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A straightforward protocol for extracting microplastics from freshwater sediment with high organic content

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Abstract – Extracting microplastics from sediment is critical in assessing pollution in freshwater environments. However, this process can be particularly challenging for clayey sediments with a significant organic matter content. This study proposes a simplified method for extracting microplastics from organic-rich clayey sediments and a complete protocol for sediment processing. To evaluate the extraction method, calcined clayey sediment was artificially enriched with varying organic content (20, 40, and 60%). Known quantities of plastic particles with distinct characteristics and compositions (PET, HDPE, PP, PE, and PS) were introduced into these artificial organic sediments, mixed with a saline solution, centrifuged, and then assessed the recovery rate. The recovery rate exceeded 83% for the five selected plastic types. Additionally, the method minimized particle loss by reducing the number of transfers between containers, a common issue in other microplastic extraction protocols. Based on the efficiency of the extraction method, we proposed and applied a protocol for environmental samples. The protocol comprises five key steps: (1) drying sediment samples, (2) density separation, (3) filtering, (4) digestion of organic matter, and (5) visual analysis. This protocol extracted a wide variety of microplastics with diverse shapes, colors, sizes, and polymeric compositions. These results demonstrate that the proposed protocol is both straightforward and effective in extracting plastic particles commonly found in clayey sediments of freshwater systems. Moreover, the protocol employs inexpensive, readily available, and environmentally friendly reagents, making it a more sustainable alternative to other methodologies for extracting microplastics from environmental matrices.

Keywords: Microplastic pollution / freshwater sediment / density separation / sample processing

1 Introduction

Microplastics are small plastic particles (<5 mm) commonly found in aquatic environments (Thompson *et al.*, 2004). Aquatic contamination occurs from a wide variety of sources, including wastewater and larger plastic debris that degrades into progressively smaller particles (Backhaus and Wagner, 2020; Horton and Dixon, 2018; Murphy *et al.*, 2016). In recent decades, microplastics have emerged as a major global environmental concern due to their widespread presence in aquatic ecosystems and bioavailability. In contaminated environments, aquatic organisms are exposed to these plastic particles, leading to several adverse effects, including behavioral changes (Fu *et al.*, 2022; Im *et al.*, 2022), oxidative stress (Buwono *et al.*, 2022; Esterhuizen *et al.*, 2022), and impaired growth (Capolupo *et al.*, 2020). Consequently, the negative impacts on the freshwater populations affect the overall dynamics of these environments.

In freshwaters, microplastics are found in the water column (Egessa *et al.*, 2020; Sulistyowati *et al.*, 2022; Warrier *et al.*, 2022), adhered to organic surfaces, like roots of aquatic plants (Rozman *et al.*, 2022; van Weert *et al.*, 2019), and sediment (Kabir *et al.*, 2022; Peng *et al.*, 2018). Detecting microplastics in environmental samples poses a significant challenge to achieving a comprehensive understanding of microplastic contamination, particularly in sediments from freshwater environments. Sediment samples are complex matrices for analyzing the presence of microplastics, especially when they contain high levels of organic matter (Prata *et al.*, 2019). The organic content can accumulate on the surface of microplastics complicating sample processing and hindering the detection of these particles.

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Table 1. Characteristics and composition of microplastics used for the proposal extraction method.

Polymer	Source	Shape	Size	Color
PET	Plastic bottle	Fragment	0.5–1 mm	Blue
HDPE	Industrial pellets	Fragment	0.5–1 mm	White
PE oxo-biodegradable	Plastic bag	Film	0.5–1 mm	Yellow
PP	Rope	Line	0.5–1 mm	Red
PS	Styrofoam	Foam	0.5–1 mm	White

Several methods have been developed to extract plastic particles from sediments (Prata, da Costa, Duarte *et al.*, 2019; Quinn *et al.*, 2017). However, the main protocols are typically designed for sediments with low organic content or sandy/gritty granulometry. Many freshwater systems, particularly in tropical regions, have high organic matter content and fine granulometry (Guyot *et al.*, 2007). As a result, the protocols commonly used for extracting microplastics from sediment samples may not be efficient for organic-rich, clayey sediments, complicating sample analysis.

