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ORIGINAL PAPER

Non-random sampling and its role in habitat conservation: a comparison of three wetland macrophyte sampling protocols

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Abstract Aquatic macrophytes provide essential spawning and nursery habitat for fish, valuable food source for waterfowl, migratory birds and mammals, and contribute greatly to overall biodiversity of coastal marshes of the Laurentian Great Lakes. Two approaches have been used to survey the plant community in coastal wetlands, and these include the grid (GR) and transect (TR) methods. These methods have been used to identify the average species richness at different sites, but their suitability for determining total species richness of a site has not been tested. In this paper, we compare the performance of these two established methods with that of the Stratified method (ST), which uses the sampler's judgment to guide them to different habitat zones within the wetland. We used the three protocols to compare species richness of six coastal wetlands of the Great Lakes, three pristine marshes in eastern Georgian Bay (Lake Huron) and three degraded wetlands in Lake Ontario, Canada. The greatest species richness was associated with the ST method, irrespective of wetland quality. The ST method was also more efficient (fewer quadrats sampled), and revealed the most number of unique (those found with only one method) and uncommon species (those found in <5% of the quadrats). Despite these statistical differences, we found that sampling method did not significantly affect the performance of a recently developed index of wetland quality, the Wetland Macrophyte Index. These results have important implications for designing macrophyte surveys to track changes in biodiversity and wetland quality.

Keywords Macrophyte · Protocol · Coastal wetlands · Conservation · Biodiversity · Species richness

Introduction

Coastal wetlands play an important role in the biological, chemical and physical cycles of aquatic ecosystems (Carpenter 1981). They are situated at the ecotone between the

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terrestrial and aquatic environments and thus provide important habitat for both aquatic and terrestrial organisms (Mitsch and Gosselink 2000). Within the Laurentian Great Lakes ecosystem, coastal wetlands provide critical habitat and a source of food for fish (Jude and Pappas 1992), amphibians, reptiles, and migratory birds (Chow-Fraser and Albert 1999; Maynard and Wilcox 1997).

Many of the important ecosystem services that wetlands provide are accomplished by the macrophytes, and these have been summarized by Cronk and Fennessy (2001). First, they act as filters to trap sediment and nutrients from terrestrial runoff and prevent them from reaching open water. Secondly, they play a vital role in the biogeochemical cycling of nutrients in wetlands where they act as both nutrient sinks and nutrient pumps. Thirdly, macrophytes help reduce shoreline damage from wind and wave action by stabilizing the sediment with their root structure. Fourthly, the physical structure of aquatic macrophytes provides essential habitat for fish and macroinvertebrates, and macrophyte species richness can be directly related to the species diversity of upper trophic levels.

Coastal wetlands face many threats from human development, and since the arrival of European settlers we have either directly or indirectly caused the destruction of 60–80% of coastal wetlands in the Great Lakes, especially in Lakes Erie and Ontario (Ball et al. 2003; Smith et al. 1991). Coastal wetlands are generally found in areas protected from wind and waves of the open lake, and for these same reasons, they are desirable for human development. Wetlands are filled in for condo and cottage development, dredged for marinas and channels and managed for recreational use (beaches and shallow areas are mowed or raked to remove aquatic vegetation). Because of the ongoing threats that wetlands face, it is important to have the appropriate tools to facilitate protection and conservation of our remaining coastal wetlands. One of the most valuable tools in our arsenal for wetland protection is knowledge about the species and the habitat they provide. The presence of rare or unique species in a wetland can heighten the need for its protection, and in some cases the presence of rare species or rare habitat alone may be sufficient justification for management agencies to save specific wetlands from further human development.

Currently very little research has been conducted to compare the effectiveness of different methods to sample wetland macrophytes in Great Lakes coastal wetlands. Methods generally vary, depending on the research goals and scope of the project. Some of the factors that researchers should consider when choosing the appropriate method include: cost, time/efficiency, available equipment (e.g., hip waders, canoe, small boat), number of personnel and the degree of expertise of the personnel (Hoel 1943). With the recent focus on accurate assessment of wetland biodiversity, it is also important to develop protocols that can locate rare species.

In ecology, random sampling is considered to be the gold standard (Rathbun and Gerritsen 2001), and is highly desirable when comparing the relative distribution of species within a given area or between different areas, or when making inferences about the whole population based on a subset of that population. It may not be appropriate when the goal is habitat conservation, because in many cases, habitat conservation is fueled by the desire to protect an area for its uniqueness, such as the presence of rare species that require specific niches. While the strength of random sampling is its ability to identify average or representative conditions, it is generally weak for identifying rare taxa, unless the site is sampled exhaustively. Plants are generally found in clumped distributions because of some underlying gradient such as depth or wave/wind exposure. A sampling design such as simple random sampling is best suited for "random" or "uniform" distribution of organisms but is less precise when some zones have greater species richness than others. A

suitable sampling design for biodiversity assessment should therefore match the research question, and produce results that maximize the ecological and statistical relevance (Eberhardt and Thomas 1991).

In this study, we consider two sampling techniques that have been reported in the literature, the *grid* method and the *transect* method. The grid method requires the researcher to set up an appropriate grid pattern using poles or flagging tape throughout the entire wetland of interest, and then sample quadrats at each line intersection of the grid (e.g., Knapton and Petrie 1999). This method is associated with the most comprehensive spatial coverage of the site, and is assumed to yield the most complete species list. Although a grid pattern is relatively easy to set up in the terrestrial portion of wetlands (e.g., emergent or wet meadow zone), it is very difficult to do so in the aquatic portion of wetlands, where the water is often too deep for poles to be inserted to establish the grid pattern, and where movement along straight grid lines is impossible when wading or paddling in canoes. This method is generally considered the most time-consuming and labour-intensive, and is therefore seldom used in wetland surveys.

In the transect method, researchers establish one or more (three being most common) straight lines of a standard length (e.g., 100 m) that extend from the wet meadow (terrestrial portion of the wetland) to the aquatic portion of the wetland ($\sim$1-m depth contour; Albert and Minc 2004). Wetland plants are then sampled along the entire transect within a standard strip width (e.g., 1-m wide; Cohen et al. 2004), or in a number of 1 $\times$ 1 m quadrats at set intervals along the transect (Bourdaghs et al. 2006). Although it is relatively easy for the researcher to identify plant species while walking within the established strip from the wet meadow to the water's edge, it is difficult to accurately identify submersed plants down to the 1-m contour unless researchers use waders or a canoe. Therefore, the transect technique is difficult to apply when sampling the aquatic portion of coastal marshes for the same reasons mentioned for the grid method above. A more serious objection is that the lower boundary of the wetland usually extends below 1 m, especially in undisturbed wetlands with good light penetration, and hence the transect method tends to underestimate the species richness of submersed aquatic vegetation, which is an important habitat component of fish (Seilheimer and Chow-Fraser 2006; Seilheimer and Chow-Fraser 2007) and benthic invertebrates (Kostuk 2006).

In the opinion of Rathbun and Gerritsen (2001), the scientific judgement of the researcher should not be ignored, even in random sampling. They suggested a stratified random sampling approach, in which a wetland is divided into appropriate vegetation zones based on the researcher's knowledge of the ecosystem, and then randomly sampled within these zones. A stratified random sampling design was also suggested to be most effective for determining tree diversity and species richness (Gimaret-Carpentier et al. 1998) when compared to random sampling in tropical forests where a strong gradient exists due to elevation. Croft and Chow-Fraser (2007) modified the stratified-random design in their *stratified* method, using their judgement to guide them to different habitat zones within the shoreline and aquatic communities of wetlands. With this method they began at one habitat zone (e.g., submergent vegetation) and sampled this by identifying all macrophyte species present in at least one quadrat, and then moved to a different habitat zone (e.g., floating, emergent stand, etc.,) and identified all species present in another quadrat within that zone. This would continue until all major habitat types were sampled, and until successive quadrats revealed no "new" species (usually from 10 to 15 quadrats).

