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NSY PORTSMOUTH
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DEPARTMENT OF THE NAVY
NORTHERN DIVISION
NAVAL FACILITIES ENGINEERING COMMAND
10 INDUSTRIAL HIGHWAY
MAIL STOP, #82
LESTER, PA 19113-2090

IN REPLY REFER TO

5090
Code 1823/FE

OCT 0 9 1997

Ms. Meghan Cassidy
U.S. Environmental Protection Agency, Region I
JFK Federal Building HBT
Boston, MA 02203-2211

Mr. Iver McLeod
Maine Department of Environmental Protection
State House Station 17
Augusta, ME 04333-0017

Subj: NOVEMBER 1997 SEEP/SEDIMENT SAMPLING FOR PORTSMOUTH NAVAL SHIPYARD, KITTERY, ME

Dear Ms. Cassidy/ Mr. McLeod:

As discussed at our July 24, 1997 technical meeting, the Navy proposed collecting and analyzing the sediment porewater for metals. In order to obtain the porewater/sediment pair for development of site specific Kds, the Navy proposes modifying the existing sediment sampling protocols. Enclosed is a brief explanation of the reasons for the changes and the proposed procedures.

Please provide any comments on the enclosed procedures by October 21, 1997.

For the Community Restoration Advisory Board (RAB) members; if you have any comments or questions on these issues, they can be provided to the Navy at a RAB meeting, by calling the Public Affairs Office at (207) 438-1140 or by writing to:

Portsmouth Naval Shipyard
Code 121.10 Bldg 44
Attn Marty Raymond
Portsmouth, NH 03804-5000

Subj: NOVEMBER 1997 SEEP/SEDIMENT SAMPLING FOR PORTSMOUTH NAVAL SHIPYARD, KITTERY, ME

If additional information is required, please contact Ms. Marty Raymond at 207-438-2536 or myself at 610-595-0567, x159.

Sincerely,

FREDERICK J. EVANS, P.E.
Remedial Project Manager
By direction of the
Commanding Officer

Copy to:
NOAA (K. Finkelstein)
US Fish & Wildlife Service (K. Munney)
ME Dept. of Marine Resources (D. Card)
NH Fish & Game (J. Nelson)
PNS (Code 106.3R, M. Raymond)
Ms. Juanita Bell
Mr. Jeff Clifford
Ms. Eileen Foley
Mr. Phil McCarthy
Mr. Guy Petty
Mr. Peter Vandermark
PNS (Code 100PAO) w/o encl
COMSUBGRU TWO (R. Jones)
Mr. Doug Bogen
Ms. Michele Dionne
Ms. Mary Marshall
Mr. Jack McKenna
Mr. Onil Roy
Ms. Carólyn Lepage
Brown and Root Environmental (L. Klink, B. Horne)

DATE: September 18, 1997

SUBJECT: PROPOSED FIELD AND LABORATORY PROTOCOLS TO DEVELOP SITE SPECIFIC KD VALUES FOR SEDIMENT AT PORTSMOUTH SHIPYARD

BACKGROUND: As part of the November 1997 seep & sediment sampling effort scheduled at Portsmouth Naval Shipyard , the Navy is attempting to collect data which will be used to develop site specific sediment partitioning coefficients (Kds) for metals in the nearshore areas. The locations for sampling will be in the same areas where previous seep and sediment samples were obtained from three earlier events (December 1996, April 1997, and August 1997). It should be noted that the protocols which are being proposed for Portsmouth Naval Shipyard were utilized in recent studies conducted by the Navy at NPL sites adjacent to Narragansett Bay in Rhode Island (NETC-Newport & NCBC-Davisville). At these two sites, numerous sediment/porewater samples were collected for chemical analysis as part of an ecological risk assessment.

Because the sampling is scheduled to occur soon, the Navy is requesting a rapid review of the proposed protocols so that equipment can be procured in time to conduct sampling. As a result, It is hoped that any significant issues can be brought to the Navy's attention by October 15, 1997. If you have any questions or concerns regarding these SOPs, please contact Fred Evans at 610-595-0567 ext 159.