Understanding the levels of microplastic pollution is crucial for the conservation of aquatic ecosystems. Therefore, developing methodologies to facilitate the investigation of microplastics in environmental matrices should be a priority. In this context, the present study proposes a simplified, low-cost, and environmentally friendly method for analyzing microplastics in organic-rich, clayey sediment samples from freshwater environments.

2 Materials and methods

In the present study, we initially tested a method for microplastic extraction from sediment samples using centrifugation associated with saline solution. Based on the efficiency of this method, we proposed a complete protocol for the processing of environmental samples including the tested extraction method. Then, we applied this protocol to clayey sediment samples collected from a tropical reservoir assessing its efficiency for real samples. These procedures are described in the following sections.

2.1 Microplastic extraction combining centrifugation and saline solution

2.1.1 Microplastic particle preparation

Five types of plastic polymers from different sources were used in the present study: polyethylene terephthalate (PET), high-density polyethylene (HDPE), oxo-biodegradable polyethylene (PE-oxo), polypropylene (PP), and polystyrene (PS) (Fig. S1). PET and PS microplastics were obtained by grinding pieces of a plastic bottle and Styrofoam, respectively. Using scissors, PE-oxo and PP particles were obtained by cutting a plastic bag and a rope, respectively. HDPE pellets were obtained directly from the industry. The particles were classified considering their origin, shape, size, and color, as displayed in Table 1. The composition of the particles was determined from the FTIR Spectrometer analysis.

2.1.2 Sediment

Artificial samples containing clayey sediment and organic matter were prepared to represent an organic-rich sediment. The sediment was collected from the Billings Reservoir, São Paulo, Brazil, and processed in the laboratory by drying, grinding, and burning at 550 °C in a muffle furnace for 6 hours to remove organic matter and potential contaminants. The burnt sediment served as the control treatment (0% organic matter). For organic-rich treatments, various organic matter contents (20%, 40%, and 60%) were achieved by adding commercial fish food (Tetramin®) to the burnt clayey sediment. The fish food was previously pulverized using a mortar and pestle. The fish food containing 88.08 ± 0.22% organic matter (determined through calcination), was used to create organic-spiked sediment samples.

2.1.3 Microplastic extraction

The recovery protocol consisted of artificial organic-rich clayey sediment samples spiked with fragments of a single polymer type (PET, HDPE, PP, PS, or oxo-biodegradable PE). Briefly, 1 g of sediment-containing microplastics and 30 plastic fragments were added to a centrifuge tube. The procedure involved adding 10 mL of saturated NaCl solution (Sigma-Aldrich, 1.2 g/cm^3, w/v) to the tube, vortexing for 30 seconds, and centrifuging at 3500 rpm for 10 minutes. The supernatant was filtered using a glass microfiber filter (Whatman 47 mm, 0.7 μm pore size) and a glass vacuum filtration apparatus. The filters were dried at 60 °C and examined under a stereomicroscope to count microplastic particles.

NaCl solution was employed for density separation due to its efficiency, cost-effectiveness, and environmentally friendly properties, as reported in the literature (Browne *et al.*, 2011; Klein *et al.*, 2015; Quinn *et al.*, 2017).

2.2 A protocol for the processing of environmental samples

Once we observed the efficacy of the extraction method, we proposed a complete protocol for environmental samples. This proposed protocol comprises five steps including the sample drying process, particle extraction, filtering, digestion, and visual analysis, as described as follows:

- ***Clayey sediment drying:*** Using a metal spatula, approximately 20 g of clayey sediment previously homogenized was spread in thin layers on parchment paper (680 cm^2) or any non-plastic surface and dried at 60 °C on the stove for approximately 4 h. Depending on the layer thickness, a