The ST method can be considered a type of adaptive cluster sampling. Adaptive cluster sampling is an effective method of sampling species that have a clumped distribution, or species that are very rare within a given area (Smith et al. 2003). Adaptive cluster sampling

often initially uses a type of regular (grid/transect) sampling technique but then requires some type of trigger (e.g., population density greater than a given number) to be present, that then calls for increased sampling in that area. In the case of wetland plant sampling the presence of one rare or uncommon species doesn't necessarily mean there will be other rare or uncommon species in the vicinity. But, the presence of a new vegetation zone or unique habitat feature does increase the likelihood of finding new species in that area. So wetlands with greater diversity of habitat features, and environmental conditions do provide greater opportunity for rare or uncommon species to occupy those niches. For the ST method the trigger that increased sampling is the presence of a vegetation zone or habitat feature that has not yet been surveyed in the wetland. A disadvantage of the adaptive cluster sampling is the inclusion of the grid or transect sampling methods in the initial phase of the sampling that would increase set-up time and ultimately increase the amount of time required for sampling, decreasing efficiency.

The impetus for this study was the observation that the transect method missed certain rare taxa that may be important when calculating the wetland macrophyte index (WMI; Croft and Chow-Fraser 2007), a biotic index that has been used to rank the quality of fish habitat in coastal wetlands according to degree of water-quality impairment stemming from human development in watersheds and along the shoreline. The *grid* method is assumed to yield a more complete species list (including the rare species) because of the comprehensive spatial coverage; however, this method is very time consuming to conduct and as mentioned earlier, very difficult to carry out in the aquatic zone of wetlands. We hypothesized that the stratified method of Croft and Chow-Fraser (2007) would be as effective as the grid method in yielding the total species richness, as well as the number of unique or rare species, but would be more efficient with respect to effort. In addition, we predict that the stratified method would yield a higher species richness, and in particular, identify more rare species, compared with the transect method. We will also determine if calculated WMI scores vary significantly among the three methods, because management of the Great Lakes coastal zone is the shared responsibility of many environmental agencies that use a variety of sampling techniques, and it is important to determine how the WMI performs with different data sources.

Study sites

Six wetlands were chosen for this study, three pristine wetlands in Georgian Bay, and three degraded wetlands in Lake Ontario (Fig. 1). The reason for including both pristine and degraded wetlands is to ensure that results of this study would be applicable across the degradation gradient, since we wanted to test how sampling methods would affect the performance of the WMI. The three pristine wetlands, Black Rock, Coffin Rock and Thunder Bay are located in Tadenac Bay, which has been privately owned by the Tadenac Club for over 100 years. There is no public access permitted in Tadenac Bay and the Club limits their numbers to a maximum of ten members at the lodge per week during the open-water season. As a result, Tadenac Bay has some of the most pristine wetlands in all the Great Lakes and is considered a reference site. The three wetlands we studied were embayment wetlands which had low nutrients and turbidity and a diverse community of submergent, emergent, floating and meadow plants.

The three degraded wetlands are all located in the lower lakes, and included Cootes Paradise marsh, Bronte Creek and Jordan Harbour. Cootes Paradise is a large (250 ha) drowned river-mouth wetland located at the western-most end of Lake Ontario. A remedial action plan (RAP) was implemented in 1992 due to high nutrients and turbidity from the

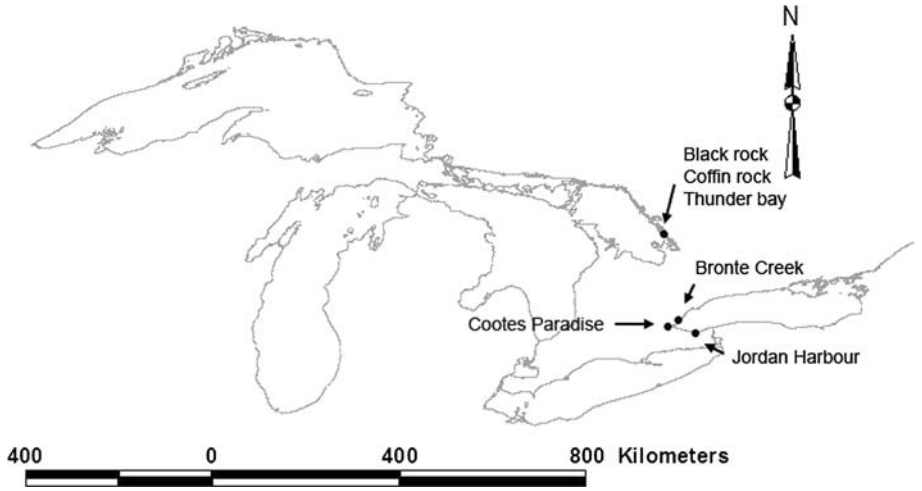

Fig. 1 Map of the Laurentian Great Lakes showing the location of the six study sites, three in eastern Georgian Bay, and three in western Lake Ontario

Dundas Sewage Treatment Plant, urban run-off, and the feeding and spawning activity of the benthivorous common carp (*Cyprinus carpio*; Lougheed et al. 1998). The two main restoration projects of the RAP consisted of marsh revegetation which began in 1994 followed by the exclusion of common carp in 1997 (Lougheed et al. 2004). According to Chow-Fraser's (2006) water quality index (WQI), the water quality within the marsh has improved from the "highly degraded" state in 1993, to the "very degraded" category between 1994 and 1998, and by 2002, has almost reached the "moderately degraded" category. Despite the improvements in water quality, the recovery of the wetland vegetation has been slow to respond (Croft and Chow-Fraser 2007). Because of the large size of Cootes Paradise, only the northern embayment known as Hopkins Bay was used in this study.

The other two wetlands included Bronte Creek and Jordan Harbour. Bronte Creek is a riverine wetland located on the north shore of Lake Ontario in the City of Oakville. Bronte Creek drains a largely urban catchment and the wetland is surrounded by houses and apartment buildings. This wetland is characterized by high turbidity levels and few submergent plants. The Jordan Harbour wetland is also a riverine wetland located on the southern shore of Lake Ontario. It is impacted by agricultural run-off from farms, vineyards and orchards in the area and is characterized by very dense submergent and floating plant growth.

Methods

Wetland surveys

Wetland macrophytes were sampled in the six wetlands with the three methods under consideration. For convenience, we will use the abbreviations 'GR', 'TR' and 'ST' when referring to the grid, transect and stratified methods. All sampling was conducted during

the height of the growing season (late June to early September) 2006. Wetlands were surveyed by canoe in the deeper areas (from 0.25 to 2 m) and by hip waders for the shallower areas and the wet meadow. Macrophyte presence/absence data were collected within 0.75 × 0.75 m quadrats at each sampling point. All macrophytes were identified to species according to Crow and Helquist (2000) and Chaade (2002). Specimens of plants that could not be identified in the field were collected, dried and pressed if necessary, and examined more thoroughly in the lab. In the deeper areas, a rake was used to collect rooted plant specimens for easier identification. The boundary of the wetland was determined before sampling began so as to ensure that the same area was equally covered with all three methods. At the time of sampling, water samples were collected to determine water turbidity with a LaMotte™ portable turbidimeter.

For the GR method, quadrats were spaced roughly 10–15 m apart in a grid pattern oriented along the longest axis of the wetland/embayment. The grid was set up with metal poles (length of 3 m). For the TR method, transect were established along a depth gradient, in order to encounter as many habitat zones as possible. At least three transects were sampled per wetland and quadrats were spaced 5–10 m apart along each transect line.

For the ST method the placement of quadrats relies on the investigator's judgement and it is important that the sampler is competent in identifying the various vegetation zones and habitat features within a coastal marsh. The zones include (but are not limited to) those listed in Table 1. Vegetation zones or patches exist within wetland due to underlying physical or chemical gradients such as depth, exposure and sediment composition. The observation that there is a visual change in appearance of the plant community (i.e., change from emergent to floating species) indicates that there is also a change in plant species composition. The transition between two vegetation zones indicates a change in environmental conditions which may mean that area also has unique species, and thus should be adequately covered during the survey. Unique habitat features may provide the necessary physical or chemical conditions required for some aquatic plant species. For example a stream emptying into a wetland provides nutrients and specific water chemistry conditions for the growth of some plant species. Ignoring such a unique habitat feature or vegetation zone may lead to an underestimation of species richness.