PROPOSED PROTOCOLS

In order to obtain the porewater/sediment pair which is most appropriate for the development of site specific modeling Kds, the Navy is proposing a modification to the existing sediment sampling protocols. For the last three rounds of sampling (December 1996, April 1997, and August 1997), sediment has been collected from the top several centimeters from areas where visible seeps discharged to the nearshore environment. This was intended to correlate seep chemistry to adjacent surficial sediment. Because the groundwater flow model assumes significant discharge beneath the surface of the sediment, more useful information could be obtained from a sample core which would provide data on the subsurface chemistry and porewater and hence a more appropriate Kd for metals. The cores would only be possible at certain locations where there is good recovery (e.g. the back channel) based on the cohesiveness of the sediment. The goal would be to obtain cores that are one foot or more in depth where possible.(see SOPS in Appendix for details on core sample collections protocols]. The enclosed SOPs for sediment sample collection indicate a number of possible methods for acquisition of cores based on depth of water and other site conditions. The Navy is proposing to use the "Drop-hammer method" where possible.

For sediment samples which cannot be collected by coring , URI personnel will obtain sediment samples using acid washed, teflon coated, titanium sampling tools. Samples will be collected from the top several centimeters of sediment and carefully placed in one liter wide mouthed acid washed plastic containers so as to minimize disturbance to the sediment-pore water matrix. (Significant disturbance to this matrix may artificially elevate the concentration of metals in the porewater). After Collection, the samples will be placed in coolers on "blue ice" at 4 degrees C until fieldwork is completed for the day. Each day, the sediment samples that are used for porewater will either be transported by truck or shipped by overnight carrier to the laboratory in Narragansett, RI. Preparation of the porewater extracts will be done within 72 hours of field collection according to the enclosed SOP. Care shall be taken to insert the airstone 8 to 10 cm into the sediment sample to avoid collection of surface water that may have accumulated in the container. SOP's for the extraction of porewater samples from the sediment collected are provided in the Appendix along with the referenced article by *Winger and Lasier*.

POINT OF CONTACT:

John W. King
URI/Graduate School of Oceanography
South Ferry Rd.
Narragansett, RI 02882-1197

I. OBJECTIVE

Collection of subtidal sediments (cores and bulk) from research vessels for chemical and toxicological analysis.

II. NECESSARY MATERIALS AND EQUIPMENT

The materials and equipment required or available for sediment sampling may vary depending on the vessel used for sampling. The investigator should determine the deployment capabilities of the vessel to be used and the equipment available prior to planning the sampling event. The Smith-MacIntyre or Van-Veen grab may be deployed from a vessel with suitable winches, and an A-Frame or Davit arrangement. The Nemesis is best operated with an A-frame. The freeze corer and standard piston corer are best operated from the URI pontoon boat with a removable hatchway in the mid-ship area.

Additional equipment needed:

- Drop-Hammer Corer
- Freeze corer 2.0 m long, 20 cm width
- Nemesis 1.3 m long 7.5 cm diameter piston corer
- standard piston corer 1m long, 10 cm diameter
- 3 m long extension rods for standard piston corer
- Smith-MacIntyre (0.1 m) grab sampler
- Stand - for the grab
- Lead weights - 4 lead weights for Smith-Mac
- Cocking bar for Smith-Mac
- Teflon Coated Van-Veen Grab
- Hard hats
- Face shield or other eye protection
- Respirator - if conditions dictate
- Standard safety equipment (i.e. - first aid kit)
- Tub - into which to dump sediment
- Titanium scoop and titanium sampling tools
- Buckets (2-3) - for rinsing
- Electrical tape
- Duct tape
- Tools - screw drivers, wrenches, hammer, other
- Sample containers - cleaned and acid stripped
- Coolers with ice, or other means of cold storage
- Water-proof sample notebook
- Water-proof markers
- Suitable protective clothing (from weather as well as contaminants)

- Foul weather gear
- Larger pipe cutter
- Boots
- Exposure suits (orange worksuits) for winter sampling
- Gloves - appropriate gloves for suspected conditions
- Differential GPS for navigation
- Line and stainless cable
- Twine/pocket knife
- Core caps
- Polycarbonate core tubes for piston cores
- Vertical extension box for sampling at 1 cm (or greater) intervals
- Methanol
- Dry ice

Whenever possible, back-up equipment should be carried on board.

