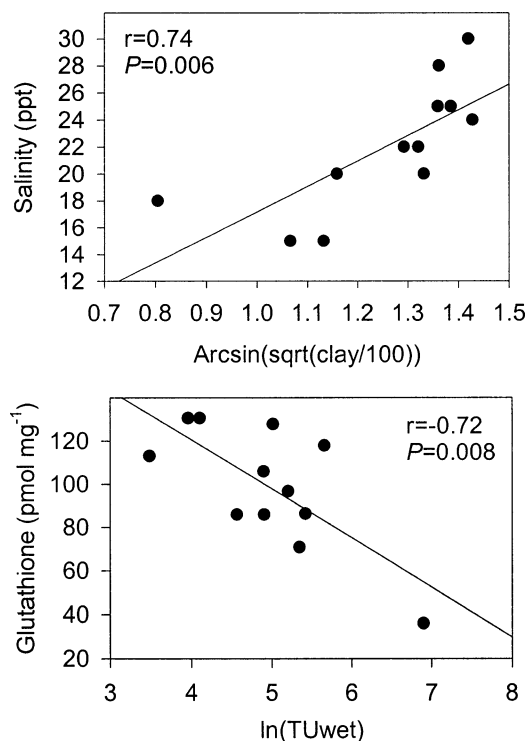


Figure 3. Significant correlations between salinity and %clay-silt (top), and glutathione and TUwet (bottom). Proportional data (% clay-silt) were angular-transformed to improve normality. The relationship between glutathione and TUdry (not shown) was very similar to the bottom plot ($r = -0.70$, $P = 0.01$).

sured, despite large differences between these creeks in levels of pollutants (Sanger *et al.* 1999a,b). Studies of fungal living-biomass (as estimated by ergosterol content) and fungal sexual productivity (as estimated by ascospore expulsion) from standing-dead leaves of *Spartina alterniflora* at two of the sites (Diesel Creek, impacted; Rathall Creek, reference) similarly found no difference between impacted and reference sites (Newell *et al.* 2000). We did find differences in glutathione concentrations between impacted and reference sites, likely because of glutathione's role in conjugating with heavy metals (Mehra *et al.* 1996, Zenk 1996). A previous study by our group at a different salt marsh site (polluted with mercury and PCBs) did not identify glutathione as a useful indicator of pollution impacts (Wall *et al.* 2001), likely because glutathione does not play a role in detoxifying the particular pollutants found at that site. Thus, glutathione is not a universal indicator of plant stress but may be a useful indicator of stress caused by selected pollutants.

A potential weakness of our approach is that we sampled at only one stretch of creekbank at each site and sampled each site on only one date. If pollutants were extremely patchy in distribution along the creeks,

Table 3. Pearson Correlation Coefficients (r and P-values) for all variables correlated across sites.

	Transpiration	Photosynthesis	% Clay-Silt	Salinity	TUwet	TUdry	Glutathione
Peroxidase	-0.06						
	0.85						
Glutathione	0.41	-0.51					
	0.19	0.09					
TUdry	-0.29	0.24	-0.75				
	0.36	0.46	0.005				
TUwet	-0.53	-0.07	0.17	-0.25			
	0.08	0.83	0.60	0.44			
Salinity	0.02	-0.12	0.22	0.04	-0.21		
	0.94	0.71	0.49	0.90	0.51		
%Clay-Silt	0.36	-0.42	0.14	0.13	-0.72	-0.26	
	0.26	0.18	0.66	0.69	0.008	0.41	
Photosynthesis	0.17	0.22	0.74	0.11	0.97	-0.70	-0.04
	0.59	0.49	0.006	0.74	0.0001	0.01	0.90

our sampling approach might have missed ‘hot spots’ of high pollutant concentrations. Although pollutant concentrations varied spatially in the studied creeks (Sanger et al. 1999a,b), the variation was not so high that it should have invalidated our approach. Similarly, if pollutant impacts varied among dates (perhaps interacting with temperature), our sampling approach might have missed dates on which maximal differences were found; however, we have no evidence that such temporal variation occurred. In any case, it was not our goal to conduct an exhaustive study of the impacts of pollutants on salt marsh systems. Rather, our goal was to determine if any of the techniques we used were suitable for rapid assessment of salt marsh health. Techniques that yield dramatically different results depending on small differences in site selection or study date may yield valuable information about spatial and temporal variation in pollutant impacts but are not likely to be robust tools for rapidly assessing the health of multiple marsh sites. In fact, if pollutant impacts are highly patchy in time or space, it is likely that no simple approach will suffice to characterize them.