Application of the ST sampling method in coastal wetlands required a few simple steps (Table 2). First, a visual inspection of the wetland was made to identify and record all of the vegetation zones (refer to Table 1 for examples). Typically, 10 to 15 quadrats were required to completely sample a wetland or embayment (if greater than 15 quadrats are required, the investigator may consider dividing the area into two or more sites). Placement of quadrats and the distance between quadrats depended on the heterogeneity of macrophyte zones in the particular wetland. The number of quadrats sampled within each vegetation zone varied depending on the diversity of species within that zone. Each zone was thoroughly covered either on foot (wearing chest waders) or by canoe, and when new species were found a new quadrat was sampled. Sampling within a wetland continued for at least ten quadrats, and was considered complete when no new species were found in two consecutive quadrats. As we have pointed out in the Introduction, the GR and TR methods are documented sampling protocols used by previous investigators of coastal wetlands (e.g., Knapton and Petrie (1999) and Cohen et al. (2004), respectively). We used satellite images of study sites and a GPS unit to assist in placement of the transects and grid pattern.

Each wetland was sampled with the three methods on the same day to reduce confounding effects of seasonal variation or differences due to meteorological conditions. The ST method was always conducted first to avoid knowledge gained from either of the other two methods to influence the selection of sites. Had we located a rare species with the GR

Table 1 Vegetation zones within Great Lakes coastal marshes (adapted from Cvetkovic 2008)

Vegetation zone	Examples of vegetation	Description
Shrubs/meadow	*Myrica gale*, *Spirea* spp., *Carex* spp.	Meadow marsh; sedge meadow
Shoreline herbaceous	*Galium* spp., *Lycopus uniflorus*, *Lobelia siphilitica*	Herbs that grow along the shoreline, and rarely inundated
Robust emergent	*Typha* spp.	Emergent band of dense vegetation occurring at the shoreline and extends into shallow water up to 0.3 m
Narow-leaved shoreline emergent	*Schoenoplectus acutus*, *Schoenoplectus validus*, *Sparganium* spp. *Carex* spp. *Juncus* spp.	Narrow-leaved emergents that are usually found occurring along the shoreline
Shallow emergent	*Zizania* spp., *Sagittaria latifolia*, *S. cuneata*, *Pontederia* spp.,	Emergent plants that are usually found below the shoreline in shallow water
Rooted basal rosettes	*Eleocharis acicularis*, *Eriocaulon aquaticum*, *Isoetes* sp. *Lobelia dortmanna*	Low-growing rosettes that occur along open shoreline areas, either emergent or submergent—not usually found in areas with robust emergent.
Rooted floating	*Nymphaea* spp., *Nuphar* spp., *Brasenia schreberi*	Floating species that can grow in deeper water up to 0.5 m
Free-floating	*Lemna* spp., *Wolfia* spp.	Unrooted floating species that can occur anywhere in the marsh
Rooted and unrooted submergent	*Ceratophyllum demersum*, *Utricularia* sp., *Najas flexilis*, *Potamogeton robbinsii*	Submerged plants in intermediate water depths
Canopy	*Bidens beckii*, *Elodea canadensis*, *Potagmogeton amplifolius*, *P. richardsonii*, *Myriophyllum spicatum*	Rooted plants that cannot tolerate low light conditions and can reach the surface of the water column

Table 2 Steps to follow when sampling coastal wetlands with the stratified (ST) method

1. Survey the wetland to identify and record the vegetation zones (Table 1), and any unique habitat features (stream outlets, changes in substrate, seeps, etc.,). The goal is to sample at *least* one quadrat within each vegetation zone and unique habitat feature
2. Beginning in the deep open water (using a boat or chest waders), sample the first quadrat using a rake or dredge (This will be the open water boundary of the wetland where canopy species will occur). Record all species found in the quadrat
3. Sample one representative quadrat within the rooted and unrooted submergent zone. A dredge or rake may be required to pull up plants that are not readily seen from above water. Record all species found in this quadrat.
4. Choose another quadrat within the submergent zone in an area different than that encountered previously. Record all species, including those that have not been recorded from the first two quadrats
5. Sample at least one quadrat in all other applicable vegetation zones listed in Table 1 (i.e., shallow emergent, robust emergent, etc.,) and record any new species found. Note that not all of the zones may be represented in every wetland
6. Continue sampling until all vegetation zones and habitat features have been sampled, including the shrubs/meadow, which marks the upland boundary of the wetland
7. Sampling is complete when all vegetation zones have been sampled at least once, and when at least ten quadrats have been completed and no further species have been identified in two consecutive quadrats

method first, it would have been difficult to ignore this information when carrying out the ST method later.

Wetland macrophyte index (WMI) scores

The Wetland Macrophyte Index (WMI) was developed with data obtained from coastal wetlands located in all five Great Lakes (Croft and Chow-Fraser 2007), by relating plant presence-absence data to measured water quality conditions in 127 coastal wetlands (154 wetland years). It was then validated with data from wetlands in Lakes Huron, Ontario and Erie, and was proven to be a robust method for determining wetland quality. The Adjusted WMI (WMIadj), used to account for presence of exotic species, provided an index of the ecological health of the wetland ecosystem in addition to the degree of water-quality impairment (Croft and Chow-Fraser 2007). The WMI has proven to be a valuable tool for determining the quality of coastal wetlands in the Great Lakes (Croft and Chow-Fraser 2007; Seilheimer et al. 2009).

Geographic data

Each quadrat sampled was georeferenced with a Garmin^{TM} Etrex GPS (4–6 m accuracy) and latitude and longitude values were imported into a GIS with ArcMap 8.2 (ESRI copyright 2002). The depth was also recorded at each quadrat with a metre stick or a weighted line marked in 10-cm depth increments.

Randomized re-sampling

To determine the relationship between species richness and sampling effort (e.g., total number of quadrats sampled), we first carried out a post hoc randomized re-sampling of the corresponding quadrats for each method in a given wetland. The first step in this procedure was to generate a randomized series of quadrats associated with the GR data by randomly selecting (random number table) 1, 3, 6, 12, 20 and 36 quadrats (with replacement). This was meant to simulate results we would have obtained had we performed a randomized sampling. We then determined the number of species that corresponded to the re-sampled quadrats (i.e., the series of 1, 3, 6, 12, 20 and 36 quadrats). For the ST and TR methods, fewer quadrats were re-sampled because fewer quadrats had been sampled originally. Species richness values were transformed (squared) to produce a linear relationship for analysis of covariance.

Statistical analysis

All statistical analyses were conducted with SAS JMP IN 5.1 (SAS Institute, Cary, North Carolina, USA). We performed one-way, two-way and three-way analysis of variance (ANOVA), and whenever appropriate, used the Tukey–Kramer post hoc test for pairwise comparison of means. A non-linear regression analysis (Y^2 transformation) was used to determine the relationship between species richness and sampling effort (i.e., number of quadrats used) for comparison of sampling methods. The relationship between square-transformed species richness data and sampling effort were determined with linear regression analysis. The slopes and intercepts were compared with an analysis of covariance (ANCOVA).

Results

A total of over 500 quadrats were sampled within the six wetlands, but the number of quadrats sampled at each site differed according to the size of the study areas. Cootes Paradise was the largest, followed by Coffin rock, Black rock, Thunder bay, Jordan harbour and Bronte creek. Hence, the number of quadrats required by the GR method in Cootes was highest (61), and that for Bronte Creek was lowest (40) (Table 3). Regardless of size, however, the total number of quadrats associated with the three methods differed, and as expected, the GR method required the most effort (mean of 51.5 ± 3.23 SE) because of its comprehensive coverage, while the TR method required fewer quadrats (mean of 31.8 ± 3.40 SE) and the ST method required the least (mean of 14.2 ± 0.65 SE).

As indicated earlier, we deliberately chose sites that differed with respect to environmental quality so that our results would have widespread applicability. Since one of the obvious differences between degraded and pristine sites is water clarity (Chow-Fraser 2006), we measured water turbidity at each site to verify their status. Turbidity levels in the three Georgian Bay sites ranged from 0.40 to 1.54 NTU, and this confirms their status as pristine wetlands (Table 3). By comparison, the other three wetlands had much higher turbidity levels; the value for Cootes Paradise was 30.8 NTU, while that for Bronte Creek and Jordan Harbour were 14.4 and 8.7 NTU, respectively, thus confirming their status as degraded sites.