III. METHODS

1. The top 1-2 cm of an undisturbed grab sample is sampled using a clean titanium scoop for chemistry and toxicity analysis. Approximately 2-3 Van-Veen or 3-5 Smith Mac grabs are needed to collect a 3-liter composite sample. The surface material is composited in a 12-liter Nalgene bucket, stirred with a titanium stirrer for ~30 seconds, and then subsampled into precleaned containers for organic, inorganic chemistry, and toxicity studies. Rinse the scoop with distilled water, acid, and methanol between grabs. The grab sampler should be "washed-down" with seawater between stations. The samples are stored on ice during collection and at -20°C upon return to the laboratory.

2. Cores are typically taken one of four ways:

   a. Taking a short core from an undisturbed grab sample:

      1) Insert "small" (15 cm) cores into the middle of an undisturbed grab sample.

      2) Place a cap on the top of the core and remove the core from the grab.

      3) Place the second cap on the core after removal from the grab.

      4) All caps and liners are precleaned with acid, methanol, and DI. Liners are capped until used.

   b. Long piston cores:

      Piston cores are used to take deeper (~1 m) inch cores. A standard piston corer, the biological corer, is used to retrieve cores from intermediate water depths (<20 m). The corer uses polycarbonate tubes and is deployed using a series of 3 meter long extension rods to push the corer into the sediment. Cores up to one meter long are recovered using this design.

Biological cores are stored in the vertical position and are transported to the lab for further analysis.

A trigger release piston corer, the Nemesis, is used in deeper water (>10 m). The landing pad design has lead weights loaded at the top and triggers upon contact with the sediment/water interface. The polycarbonate core barrel is then forced into the sediment to recover 1 to 1.3 meter cores from any water depth. The Nemesis cores are stored vertically and transported to the lab for analysis. Both types of piston are sampled by using a vertical extrusion box that extrudes the core out of the top of the core tube in 1 cm (or greater) increments without disturbing the core.

c. Freeze cores:

A freeze corer is used to retrieve undisturbed sediments even in flocculent or gaseous sediments. This corer is a two-meter long, stainless steel rectangular box lined with plywood that functions as insulation. The box is filled with a combination of one part methanol and three parts dry ice and creates a freezing surface to which the sediment adheres. The corer is lowered through the water column and then pushed into the sediment column where it remains for 15-20 minutes. During this interval a thickness of 1-4 cm of sediment freezes to the exterior of the box. Frozen slab cores are recovered at lengths from 0.5 to one meter. Cores were wrapped in foil and kept frozen for analysis in the lab. Cores are sectioned using a titanium "hot wire" cutting device in increments of 1 cm (or greater).

d. Drop-hammer cores:

A drop-hammer corer is used to retrieve undisturbed sediments in intertidal or very shallow water environments. This corer has a polycarbonate barrel, a stainless steel cutting shoe and core catcher. The core barrel is pounded into the sediment to the desired depth or to refusal. The top of the core barrel is cut off with the pipe cutter and capped and taped. A shovel is used to dig down next to the core barrel until the bottom can be capped. The core is retrieved, measured for length, and transported on ice.

IV. TROUBLESHOOTING

When using the Smith-MacIntyre grab in very soft sediments, it may be necessary to attach plywood "shoes" to the landing pads. This is necessary to increase the sensitivity of the "firing" mechanism.

V. STATISTICAL ANALYSIS AND DATA USAGE

Not applicable.

VI. REFERENCES

None.

Point of Contact:
Cornelia Mueller
Greg Tracey
Science Applications International Corporation
165 Dean Knauss Drive
Narragansett, RI 02882

1. OBJECTIVE

This document describes the procedures used to extract pore-water from estuarine and freshwater sediments using a vacuum-operated system following methods described in Winger and Lasier (1991). Actual volumes needed for analyses and biological testing and holding requirements are task-specific and require consultation with the Project Manager.

2. SAFETY

Sediments may be contaminated with hazardous biological or chemical constituents. Proper attire should be worn.