Using techniques similar to those used here, we previously found few differences in these same bacterial, fungal, and plant indices between a salt marsh in Brunswick, Georgia that had high levels of mercury and PCBs and an adjacent reference site (Newell and Wall 1998, Wall et al. 2001). The repeated failure of these indices to detect differences between sites with and without known high levels of pollutants suggests either that the pollutants have no impact on marsh health (which is unlikely given that the compounds were at high concentrations and have known toxic effects in other systems) or that these indices are not sensitive indicators of marsh health. Our results contrast with those of Lerberg et al. (2000), who worked

at the same creeks around Charleston harbor and found impacts of pollutants on benthic invertebrate abundance and community structure. Similarly, Horne et al. (1999) reported differences in meiofaunal community structure between the Brunswick, GA impacted marsh where we worked previously (Wall et al. 2001) and a reference site. This contrast suggests that benthic invertebrate communities are better indicators of pollution in salt marsh systems than are plants or associated fungi. Benthic invertebrates live intimately associated with sediments, and some taxa feed by passing sediments through their digestive tracts. The chemistry of the digestive tract may be more acidic than that of the marsh environment, which might lead to desorption of some toxic chemicals from sediments, making them more available to invertebrates. As a result, benthic invertebrates are chronically exposed to high levels of pollutants. In contrast, emergent plants and the fungi that decompose senescent leaves are exposed primarily to pollutants in the water column, which are typically present at much lower concentrations than in the sediment, and are exposed only for part of each day when covered by the tide. Plant roots and rhizomes are, of course, continually exposed to pollutants in the sediment; however, roots and rhizomes of salt marsh plants must be adapted to tolerate low redox potentials and high levels of sulfides naturally present in marsh sediments, and these adaptations may confer benefits in terms of tolerating anthropogenic pollutants. In general, previous studies have shown that *Spartina alterniflora* is fairly resistant to a variety of anthropogenic pollutants, including PAHs and crude oil (Lee et al. 1981, Li et al. 1990, Carmen et al. 1996, Lin and Mendelssohn 1996). In contrast, Lewis et al. (2001) argued that toxicity tests using seeds and seedlings of estuarine plants, including *Spartina alterniflora*, may be more sensitive to pollution impacts than toxicity tests

using animals; however, they did not address the utility of studies with mature plant clones nor of studies of *in situ* benthic invertebrate communities.

Finally, the sites studied here and at the Brunswick site studied previously all represent areas that have a long history (several decades) of impacts. It may be that *Spartina alterniflora* and its associated fungal communities have adapted over time to the chemical insults at these sites. In contrast, benthic invertebrates may disperse too widely, either as larvae or with the tides as adults, to adapt to pollutants at such a local scale. If so, assays with naïve organisms transplanted to the sites or exposed to sediments from the sites might reveal larger differences between impacted and reference sites. Our Microtox assays, however, took the latter approach and detected no differences between sites.

In summary, our results presented in this paper and elsewhere (Newell and Wall 1998, Newell *et al.* 2000, Wall *et al.* 2001) suggest that the lower trophic levels of salt marsh food webs (angiosperms and decomposer fungi) are resistant to high levels of pollution and that indices focusing on these trophic levels show little promise for rapidly assessing the health of salt marsh systems. Studies focusing on benthic invertebrates seem to be more sensitive indicators of pollution and should be retained as one of the methods of choice for assessing the health of potentially impacted salt marsh sites. However, although the benthic invertebrate community structure may be altered by pollution, this primarily involves the replacement of pollution-sensitive species with pollution-tolerant species (Horne *et al.* 1999, Lerberg *et al.* 2000) rather than a loss of invertebrate biomass. As a result, little prevents pollutants from rapidly moving up the salt marsh food web to top consumers such as fish and birds, where they can bioaccumulate (Smith and Weis 1997, Maruya and Lee 1998, Horne *et al.* 1999, Burke *et al.* 2000, Newell *et al.* 2000) and pose greater threats to these taxa and to humans.

ACKNOWLEDGMENTS

Financial support of this research was provided by U.S. Environmental Protection Agency award R825147-01-0 and Sea Grant award NA66RG0282. We thank staff of the South Carolina Department of Natural Resources for generously providing access to sites and sharing data. We thank M. Dionne, A. F. Holland, S. Y. Newell, D. M. Sanger, and two anonymous reviewers for helpful comments on the manuscript. This is UGAMI Contribution number 897.