Species richness

The total number of plant species identified with all three methods (GR, ST and TR) ranged from site to site, with generally higher species richness associated with pristine sites (43–50), than with degraded sites (17–32) (Table 4). To account for this effect of wetland quality, we carried out a two-factor ANOVA, which tested the effect of sampling method, wetland quality, and the interaction between these. Both sampling method and wetland quality had a statistically significant effect on species richness ($P = 0.0064$ and < 0.0001, respectively), but there was no significant interaction between these factors ($P = 0.7604$; Table 5). The ST method identified significantly more species than did the other two (30.83 ± 3.93 compared with 23.83 ± 4.53 and 20.33 ± 3.99 for GR and TR, respectively; Fig. 2a). We also found that regardless of methods used, mean species richness for the pristine sites was significantly higher than that for degraded sites (Fig. 2b). In the case

Table 3 Area, turbidity and total number of quadrats sampled with each method in the six study sites

Wetland	Size of study area (ha)	Water turbidity	No of quadrats sampled		
			GR	ST	TR
Pristine sites					
Black rock	1.87	1.54	58	15	24
Coffin rock	1.64	0.40	49	17	45
Thunder bay	1.52	1.07	55	13	35
Degraded sites					
Bronte creek	0.49	14.4	40	13	24
Cootes paradise	0.47	30.8	61	14	36
Jordan harbour	0.37	8.7	46	13	27
Average for all sites			51.5	14.2	31.8

GR Grid, *ST* Stratified and *TR* Transect

Table 4 Summary of total number of submergent, emergent, wet meadow and floating species recovered in each wetland by the three methods (*GR* grid; *ST* stratified; *TR* transect), and when data from all three methods were combined (COMB)

Macrophyte habit	Method	Black rock	Coffin rock	Thunder bay	Bronte creek	Cootes paradise	Jordan harbour
All taxa	GR	35 (70.0)	33 (70.2)	32 (74.4)	10 (58.8)	12 (46.2)	21 (65.6)
	ST	42 (84.0)	41 (87.2)	32 (74.4)	17 (100.0)	25 (96.1)	28 (87.5)
	TR	29 (58.0)	30 (63.8)	27 (62.7)	8 (47.1)	10 (38.5)	18 (56.2)
	COMB	50	47	43	17	26	32
Emergent taxa	GR	7 (70.0)	5 (62.5)	6 (60.0)	5 (83.3)	6 (46.2)	3 (60.0)
	ST	9 (90.0)	7 (87.5)	8 (80.0)	6 (100.0)	8 (96.1)	5 (100.0)
	TR	5 (50.0)	5 (62.5)	7 (70.0)	4 (66.6.1)	5 (38.5)	2 (40.0)
	COMB	10	8	10	6	9	5
Floating taxa	GR	4 (80.0)	5 (100.0)	3 (75.0)	2 (100.0)	2 (66.7)	4 (80.0)
	ST	4 (80.0)	5 (100.0)	4(100.0)	2 (100.0)	3 (100.0)	5 (100.0)
	TR	5 (100.0)	2 (40.0)	3 (75.0)	2 (100.0)	1 (33.3)	4 (80.0)
	COMB	5	5	4	2	3	5
Wet meadow taxa	GR	8 (57.1)	6 (75.0)	9 (90.0)	1 (16.7)	2 (22.2)	9 (60.0)
	ST	12 (85.7)	5 (62.5)	6 (60.0)	6 (100.0)	9 (100.0)	12 (80.0)
	TR	4 (28.6)	5 (62.5)	4 (40.0)	1 (16.7)	1 (11.1)	7 (46.7)
	COMB	14	8	10	6	9	15
Submergent taxa	GR	16 (76.2)	17 (65.4)	14 (77.8)	2 (66.7)	2 (40.0)	5 (71.4)
	ST	17 (80.9)	24 (92.3)	14 (77.8)	3 (100.0)	5 (100.0)	6 (85.7)
	TR	15 (71.4)	18 (69.2)	13 (72.2)	1 (33.3)	3 (60.0)	5 (71.4)
	COMB	21	26	18	3	5	7

Numbers in bracket correspond to the percentage of species identified by each method relative to the total number of species recovered by all methods combined

of GR and TR, there were twice as many species identified in pristine as in degraded sites. Lack of a significant effect between factors indicated that the effect of sampling method was not dependent on wetland quality.

Differences in total plant species richness among sites noted in Table 3 were mostly attributed to the much higher number of submergent taxa in pristine wetlands (18–26) compared to degraded wetlands (3–7); the number of emergent, floating and wet meadow taxa did not appear to vary as greatly across sites as did submergent taxa (Table 4). We wanted to determine how species richness of these various plant groups was affected by sampling method and wetland quality and carried out a three-way ANOVA that accounted for plant group, wetland quality, and sampling method, as well as all possible interactions among these factors. Both plant group and wetland quality had a significant effect on species richness, and there was also a significant interactive effect between these ($P < 0.0001$ for all sources; Table 6). Consistent with results of the two-way ANOVA, there were twice as many species in pristine sites as in degraded sites (8.38 vs. 4.13 (least squared mean values), respectively), although as noted previously, the major difference was noted for submergent taxa (Fig. 3).

We also confirmed that significantly more species were identified through the ST method (7.71) (Least squared mean values) than through the GR (5.96) or TR (5.13) method ($P = 0.0008$) when all data were examined together in the three-way ANOVA

Table 5 Summary of two-way ANOVA testing the effect of sampling method, wetland quality and interaction between sampling method and wetland quality on species richness of macrophytes in wetlands

Source	DF	SS	MS	*F*	*P*
Model	5	1,638.677	327.73	15.165	0.0001
Sampling method	2	343.000	171.50	7.936	0.0064
Wetland quality	1	1,283.556	1,283.56	59.393	0.0001
Sampling method × wetland quality	2	12.111	6.06	0.280	0.7604
Error	12	259.333	21.611		
Total	17	1,898.000			

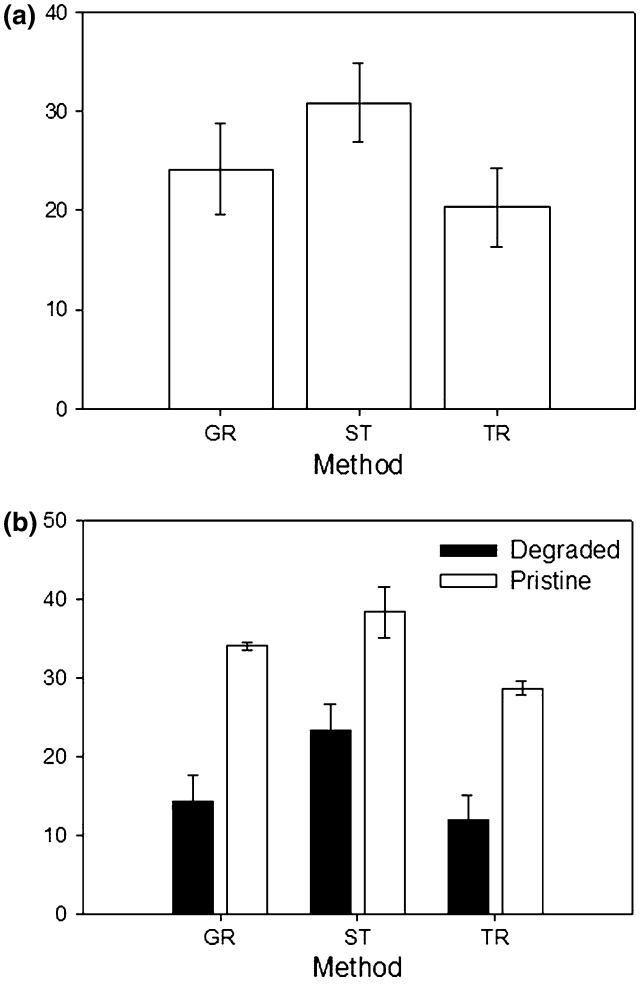

Fig. 2 Comparison of mean species richness (±SE) associated with three sampling methods for **a** all six sites in this study and **b** when sites are grouped according to environmental quality (*open pristine sites*; *solid degraded sites*)