3. MATERIALS

Fused-glass air stone
Hose clamp
Medical grade silastic tubing (3/16" I.D.)
30 or 60 cc polypropylene syringe with catheter tip
Piece of wood cut to fit between the end of the syringe and the lip of the plunger
0.45 µm filter unit
50-ml centrifuge tube
Deionized water in squirt bottle

4. METHODS

4.1 Label the syringes and 50-ml centrifuge tubes with data transcribed from the sample container.

4.2 Use tubing to attach fused-glass air stone to the syringe.

4.3 Insert air stone 8 to 10 cm into the sediment sample.

4.4 Secure the tubing to container with elastic bands.

4.5 Attach hose clamp to the tubing.

4.6 Create a vacuum by loosening the hose clamp retracting the plunger of the syringe.

4.7 Brace the plunger in the retracted position by inserting a piece of wood between the end of the syringe and the lip of the plunger.

4.8 Collect 2 to 5 ml of pore-water.

4.9 Maintain vacuum by tightening hose clamp.

4.10 Remove the brace.

4.11 Remove the syringe.

4.12 Discard pore-water.

4.13 Rinse syringe and plunger with deionized water from squirt bottle.

4.14 Reassemble and reattach syringe to tubing.

4.15 Reestablish the brace.

4.16 Loosen the hose clamp.

4.17 Fill the syringe. For fine-grained sediments, 12 - 18 hours may be required. Store sample in at 4°C in the dark during extraction.

4.18 Remove the airline tubing from the syringe when the syringe is full.

4.19 Examine pore-water for turbidity. Filter if necessary. Multiple collections require homogenization before filtration.

4.20 Dispense the pore-water from the syringe or the filter unit into a pre-labeled 50-ml centrifuge tube. Multiple collections require homogenization before dispensation into 50-ml tubes.

4.21 Store sample at 4°C in the dark for further analyses.

4.22 Sediments are discarded in waste barrels for disposal. Barrels are sampled and analyzed to establish proper disposal procedures. Disposal procedures are performed in accordance with local, state, and federal disposal regulations.

5.0 QA/QC

Any deviations from this SOP are documented and reported to the Program Manager.

6.0 REFERENCES

Winger, PV and PJ Lasier. 1991. A Vacuum-Operated Pore-Water Extractor for Estuarine and Freshwater Sediments. Arch. Environ. Contam. Toxicol. 21, 321-324.

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Arch. Environ. Contam. Toxicol. 21, 321–324 (1991)

Archives of Environmental Contamination and Toxicology

A Vacuum-Operated Pore-Water Extractor for Estuarine and Freshwater Sediments

Parley V. Winger and Peter J. Lasier

U.S. Fish and Wildlife Service, National Fisheries Contaminant Research Center, University of Georgia, School of Forest Resources, Athens, Georgia 30602, USA

Abstract. A vacuum-operated pore-water extractor for estuarine and freshwater sediments was developed and constructed from a fused-glass air stone attached with aquarium airline tubing to a 30 or 60 cc polypropylene syringe. Pore water is extracted by inserting the air stone into the sediment and creating a vacuum by retracting and bracing the syringe plunger. A hand-operated vacuum pump attached to a filtration flask was also evaluated as an alternative vacuum source. The volume and time to extract pore water varies with the number of devices and the sediment particle size. Extraction time is longer for fine sediments than for sandy sediments. Four liters of sediment generally yield between 500 and 1,500 mL of pore water. The sediment that surrounds and accumulates on the air stone acts as a filter, and, except for the first few milliliters, the collected pore water is clear. Because there is no exposure to air or avenue for escape, volatile compounds and *in situ* characteristics are retained in the extracted pore water.

A major thrust in current environmental toxicology is the evaluation of contaminants in sediments, particularly, in pore (interstitial) water. Most contaminants that enter aquatic systems are ultimately deposited in the bottom sediments (Salomons *et al.* 1987). The processes of sediment transport and deposition transfer pollutants from the water column into the bottom sediments, but the sediments also provide a source of contaminants to the surface waters (Davies-Colley *et al.* 1985; Salomons 1985). Consequently, the effects of contaminants in sediments on the environment must be identified for management and protection of aquatic resources.