LITERATURE CITED

- Biggs, R. B., T. B. DeMoss, M. M. Carter, and E. L. Beasley. 1989. Susceptibility of U.S. estuaries to pollution. *Aquatic Sciences* 1: 189–207.
- Byl, T. D., H. D. Sutton, and S. J. Klaine. 1994. Evaluation of peroxidase as a biochemical indicator of toxic chemical exposure in the aquatic plant *Hydrilla verticillata* Royale. *Environmental Toxicology and Chemistry* 13:509–515.
- Burke, D. J., J. S. Weis, and P. Weis. 2000. Release of metals by the leaves of the salt marsh grasses *Spartina alterniflora* and *Phragmites australis*. *Estuarine, Coastal and Shelf Science* 51: 153–159.
- Canfield, T. J., N. E. Kemble, W. G. Grumbaugh, F. J. Dwyer, C. G. Ingersoll, and J. F. Fairchild. 1994. Use of benthic invertebrate community structure and the sediment quality triad to evaluate metal-contaminated sediment in the Upper Clark Fork River, Montana. *Environmental Toxicology and Chemistry* 13:1999–2012.
- Carmen, K. R., J. C. Means, and S. C. Pomarico. 1996. Response of a sedimentary bacteria in a Louisiana saltmarsh to contamination by diesel fuel. *Aquatic Microbial Ecology* 10:231–241.
- Carr, R. S., J. W. Williams, and C. T. B. Fragata. 1989. Development and evaluation of a novel marine sediment pore water toxicity test with polychaete *Dinophylus gyrociliatus*. *Environmental Toxicology and Chemistry* 8:533–543.
- DeLaune, R. D. and C. J. Smith. 1985. Release of nutrients and metals following oxidation of freshwater and saline sediment. *Journal of Environmental Quality* 14:164–168.
- DelValls, T. A., L. M. Lubián, J. M. Forja, and A. Gómez-Parra. 1997. Comparative ecotoxicity of interstitial waters in littoral ecosystems using Microtox® and the rotifer *Brachinus plicatilis*. *Environmental Toxicology and Chemistry* 16:2323–2332.
- Gambrell, R. P. 1994. Trace and toxic metals in wetlands—a review. *Journal of Environmental Quality* 23:883–891.
- Griffith, O. W. 1980. Determination of glutathione and glutathione disulfide using glutathione reductase and 2-aminopyridine. *Analytical Biochemistry* 106:207–212.
- Horne, M. T., N. J. Filey, and M. D. Sprenger. 1999. Polychlorinated biphenyl- and mercury-associated alterations on benthic invertebrate community structure in a contaminated salt marsh in southeast Georgia. *Archives of Environmental Contamination and Toxicology* 37:317–325.
- Kato, M. and S. Shimizu. 1985. Chlorophyll metabolism in higher plants. VI. Involvement of peroxidase in chlorophyll degradation. *Plant Cell Physiology* 26:1291–1301.
- Klemm, D. J., C. J. Strobel, L. B. Lobring, J. W. Eichelberger, and A. Alford-Stevens. 1993. Environmental Monitoring and Assessment Program (EMAP) Laboratory Methods Manual Estuaries. U.S. Environmental Protection Agency, Cincinnati, OH, USA. EPA/620/R-93/xxx (draft).
- Lamberson, J. O., T. H. DeWitt, and R. C. Swartz. 1992. Assessment of sediment toxicity to marine benthos. p. 183–240. *In* G. A. Burton, Jr. (ed.) *Sediment Toxicity Assessment*. Lewis Publishers, Boca Raton, FL, USA.
- Lee, R. F., B. Dornseif, F. Gonsoulin, K. Tenore, and R. Hanson. 1981. Fate and effects of a heavy fuel oil spill on a Georgia salt-marsh. *Marine Environmental Research* 5:125–143.
- Lerberg, S. B., A. F. Holland, and D. M. Sanger. 2000. Responses of tidal creek macrobenthic communities to the effects of watershed development. *Estuaries* 23:838–853.
- Lewis, M. A., D. E. Weber, R. S. Stanley, and J. C. Moore. 2001. The relevance of rooted vascular plants as indicators of estuarine sediment quality. *Archives of Environmental Contamination and Toxicology* 40:25–34.
- Li, Y., J. T. Morris, and D. C. Yoch. 1990. Chronic low level hydrocarbon amendments stimulate plant growth and microbial activity in saltmarsh microcosms. *Journal of Applied Ecology* 27: 159–171.
- Lin, Q. and I. A. Mendelssohn. 1996. A comparative investigation of the effects of south Louisiana crude oil on the vegetation of fresh, brackish and salt marshes. *Marine Pollution Bulletin* 32: 202–209.