Table 6 Summary of three-way ANOVA testing the effect of sampling method, wetland quality, plant group, and all possible interactions among these factors on species richness of macrophytes in wetlands

Source	DF	SS	MS	*F*	*P*
Model	23	1,313.986	57.129	11.331	0.0001
Sampling method	2	83.444	41.722	8.275	0.0008
Wetland quality	1	325.125	325.125	64.487	0.0001
Plant group	3	411.931	137.310	27.235	0.0001
Sampling method × wetland quality	2	3.000	1.500	0.297	0.7440
Plant group × wetland quality	3	448.931	149.644	29.681	0.0001
Plant group × sampling method	6	23.444	3.907	0.775	0.5940
Plant group × wetland quality × sampling method	6	18.111	3.018	0.598	0.7290
Error	48	242.000	5.042		
Total	71	1,555.986			

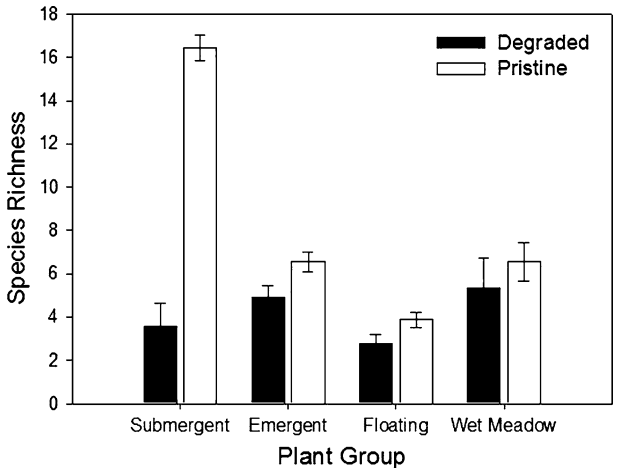

Fig. 3 Comparison of mean species richness (±SE) associated with four plant groups sampled in pristine (*open bars*) and degraded (*solid bars*) sites

model. However, once the data were sorted by plant group, sampling method was only significant for emergent taxa ($P = 0.0209$), even though the significant effect of wetland quality was evident for both submergent ($P < 0.0001$) and emergent ($P = 0.0128$) taxa. For both pristine and degraded wetlands, mean emergent species richness associated with the ST method (8.0 and 6.3, respectively) were consistently higher than those for the GR (6.0 and 4.7, respectively) and TR (6.0 and 3.67, respectively). We found no significant effect of sampling method or wetland quality on species richness of wet meadow or floating taxa (Fig. 4).

Unique species

The number of unique species (those found with only a single method; Table 7) varied among the three sampling methods, and was highest for the ST method (9.00 ± 0.816 [9.67 ± 1.45

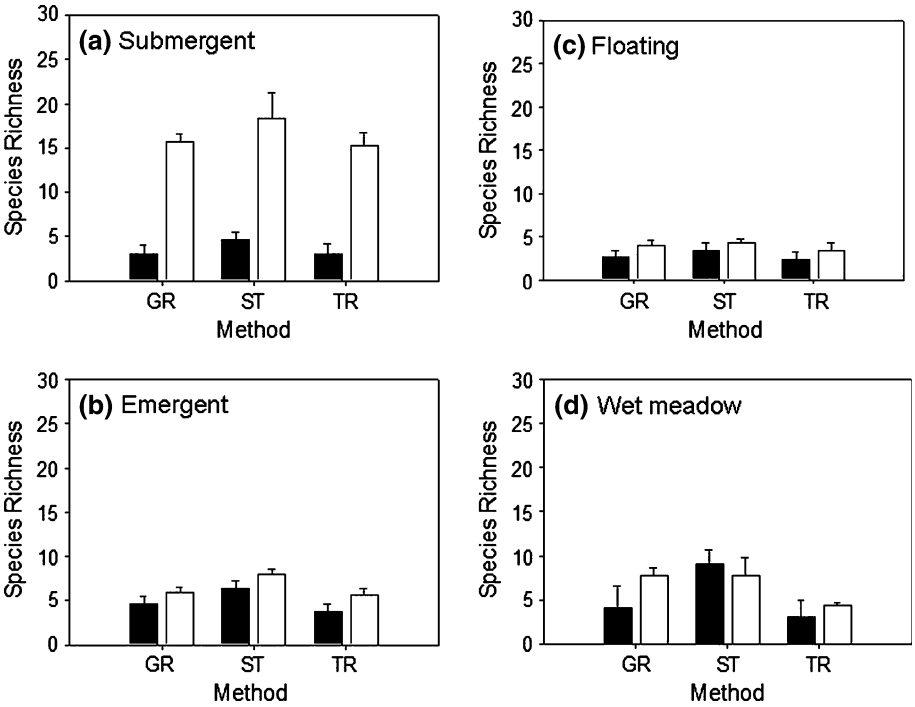

Fig. 4 Comparison of mean species richness (±SE) associated with three sampling methods for pristine (*open bars*) and degraded (*solid bars*) sites. Data are presented separately for **a** submergent taxa **b** emergent taxa **c** floating taxa and **d** wet meadow taxa

adjusted for quality]; two-way ANOVA; $F = 39.98$; $P < 0.0001$; $df = 2$), irrespective of wetland quality (mean of 8.33 ± 0.88 vs. 9.67 ± 1.45 for pristine and degraded wetlands, respectively). There were also significant differences among plant groups (Table 8), with the greatest number associated with the wet meadow group (1.83 ± 0.57), followed by the submergent (1.17 ± 0.29), emergent (0.89 ± 0.28) and floating (0.11 ± 0.08) (one-way ANOVA; $P = 0.0101$; Tukey–Kramer post hoc test; $P < 0.05$). A three-way ANOVA was carried out to determine the effect of sampling method, plant group, and wetland quality on the number of unique species. There was a significant effect of plant group and sampling method on the number of unique species, but no significant effect of wetland quality (Table 9). When analyses were run separately for each plant group, we found a significant effect of sampling method on the number of unique species for emergent ($P < 0.0001$) and wet meadow ($P = 0.0005$) taxa. In both cases, the ST method identified significantly more unique taxa than the other two methods (Tukey–Kramer test; $P < 0.05$).

Uncommon species

The presence of rare or endangered species is often used by managers to justify protecting habitat. Based on data from 62 wetlands (1,099 quadrats) that were sampled in Georgian Bay in 2005 and 2006, we calculated frequency of occurrence for the 136 species that were found. We considered a species uncommon if it was found in fewer than 5% of the quadrats sampled. The mean number of uncommon species was significantly higher for ST (20.7), compared with the mean for TR (8.0) and GR (10.3) (ANOVA; $P < 0.0089$). Some

Table 7 Comparison of the number of species unique to a single method on a site-by-site basis

Wetland	Sampling method		
	GR	ST	TR
Pristine sites			
Black rock	6	10	1
Coffin rock	1	8	2
Thunder bay	4	7	3
Degraded sites			
Bronte creek	0	7	0
Cootes paradise	0	12	0
Jordan harbour	1	10	1

Table 8 Number of unique species grouped by plant groups: submergent (SUB), emergent (EM), floating (FL), and wet meadow (WM)

Wetland	Sampling method	Macrophyte group			
		SUB	EM	FL	WM
Pristine sites					
Black rock	GR	4	1	0	1
	ST	1	3	0	6
	TR	1	0	0	0
Coffin rock	GR	0	0	0	1
	ST	4	3	0	1
	TR	1	0	0	1
Thunder bay	GR	1	0	0	3
	ST	2	3	1	1
	TR	2	1	0	0
Degraded sites					
Bronte creek	GR	0	0	0	0
	ST	1	1	0	5
	TR	0	0	0	0
Cootes paradise	GR	0	0	0	0
	ST	2	2	1	7
	TR	0	0	0	0
Jordan harbour	GR	0	0	0	1
	ST	1	2	1	6
	TR	1	0	0	0

examples of uncommon species encountered in Georgian Bay wetlands were creeping spearwort (*Ranunculus reptans*), floating heart (*Nymphoides cordata*), flat-leaved bladderwort (*Utricularia intermedia*), creeping bladderwort (*Utricularia gibba*), horned bladderwort (*Utricularia cornuta*), and quillwort (*Isoetes* spp.).