To have a toxic effect on aquatic organisms, contaminants must be biologically available. In aquatic systems, the major route of exposure is solution. However, to elicit a toxic response, contaminants in sediments have to be sufficiently high for the equilibrium-partitioning concentration in pore water to exceed toxic levels (Adams *et al.* 1985; O'Donnel *et al.* 1985). Assessing the toxicity of pore water directly is a logical step in evaluating sediment and bioavailability of contaminants. In fact, equilibrium-partitioning of contaminants in pore water is a potential means of assessing sediment quality (Shea 1988). Unfortunately, techniques for isolating or collecting pore water for chemical analyses or toxicity testing require elaborate equipment and may alter the chemical characteristics of the sediment and pore water (Bender *et al.* 1987; Knezovich and Harrison 1988). In addition, most techniques do not produce enough pore water for standard toxicity testing.

A variety of methods have been used to extract pore water. Most early investigators were concerned with general chemistry of sandy intertidal and shoreline areas, and pore water was obtained by draining the sediment (Bruce 1928). Other methods of pore-water extraction have been developed and include various filtering devices (Borden 1931; Cole 1932; Pennak 1940; Ishida 1953; Jones 1955; Eriksen 1963; Brafield 1964; Saager *et al.* 1990), mechanical squeezers (Krnikov and Komarova 1954; Luszczynski 1961; Siever 1962; Hartman 1965; Manheim 1966; Reeburgh 1967; Presley *et al.* 1967; Bender *et al.* 1987; Jahnke 1988; Carr *et al.* 1989), and dialysis (Hesslein 1976; Carignan *et al.* 1985). Centrifugation and elutriate preparations are also commonly used for collecting water for the assessment of contaminants and toxicity in the liquid fraction of sediments (Shuba *et al.* 1977; Dutka and Kwon 1988; Geisy *et al.* 1988; Ankley *et al.* 1990; Saager *et al.* 1990). Many described methods for collecting pore water require extensive sample manipulations in addition to filtration or centrifugation prior to testing or analysis. However, vacuum-operated devices (Johnson 1967; Creaser 1971; Makemson 1972; Sayles *et al.* 1976; Knezovich *et al.* 1987; Whitman 1989) have the potential to alleviate many of the problems associated with sediment sample manipulation and turbidity of extracted pore water. The vacuum-operated pore-water extractor described in this paper is inexpensive, easily constructed, and effective for sediments of all particle sizes. It provides sufficient quantities of pore water for toxicity testing and chemical analyses, and extracts do not require filtration or centrifugation to clear the pore water prior to toxicity testing or analyses.

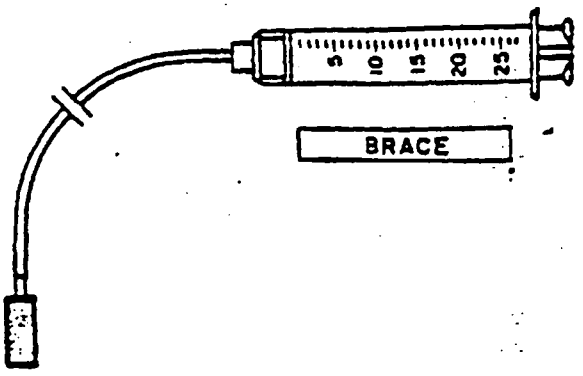

Fig. 1. Sediment pore-water extractor constructed from a syringe, brace, tubing, and an air stone

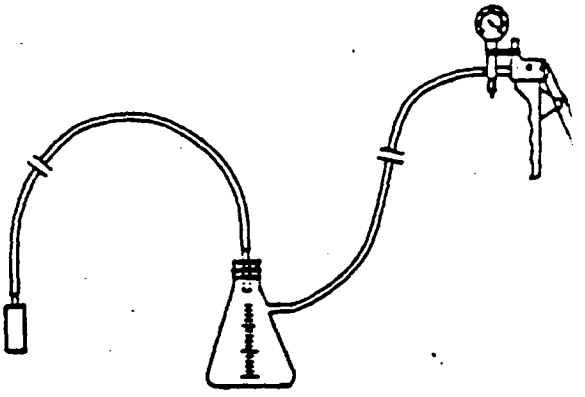

Fig. 2. Sediment pore-water extractor constructed from a hand operated vacuum pump, tubing, filtration flask, and an air stone