- Long, E. R. 2000. Degraded sediment quality in U.S. estuaries: a review of magnitude and ecological implications. *Ecological Applications* 10:338–349.
- Luoma, S. N. and K. T. Ho. 1992. The appropriate uses of marine and estuarine sediment bioassays. p. 193–226. *In* P. Callow (ed.) *The Handbook of Ecotoxicology*, Vol. 1. Blackwell Scientific, Cambridge, MA, USA.
- Maruya, K. A. and R. F. Lee. 1998. Aroclor 1268 and toxaphene in fish from a southeastern U.S. estuary. *Environmental Science and Technology* 32:1069–1075.
- Mehra, R. K., J. Miclat, V. R. Kodati, R. H. T. C. Abdullah, and P. Mulchandani. 1996. Optical spectroscopic and reverse-phase HPLC analyses of Hg(II) binding to phytochelatins. *Biochemistry Journal* 314:73–82.
- Microbics Corp. 1992. *Microtox® Manual*, a toxicity testing handbook. Microbics Corp., Carlsbad, CA, USA.
- Microbics Corp. 1995. *Microtox® Manual*. *Microtox® Acute Toxicity Solid-Phase Test*. Microbics Corp., Carlsbad, CA, USA.
- Newell, S. Y. and V. D. Wall. 1998. Response of saltmarsh fungi to the presence of mercury and polychlorinated biphenyls at a Superfund site. *Mycologia* 90:777–784.
- Newell, S. Y., V. D. Wall, and K. A. Maruya. 2000. Fungal biomass in saltmarsh grass blades at two contaminated sites. *Archives of Environmental Contamination and Toxicology* 38:268–273.
- Olsen, R., N. H. Cutshall, and I. L. Larsen. 1982. Pollutant-particle associations and dynamics in coastal marine environments: a review. *Marine Chemistry* 11:501–533.
- Ringwood, A. H., M. E. DeLorenzo, P. E. Ross, and F. Holland. 1997. Interpretation of *Microtox®* solid phase toxicity tests: Effects of sediment composition. *Environmental Toxicology and Chemistry* 16:1135–1140.
- Sanger, D. M., A. F. Holland, and G. I. Scott. 1999a. Tidal creek and salt marsh sediments in South Carolina coastal estuaries: II. Distribution of organic contaminants. *Archives of Environmental Contamination and Toxicology* 37:458–471.
- Sanger, D. M., A. F. Holland, and G. I. Scott. 1999b. Tidal creek and salt marsh sediments in South Carolina coastal estuaries. I. Distribution of trace metals. *Archives of Environmental Contamination and Toxicology* 37:445–457.
- Siegel, B. Z. 1993. Plant peroxidases—an organismic perspective. *Plant Growth Regulation* 12:303–312.
- Simpson, R. L. and R. E. Good. 1985. The role of tidal wetlands in the retention of heavy metals. p. 164–175. *In* H. A. Groman, T. R. Henderson, E. J. Meyers, D. M. Burke, and J. A. Kusler (eds.) *Proceedings of the Conference—Wetlands of the Chesapeake*. Environmental Law Institute, Washington, DC, USA.
- Smith, G. M. and J. S. Weis. 1997. Predator-prey relationships in mummichogs (*Fundulus heteroclitus* (L.)): effects of living in a polluted environment. *Journal of Experimental Marine Biology and Ecology* 209:75–87.
- Strobel, C. J., D. J. Klemm, L. B. Lobring, J. W. Eichelberger, and A. Alford-Stevens. 1995. Environmental monitoring and assessment program (EMAP) Laboratory Methods Manual Estuaries. Vol. 1. Biological and Physical analyses. U.S. Environmental Protection Agency, Cincinnati, OH, USA. EPA/620/R-95/008.
- Swartz, R. 1989. Marine sediment toxicity tests. p. 115–129. *In* *Contaminated Marine Sediments—Assessment and Remediation*. National Research Council, National Academy Press, Washington, DC, USA.
- Traunspurger, W. and C. Drews. 1996. Toxicity analysis of freshwater and marine sediments with meio- and macrobenthic organisms: a review. *Hydrobiologia* 328:215–226.
- Wall, V. D., J. J. Alberts, D. J. Moore, S. Y. Newell, M. Pattanayek, and S. C. Pennings. 2001. The effect of mercury and PCBs on organisms from lower trophic levels of a Georgia salt marsh. *Archives of Environmental Contamination and Toxicology* 40:10–17.
- Zenk, M. H. 1996. Heavy metal detoxification in higher plants—a review. *Gene* 179:21–30.

Manuscript received 20 July 2001; revisions received 25 January 2002; accepted 1 March 2002.