Randomized re-sampling

Species richness was plotted against number of quadrats sampled in Fig. 5; the solid horizontal line in this figure indicates the total number of species found in a wetland by all

Table 9 Summary of three-way ANOVA testing the effect of sampling method, wetland quality, plant group, and all possible interactions among these factors on the number of unique species in wetlands

Source	DF	SS	MS	*F*	*P*
Model	23	138.667	6.029	7.001	0.0001
Sampling method	2	53.083	26.541	30.822	0.0001
Wetland quality	1	2.000	2.000	3.323	0.1340
Plant group	3	27.444	9.148	10.624	0.0001
Sampling method × wetland quality	2	3.583	1.791	2.081	0.1360
Plant group × wetland quality	3	8.111	2.704	3.139	0.0337
Plant group × sampling method	6	28.472	4.745	5.511	0.0002
Plant group × wetland quality × sampling method	6	15.972	2.662	3.091	0.0122
Error	48	41.333	0.861		
Total	71	180.000			

three methods, while the dashed line represents 80% of the total. Non-linear regression equations fitted through each set of method-wetland data were all highly significant, with correspondingly high r^2 values (Table 10). We performed an ANCOVA (with square transformed data) and found significant differences in slopes among the three methods ($P < 0.0010$). Slopes for the ST method were always steeper than those for the other two methods, indicating that a greater number of species were identified for a given effort. Much lower efficiency was associated with the GR and TR methods as indicated by the much lower slopes.

We used the slopes obtained from Table 10 to estimate the number of quadrats required to find 80% of the total species richness for each wetland (Table 11). For the ST method, it would require 8–16 quadrats, which is at least seven times fewer quadrats compared with the TR and GR methods (54–146 and 54–179, respectively). Even if we exclude the unusually high numbers for Cootes Paradise (146 and 179, respectively for TR and GR), the average number of quadrats required to find 80% of the species within a wetland would be 13 for ST, 62 for GR and 66 for TR.

Wetland macrophyte index (WMI)

We used the species information to calculate both WMI and WMIadj scores for all sites and methods (Table 12). Sampling method had no significant effect on either set of scores (Kruskal Wallis, $n = 6$; $P > 0.05$), and this was true when all sites were combined for the analysis or when they were analyzed separately by wetland quality.

Discussion

The wetlands chosen for this study were selected because of the species diversity and water quality within the wetlands. Wetlands in Georgian Bay are characterized by low nutrients and low turbidity, which result in high species diversity (Chow-Fraser 2006). The turbidity levels in the three Georgian Bay sites ranged from 0.40 to 1.56 NTU (Table 3). The primary environmental factors that currently impact the aquatic plant communities in Georgian Bay are wind and wave exposure and prolonged lowering of the water levels.

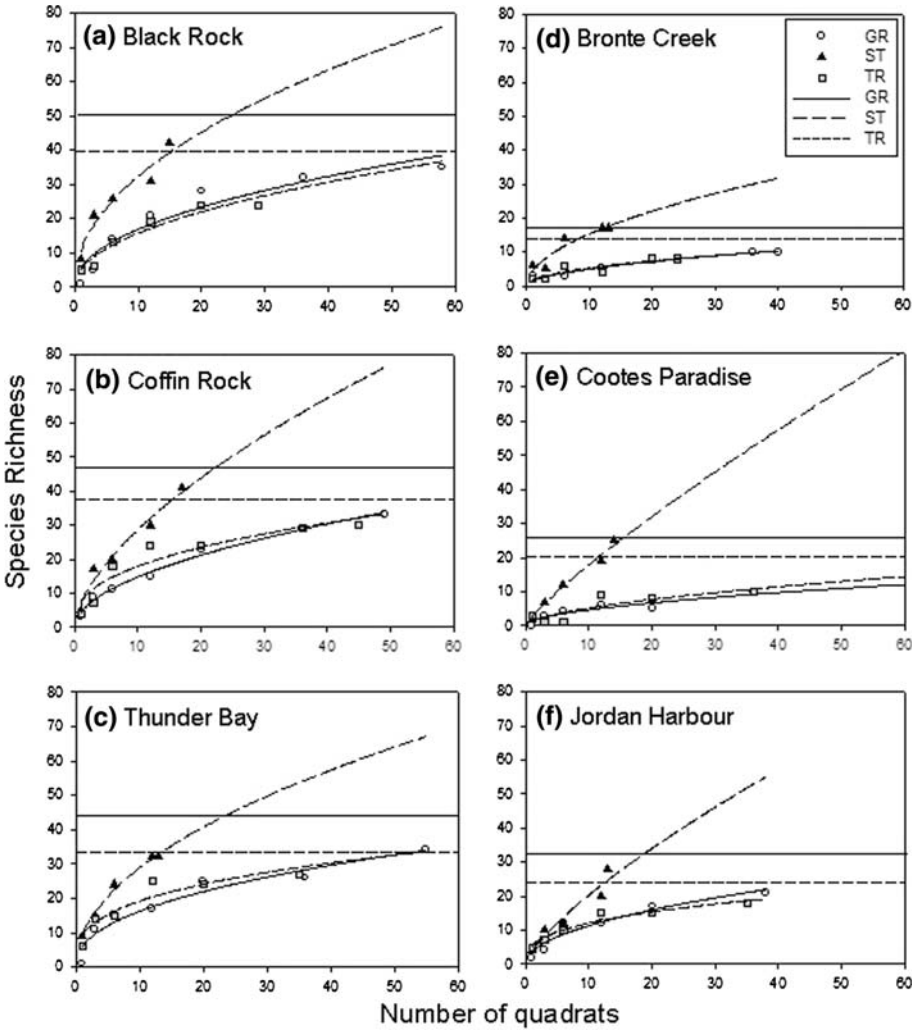

Fig. 5 Cumulative number of species identified by each method for **a** Black Rock **b** Coffin Rock **c** Thunder Bay **d** Bronte Creek **e** Cootes Paradise Marsh and **f** Jordan Harbour. *Horizontal lines* indicate 100% (*solid line*) and 80% of species (*dashed line*) identified by all three methods. See Table 10 for corresponding r^2 values of method-specific regressions

Conversely, wetlands in Lake Ontario and Lake Erie have much higher nutrient and turbidity levels that have been attributed to urban and agricultural run-off, and this can greatly affect the type of submergent plant communities that are present (Chow-Fraser 2006; Croft and Chow-Fraser 2007; McNair 2006; McNair and Chow-Fraser 2003). For example, the turbidity level for Cootes Paradise was very high (30.8 NTU) because of high levels of nutrients from urban sources (waste-treatment facility and urban runoff) and resuspension by wind and waves and bioturbation from common carp (*Cyprinus carpio*; Chow-Fraser 2005). Bronte creek also had relatively high turbidity (14.43 NTU), because the wind can easily re-suspend the sediment in this shallow (mean depth of 30 cm) wetland. Jordan Harbour, by comparison, had the lowest turbidity (8.73 NTU) of Lake Ontario

Table 10 Summary of statistics associated with regression analysis relating species richness to total quadrats *y* species richness, *x* total quadrats

Wetland	Method	Equation	r^2	*P*
Pristine				
Black rock	GR	$y^2 = 22.368x + 93.416$	0.902	0.0010
	ST	$y^2 = 102.907x + 19.681$	0.912	0.0114
	TR	$y^2 = 22.134x + 28.581$	0.910	0.0031
Coffin rock	GR	$y^2 = 22.882x - 1.571$	0.991	0.0001
	ST	$y^2 = 97.071x - 98.153$	0.964	0.0029
	TR	$y^2 = 19.229x + 130.973$	0.863	0.0025
Thunder bay	GR	$y^2 = 19.643x + 68.624$	0.952	0.0002
	ST	$y^2 = 81.596x + 14.824$	0.986	0.0006
	TR	$y^2 = 19.201x + 151.424$	0.772	0.0211
Degraded				
Bronte creek	GR	$y^2 = 2.681x - 0.758$	0.971	0.0001
	ST	$y^2 = 23.395x + 3.327$	0.918	0.0101
	TR	$y^2 = 2.664x + 2.033$	0.803	0.0156
Cootes paradise	GR	$y^2 = 2.419x - 0.888$	0.955	0.0010
	ST	$y^2 = 44.002x - 80.211$	0.929	0.0082
	TR	$y^2 = 2.915x + 4.770$	0.754	0.0247
Jordan harbour	GR	$y^2 = 11.725x + 16.666$	0.948	0.0010
	ST	$y^2 = 53.199x - 83.551$	0.830	0.0314
	TR	$y^2 = 8.642x + 47.086$	0.889	0.0048