Materials and Methods

The pore-water extractor that was developed consists of a fused-glass air stone attached by aquarium airline tubing to a 30 or 60 cc (other size syringes can be used) polypropylene syringe (Figure 1). The syringe provides the vacuum and also serves as the collecting chamber for the pore water. An alternate vacuum source is a commercial hand-operated vacuum pump. The hand-operated pump is attached with tubing to an Erlenmeyer or filtration flask which serves as the collecting chamber (Figure 2).

To extract pore water, the air stone was inserted 8 to 10 cm into the sediment sample and vacuum created by retracting the plunger of the syringe. The plunger is braced in the retracted position by inserting a piece of wood, cut to the proper length, between the end of the syringe and the plunger lip. When full, the airline tubing is removed from the syringe and the syringe is either capped for storage or emptied and reset for another extraction.

To test the device, pore water was extracted from the sediment samples from various freshwater and estuarine habitats in the Southeast (Table 1). Sediment samples were collected with either an Ekman or Ponar grab.

Sediment particle size was determined by wet sieving with U.S. Standard Sieves (500 μm, 250 μm, 125 μm, and 63 μm) and a 1.2 μm glass-fiber filter. Mean particle size was calculated by Inman's (1952) method; the resulting phi values were converted from $-\log_2$ back to metric units. An estimate of percent organic matter was determined by loss on ignition at 600°C for 4 h. Moisture content was determined as the percent difference between wet and oven-dried (105°C for 24 h) weight.

Results and Discussion

The vacuum-operated extractor collected pore water from bottom sediments at rates from 11 to >100,000 mL/h (Table 1). Extraction rates varied with sediment particle size and were lowest for sediments with the highest percentages of fines and smallest mean particle sizes. The amount of organic matter or percent moisture in the sediment did not influence extraction rates. Extraction of pore water from sandy sediments (mean particle size >140 μm) was very rapid—the syringes filled almost immediately when the vacuum was applied.

The required volume and time to extract pore water varies with the number of devices used and the sediment particle size. About 150 mL of pore water/h can be extracted fro most sediments with six 30-cc syringes or two-hand operai vacuum pumps. Four liters of sediment generally yield b tween 500 and 1,500 mL of pore water, but sediments wi large particle sizes (sand) do not contain as much pore wat as sediments with finer particles.

The first few milliliters of extracted pore water may b turbid. This water is discarded by first bending the airlir tubing, which retains and maintains the vacuum at the s stone, and then releasing the vacuum at the syringe by pul ing the tubing from the end of the syringe. After dispensir the turbid pore water, the airline tubing is re-affixed to th syringe. The bend in the tubing is then released and a fres vacuum applied by retracting and bracing the syring plunger.

The initial turbidity in the extracted pore water from som sediment samples apparently occurs before the finer sedi ment particles have accumulated on the surface of the ai stone. The sediment that surrounds and accumulates on th air stone serves as a filter and screens the particulate matter After discarding the first few milliliters of turbid sample, th rest of the collected pore water is generally clear. Mitchell e al. (1989) also found that pore water extracted with a vac uum-operated device became clear after the initial sample An exception is sandy sediments (larger particle sizes)— larger volumes need to be extracted before the pore wate becomes clear.

Pore-water extractors that drastically disturb the sedi ments (causing aeration and change in redox potential) anc also require filtration or centrifugation may not be ade quately sampling the dissolved fraction (or the actual con stituents) of contaminants in the pore water (Knezovich e al. 1987; Puls and Barcelona 1989), although there is no sat isfactory method for determining unambiguously the parti tioning of contaminants in sediments (Tessier and Campbel 1987). Major problems of most pore-water extractors includ contamination of pore water with overlying water and solid solution reactions. Contaminants in elutriate or pore wate rapidly partition back onto the underlying sediments follow ing squeezing or centrifugation (Bender et al. 1987). Chang ing from anoxic to oxic conditions in the sediments cou also alter the state (species) of contaminants in the p