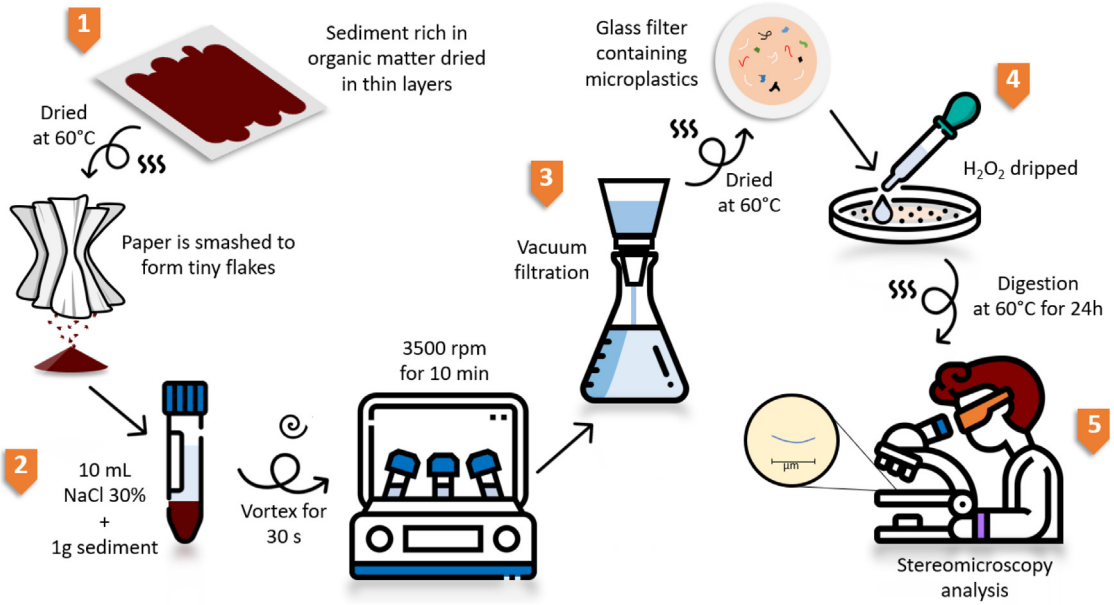

Fig. 1. The diagram of the sample processing protocol for extraction of microplastics from organic-rich clayey sediments.

longer time drying may be required. After drying, the parchment paper containing the dried sediment was gently smashed to form fine sediment. This process eliminates the necessity of mechanical action, e.g. maceration using mortar and pestle, to pulverize the sediment. A saturated solution of NaCl (Sigma-Aldrich, 1.2 g/cm^3, w/v) was used for density separation. The visual aspect of step 1 is displayed in Figure S2.

– ***Particle extraction in saline solution by centrifugation:*** 1 g of environmental sediment sample and 10 mL of NaCl solution (Sigma-Aldrich, 1.2 g/cm^3, w/v) were added to a centrifuge tube and shaken for 30 seconds using a vortex mixer to promote homogenization. The mixture was centrifuged at 3500 rpm for 10 min. Plastic centrifuge tubes can be used in this step, but some strategies need to be adopted, such as a control experiment to assess possible cross-contamination from the tubes. To avoid cross-contamination from the tubes, we recommend prioritizing glass tubes. However, it is important to verify the suitability of the glass tubes that will be used, because some tubes may not resist these centrifugation conditions.

– ***Filtering in a vacuum system:*** The supernatant is filtered using a glass microfiber filter (47 mm) with a glass vacuum filtration apparatus. The filter is abundantly rinsed using filtered ultrapure water (Whatman 47 mm, pore size 0.6 μm). After centrifugation, the supernatant needs to be immediately filtered. We recommend that the centrifugation step be repeated three times to enhance microplastic recovery. Between filtrations, the filtration system should be maintained covered to avoid contamination from the air. The same glass filter can be used for the three filtration steps, that is, one filter per sediment sample. A new filter could be used if the glass filter appears loaded. Then, the filters should be stored in Petri dishes and dried on a stove at 60 °C.

– ***Digestion method for organic matter removal:*** Organic matter digestion was performed using H_2O_2 (Sigma-Aldrich). Drops of H_2O_2 were carefully added to the dried filter at a volume sufficient to embed the filter without promoting overflow of the H_2O_2 (approximately 0.5 mL) and consequently dragging particles. The filter was placed on a stove and maintained for 12 hours. After this period, we recommend a rapid inspection under stereo microscopy to verify whether the organic content was digested, easily identified by the loss of dark color, and disaggregation of remaining small agglomerates. If necessary, the digestion can proceed for an additional 12 hours by adding some drops of H_2O_2.

– ***Visual analysis:*** The entire filter was analyzed under a stereo microscope (50× magnification) to count and identify all microplastic particles present in the sample that were characterized according to their shape, size, and color. We suggest the analysis of the entire filter because the distribution of particles is not homogeneous due to the morphological diversity of microplastics. After the visual identification, it is highly recommended that the chemical characterization be carried out using analytical methods already established in the polymer literature, such as spectroscopic and thermal analysis (Huang *et al.*, 2023).