Table 11 Number of quadrats required by each sampling method to survey 80% of the total species richness for each site

Wetland	Number of quadrats		
	GR	ST	TR
Pristine sites			
Black rock	67	15	71
Coffin rock	62	16	67
Thunder bay	57	14	54
Degraded sites			
Bronte creek	69	8	69
Cootes paradise	179	12	146
Jordan harbour	54	14	70
Average	62 (81)	13	66 (80)

Values calculated using equations in Table 10. Average values at the bottom of the table are for the number of quadrats excluding data for the GR and TR from Cootes Paradise; numbers in brackets are averages calculated without excluding Cootes data

wetlands in this study, mainly because it is well protected, is deeper and has a well-established community of submergent vegetation that does not permit sediment to be easily re-suspended by wind and wave action. Given this heterogeneous range of environmental conditions encountered in these six wetlands, differences emerging from this study should have widespread applicability despite the small sample size (Appendix 1).

Sampling with the ST method has been shown to produce the highest species richness compared to TR and GR. Although species richness depends on wetland quality, with there being higher species richness in the more pristine sites, the effectiveness of any given method was not dependent on the wetland quality. The fact that sampling method was only significant

Table 12 Comparison of WMI and WMIadj scores (calculated according to Croft and Chow-Fraser 2007) for data obtained from the three sampling methods in each wetland in this study

Wetland	All		GR		ST		TR	
	WMI	WMIadj	WMI	WMIadj	WMI	WMIadj	WMI	WMIadj
Black rock	3.70	3.70	3.70	3.70	3.76	3.76	3.65	3.65
Coffin rock	4.03	3.85	4.01	4.01	3.96	3.78	4.09	4.09
Thunder bay	3.84	3.64	3.85	3.63	3.92	3.92	3.78	3.78
Bronte creek	1.56	1.12	1.45	0.95	1.67	1.22	1.66	1.66
Cootes paradise	1.34	0.99	1.28	1.28	1.45	1.10	1.35	1.35
Jordan harbour	1.79	1.47	1.77	1.39	1.78	1.44	1.30	0.92

for the emergent group points to the advantage that samplers' judgment had over more random methods. Many emergent species can be visually located from a long distance away, and can be positively identified upon closer inspection. There was no apparent advantage of using the stratified method over the other methods for the submergent species because the submergent species are harder to locate from a long distance. When sampling the submergent zone the species that are found are largely limited by the path that the sampler takes (i.e., straight transect or a judgment guided route through the zone). The submergent species that can be noted from the boat is often limited by the glare off the water, so generally even in very clear water only the species directly beside the boat or within several meters can be seen. In very turbid wetlands, the submergent species cannot be visualized from the water surface so they must be retrieved with a rake to be identified; their sparse distribution also make it difficult to locate these, regardless of the sampling method used.

The number of unique species found with each method provides us with some valuable information about the effectiveness of the method. The ST method found a greater number of unique species compared to the other two methods, regardless of the wetland quality. More unique species were found with the stratified method because we were actively looking for species that had not yet been identified. Sampler judgment was used to assist in locating different habitat zones (e.g., stream mouth, sandy or rocky areas, beaver lodge) and this greatly increased the probability of finding species that are associated with those specific features. By comparison, the grid and transect method would only include unique species if they happened to fall along the transect or grid lines.

Because this method requires human judgment to determine where to locate sampling quadrats, it is important that we assess the consistency of data generated by multiple investigators when using the ST protocol. To address this, we compared data collected with the ST method by three investigators (S. McNair and V. Lougheed, who were Ph.D candidates at the time of data collection, and the first author who was conducting an M.Sc. project). There was no significant difference in WMI scores and species richness between pairs of investigators of the same set of wetlands sampled (Wilcoxon Signed Rank test between VL and MC: WMI $P = 0.66$, Species Richness $P = 0.19$; Wilcoxon Signed Rank test between SM and MC: WMI $P = 0.47$, Species Richness $P = 0.25$; Wilcoxon Signed Rank test between SM and VL: WMI $P = 0.23$, Species Richness $P = 0.43$) We feel that the skill of identifying different vegetation zones or habitat features in wetlands is far easier than that required to identify wetland plants. Therefore, under most circumstances, a field botanist or wetland ecologist who is proficient at identifying wetland plants will have no difficulty determining what vegetation communities or unique habitat features must be sampled in order to satisfy the ST protocol.

The randomized re-sampling of this large data set has provided valuable information regarding sampling effort and sampling method. The greatest disparity among slopes of species richness to sampling effort existed for Cootes Paradise Marsh (see Fig. 5e), where slopes for the GR and TR methods were extremely low compared with that of ST. This reflects the very sparse distribution of the submergent macrophyte species in the highly degraded wetland. The high water turbidity in Cootes Paradise only permits the growth of a few submergent species which are sparsely distributed. Consequently, only five or six quadrats within the transect or grid may contain any submergent macrophytes. Thus, the GR method required seven times more quadrats to be sampled to identify 80% of the species. This illustrates the much greater efficiency associated with the ST method for surveying macrophyte species richness in degraded wetlands. The amount of time required to sample a wetland is a major logistical constraint on most research programs because investigators are generally trying to maximize their sample size. The fewer the quadrats that have to be sample the less amount of time it will take.

The Wetland Macrophyte Index (WMI; Croft and Chow-Fraser 2007) utilize information associated with the functional ecology of wetland plants. In a given wetland, there may be several species occupying the same niche and are therefore ecological analogues. Hence, the presence or absence of rare or unique species do not generally affect WMI scores as long as a minimum of one ecological analogue is sampled. This is why species richness is generally not the best indicator of ecosystem health. A wetland with high species diversity that are all indicative of poor water conditions would still yield a low WMI score, whereas one with low species diversity that are indicative of good water quality will yield a high WMI score. For instance, in Black rock, quillwort (*Isoetes* spp.) was found with the stratified method whereas *Potamogeton epihydrus* was found with the grid method. Both species are found exclusively in sites with good water-quality conditions and are therefore ecological analogues, even though they are found in very different habitats (quillwort is a basal rosette in shallow habitats while the pondweed is a canopy forming plant growing in deeper water). Consequently, WMI scores associated with both methods were similar. This redundancy within wetlands is important for maintaining ecosystem integrity in the face of changing environmental conditions.

Conclusions

Many factors must be considered when choosing an appropriate sampling protocol to survey coastal wetlands. The most important consideration is the purpose or goal of the study, and random sampling should not automatically be assumed to be the most suitable. We predicted that the ST and GR would produce similar results with respect to total species richness, as well as the number of unique and uncommon species, but that the GR method would take more time and effort. Not only was the ST method more efficient in finding the greatest species richness, it also required fewer quadrats and therefore took less time, and was also more useful for locating uncommon and unique species.

Croft and Chow-Fraser (2007) showed that the WMI is a valuable tool for assessing the degree of human disturbance in coastal wetlands. One of the explicit goals of this study was to test the sensitivity of the WMI to variation in sampling methodology. We have confirmed that WMI scores calculated with data collected by these three methods are statistically homogeneous. This highlights the robustness of the WMI because different species can be ecological analogues and indicate the same water-quality conditions. It is important to note that although the WMI is suitable for determining the quality of the

wetland in relation to water-quality impairment, it cannot be used to convey information about rare species that would be valuable for conservation purposes.

In this study, species richness varied according to the sampling method used; therefore we caution against cross-study comparisons of species richness when more than one sampling protocol is involved, especially when it is used as a metric in indices of biotic integrity. We recommend the use of the ST method for routine monitoring and surveys because it will yield information both for indices calculation, and for targeting wetlands for protection and conservation. More importantly, we recognize that the use of random sampling methods will not necessarily reveal the rare and unique species that are important considerations for designating habitats for protection.