Table 1. Extraction rate (mL/h) for the vacuum-operated pore-water extractor from sediments characterized by percent moisture, percent organic matter, and sediment particle size

Station	Rate (mL/h)	Organic matter (%)	Moisture (%)	Percent composition by particle size (μm) 250	125	63	1	Mean size (μm)
Charleston Harbor	18	14.4	72	6.6	16.0	9.9	67.5	21
Savannah Harbor 1	27	15.7	78	1.1	5.3	5.5	88.1	10
Savannah Harbor 2	27	18.2	81	0.4	1.0	13.6	85.0	12
S. Thurmond Lake	25	6.5	41	13.6	38.9	16.4	31.1	5
St. Simons Ext. 1	22	16.7	77	10.7	15.3	11.6	62.4	24
St. Simons Ext. 2	15	12.8	68	6.1	41.6	9.4	42.9	31
St. Andrews Bay 1	35	15.3	69	7.9	21.3	24.0	46.8	28
St. Andrews Bay 2	49	19.5	75	8.5	27.2	15.2	49.1	28
St. Andrews Bay 3	35	9.2	61	22.3	33.9	13.3	28.3	51
Perdido River	108,000	0.2	24	96.5	2.8	0.1	0.3	380
Galveston Bay 1	11	6.4	53	4.2	30.0	18.4	47.4	27
Galveston Bay 2	12	2.6	34	1.8	44.8	26.0	27.4	48
Galveston Bay 3	6,750	3.0	27	2.5	67.4	20.6	20.6	140

water. Salomons *et al.* (1987) indicated that filtration of anoxic sediments (causing a shift to the oxic condition) could possibly cause iron oxidation and colloidal formation with subsequent sorption of previously dissolved contaminants. Consequently, not only could filtering cause aeration and loss of dissolved species, but the progressive clogging of the filter with the colloids results in a changing of the generally accepted level of separation between dissolved and particulate matter (Salomons *et al.* 1987).

Volatile compounds of sediments are retained in the pore-water extract or in the sampler described, because the system is enclosed and no avenue of escape exists. Knezovich and Harrison (1987, 1988) showed that a vacuum-operated device similar to the one described here was effective in collecting volatiles from pore water. The ability of a sampler to retain these fractions may be particularly important for aquatic sediments contaminated with volatile compounds. Although this was not tried, a carbon trap could be included in the device to collect volatile compounds as they are extracted with the pore water.

The extractor with the hand-operated vacuum pump was just as effective in obtaining pore water as the syringe system. This system was, however, more expensive and more cumbersome to use than the syringe. Attachment of a carbon trap for capture of volatile compounds would probably be easier to install on the hand-operated system than on the syringe extractor—a carbon trap could be inserted into the tubing that connects the filtration flask to the hand-operated vacuum pump.

The vacuum-operated extractor described here can be used to collect pore water *in situ* or from sediment samples in the field, thereby alleviating the need to return large quantities of sediment to the laboratory. We prefer extracting pore water from consolidated sediment samples collected with a Ponar grab. The extracted pore water is essentially the same as it is *in situ*, because exposure to oxidizing conditions is limited to the external surfaces of the sediment sample. Pore water was also extracted from sediment samples while they were still in an Ekman grab—lift the top flaps of the sampler, decant the surface water, and insert an air

Glass syringes and teflon tubing are more expensive options if there is concern about possible adsorption of organic contaminants to the walls of the extractor. However, the small amount of contaminants adsorbed to the extractor will probably not reduce or alter the toxicity of the pore water or be detectable analytically in light of the natural variability inherent in field samples.

The vacuum-operated pore-water extractor described in this paper provides an inexpensive, but highly versatile and easy device for collecting pore water for toxicity testing and chemical analyses. The major advantages of this method are: large volumes of pore water can be collected within a reasonable amount of time; it is effective for all types of aquatic sediment samples; it eliminates the need for extensive sample manipulation and processing; and pore water can be extracted from essentially undisturbed sediments, thereby minimizing activities that could alter the *in-situ* chemical characteristics of the pore water.

Acknowledgments. We thank S. Carr, M. Brim, H. Geiner, and R. Jarvis for assisting in the collection of sediment samples.

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