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Appendix 1

See Table 13.

Table 13 Summary of depth measurements (cm) for species found in all six wetlands

Taxon	Common name	*N*	Mean	Median	Min	Max	Range
Porifera							
Freshwater sponges	Sponges	17	49.41	46	3	89	86
Emergent taxa							
Eleocharis acicularis	Needle spike rush	2	32.50	32.5	20	45	25
Eleocharis robbinsii	Robbins' spike-rush	8	17.25	18	3	26	23
Eleocharis smallii	Marsh spikerush	45	14.80	14	0	62	62
Polygonum amphibium	Water smartweed	5	9.80	5	1	30	29
Polygonum lapathifolium	Dock-leaved smartweed	2	4.00	4	3	5	2
Polygonum sp.	Smartweed	1	25.00	25	25	25	0
Pontederia cordata	Pickerel weed	40	18.60	18	2	59	57
Ranunculus reptans	Creeping spearwort	2	1.00	1	1	1	0
Sagittaria latifolia	Broad arrowhead	18	17.11	16.5	2	33	31
Sagittaria sp.	Arrowhead species	1	38.00	38	38	38	0
Schoenoplectus acutus	Hardstem bulrush	47	23.04	23	0	68	68
Schoenoplectus americana	Three-square bulrush	5	22.20	26	5	30	25
Schoenoplectus validus	Softstem bulrush	16	10.31	7.5	0	21	21
Sparganium androcladum	Branched burreed	16	28.94	27	3	59	56
Sparganium eurycarpum	Giant burreed	9	14.00	5	0	50	50
Sparganium sp.	Burreed	3	4.00	1	0	11	11
Typha angustifolia	Narrow-leaf cattail	29	16.62	15	0	60	60
Typha latifolia	Broadleaf cattail	7	20.86	20	10	29	19

Table 13 continued

Taxon	Common name	*N*	Mean	Median	Min	Max	Range
Typha sp.	Cattail	1	3.00	3	3	3	0
Typha xglauca	Hybrid cattail	42	26.55	28	0	50	50
Utricularia cornuta	Horned bladderwort	1	1.00	1	1	1	0
Zizania aquatica	Annual wild rice	2	25.50	25.5	23	28	5
Zizania palustris.	Wild rice	79	35.19	33	1	91	90
Free-floating							
Azolla caroliniana	Mosquito fern	13	46.15	49	10	63	53
Lemna minor	Lesser duckweed	57	36.66	30	0	100	100
Ricciocarpus natans	Purple fringed liverwort	2	22.00	22	19	25	6
Floating rooted							
Brasenia schreberi	Water shield	15	47.13	46	16	88	72
Nuphar variegata	Common yellow pond lily	37	23.24	10	0	82	82
Nymphaea odorata	Fragrant water lily (white)	182	33.72	29	1	92	91
Nymphoides cordata	Little floating hearts	12	65.25	68	30	89	59
Potamogeton natans	Broad-leaved pondweed	8	49.50	46	11	91	80
Sparganium fluctuans	Floating burreed	15	53.60	58	34	69	35
Macroalgae							
Chlorophyta	Filamentous algae	53	45.91	45	15	88	73
Chara sp.	Muskgrass	31	48.61	46	3	91	88
Nitella sp.	Stonewort	4	44.75	32	24	91	67
Meadow							
Amblystegiaceae family	Moss	1	5.00	5	5	5	0
Asclepias incarnata	Swamp milkweed	1	0.00	0	0	0	0
Calamagrostis canadensis	Canada blue joint	6	0.00	0	0	0	0
Carex sp.	Sedge	97	4.58	0	0	45	45
Cirsium sp.	Thistle	2	0.00	0	0	0	0
Cuscuta gronovii	Swamp dodder	3	0.00	0	0	0	0
Dulichium arundinaceum	Three-way sedge	56	9.20	8	0	28	28
Epilobium ciliatum	Northern willow herb	1	28.00	28	28	28	0
Eupatorium perfoliatum	Boneset	6	0.33	0	0	1	1
Glyceria grandis	American manna grass	10	5.00	1	0	25	25
Impatiens capensis	Spotted jewelweed	18	6.22	0	0	45	45
Iris versicolor	Blue-flag iris	6	1.50	0	0	8	8
Juncus effusus	Soft rush	37	2.57	1	0	20	20
Juncus sp.	Rush	48	14.92	9	0	77	77
Lobelia cardinalis	Cardinal flower	1	0.00	0	0	0	0
Lycopus uniflorus	Northern water horehound	2	9.00	9	0	18	18
Lythrum salicaria	Purple loosestrife	3	3.33	5	0	5	5
Mimulus ringens	Square stemmed monkey flower	5	19.00	3	0	63	63
Myrica gale	Sweet gale	3	0.00	0	0	0	0
Onoclea sensibilis	Sensitive fern	5	0.00	0	0	0	0
Rorippa palustris	Marsh yellow cress	1	18.00	18	18	18	0
Rubus idaeus	Raspberry	1	0.00	0	0	0	0

Table 13 continued

Taxon	Common name	*N*	Mean	Median	Min	Max	Range
Salix sp.	Willow	1	0.00	0	0	0	0
Solanum dulcamara	Climbing night shade	2	1.50	1.5	0	3	3
Urtica dioica	Stinging nettle	5	9.00	0	0	45	45
Verbena hasta	Blue vervain	6	4.83	0	0	29	29
Submergent unrooted							
Ceratophyllum demersum	Coontail	29	56.03	55	28	88	60
Utricularia gibba	Creeping bladderwort	12	22.92	19	3	59	56
Utricularia intermedia	Flatleaved bladderwort	11	28.73	21	2	91	89
Utricularia purpurea	Purple bladderwort	9	51.89	47	34	81	47
Utricularia sp.	Bladderwort	1	38.00	38	38	38	0
Utricularia vulgaris	Common bladderwort	10	43.20	44.5	21	60	39
Submergent rooted							
Bidens beckii	Water marigold	5	50.60	40	30	86	56
Callitriche sp.	Water starwort	7	12.43	9	1	33	32
Elodea canadensis	Canadian waterweed	19	42.05	38	13	88	75
Eriocaulon aquaticum	Pipewort	44	26.82	25.5	0	70	70
Hippurus vulgaris	Mare's tail	4	31.00	34	18	38	20
Isoetes sp.	Quillwort	1	40.00	40	40	40	0
Myriophyllum heterophyllum	Two-leaf water milfoil	2	29.00	29	13	45	32
Myriophyllum sibiricum	Common water milfoil	7	36.71	38	10	58	48
Myriophyllum spicatum	Eurasian water milfoil	57	60.58	59	28	100	72
Myriophyllum tenellum	Slender water milfoil	6	69.50	76.5	21	89	68
Najas flexilis	Slender water nymph	60	50.87	47.5	13	91	78
Potamogeton amplifolius	Large-leaved pondweed	4	88.25	86	76	105	29
Potamogeton crispus	Curly-leaf pondweed	3	27.67	27	26	30	4
Potamogeton epiphydrus	Ribbon-leaf pondweed	2	39.50	39.5	33	46	13
Potamogeton filiformis	Thread leaf pondweed	3	28.00	28	26	30	4
Potamogeton foliosus	Leafy pondweed	12	31.42	29	23	55	32
Potamogeton gramineus	Variable pondweed	59	41.36	38	3	89	86
Potamogeton pusillus	Slender pondweed	2	48.00	48	46	50	4
Potamogeton richardsonii	Clasping-leaved pondweed	2	67.00	67	46	88	42
Potamogeton robbinsii	Fern-leaf pondweed	42	43.79	39.5	3	88	85
Potamogeton spirillus	Northern snailseed pondweed	10	41.10	38	15	76	61
Sagittaria graminea	Grassy arrowhead	71	31.76	29	0	77	77
Schoenoplectus subterminalis	Water bulrush	101	30.49	28	9	91	82
Stuckenia pectinatus	Sago pondweed	4	58.50	57	39	81	42
Utricularia subulata	Slender bladderwort	18	61.67	62	27	105	78
Vallisneria americana	Tape grass	20	62.65	63.5	30	105	75

Species have been divided into groups according to morphology. *N* indicates the number quadrats in which species were found

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