Figure 1 summarizes the 5-step protocol for extraction of microplastics from sediment samples.

The visual aspect of steps 2, 3, and 4 is displayed in Figure S3. The description of the technique for visual inspection of the entire filter is shown in Figure S4. Additionally, a control experiment following all the proposed steps is essential. The control experiment will allow us to determine the levels of crossing contamination to which samples are susceptible along the experiments.

2.3 Application of the proposed protocol on environmental samples

To evaluate the protocol's efficiency in detecting microplastics in environmental samples, the methodology was applied to two organic-rich clayey sediment samples. The samples were collected in 2022 from the Billings Reservoir in São Paulo, Brazil (–23.810601; −46.535413). This freshwater reservoir is an important aquatic ecosystem supplying water to the Metropolitan Region of São Paulo but is frequently impacted by domestic and industrial effluents (Gargiulo *et al.*, 2022; Milz *et al.*, 2022). Sediment samples were collected using a grab sampler and then transferred to 100 mL glass flasks previously cleaned with acetone and stored at 4°C until further analysis. The sediment samples comprised the superficial sediment layer (up to 5 cm). The organic content of the sample was determined at 44.7% by calcination and classified as clayey due to the high clay content (97.3%). The humidity was determined at 81.9%.

In the laboratory, the sediment was dried at 60 °C in thin layers. Subsequently, 10 g of dried sediment was divided into 10 tubes (1 g per tube), each filled with 10 mL of saturated NaCl solution. The sample processing followed the protocol described in Section 2.3. Filters were analyzed under a stereomicroscope (Zeiss) for physical identification (size, shape, and color) and particle counting. To avoid biased results, we recommend visually analyzing the entire filter, as the distribution of microplastics across the filter surface is not uniform. A simple technique was devised to guide filter analysis under the stereomicroscope: the filter was placed in a Petri dish with pre-drawn lines to assist in systematically scanning the entire surface (Fig. S3).

The plastic particles were classified according to their shape, size, and color. The following shape groups were adopted: fibers, fragments, films, and foams (Tab. S1). Regarding color, particles were grouped based on their predominant color. The color identification was performed under visual analysis. Transparent, grayish, or organic-matter-colored particles were classified as "colorless". The colorless class was adopted due to the possible loss of pigmentation after the sample processing. Only microplastics larger than 100 μm were considered for this study to ensure reliable identification. The size measurement was performed using the measurement tools in Zen 3.6 software, and filters were analyzed in their entirety to characterize all particles retained on the filters.

For chemical characterization, 42 particles larger than 200 μm were randomly selected from the filters. The minimum size of characterized particles (>200 μm) was chosen to allow for handling and transfer for chemical analysis. Particle composition was determined using a Micro-Raman Renishaw inVia spectrometer ($\lambda_0 = 532$, 633, and 785 nm) with an Olympus 100× objective with 0.85 of numerical aperture.

2.4 Quality assurance and quality control procedures

To minimize potential sample contamination, quality assurance, and quality control (QA/QC) procedures were applied at all stages of the present study. All water and solutions were filtered with a Whatman® glass microfiber filter (47 mm, 1.6 μm pore size) before use. The collection instruments and laboratory glassware were washed using filtered ultrapure water, and cleaned acetone before use. During sample processing, the laboratory door remained closed to reduce airborne contamination sources, and only 100% cotton attire was worn at all stages of this work to avoid contact with external synthetic fibers. Samples were always covered with aluminum foil to prevent airborne contamination during collection, processing, and analysis. After vacuum filtration, filters were stored in closed Petri dishes and dried at 60 °C before analysis. All sample processing was carried out in a fume hood with the airflow activated. A control experiment following the same procedures without sediment was performed. NaCl solution (10 mL) was centrifuged, filtered in a glass microfiber filter, and then analyzed in stereomicroscopy. We detected 7 colorless fibers (100–300 μm) which were subtracted from the total count of transparent fibers obtained in each sediment sample.

2.5 Data analysis

Microplastic recovery data was expressed as the mean percentage ± standard deviation. Data normality was assessed using the Anderson-Darling test ($p > 0.05$). Significant differences were verified using the Fisher one-way ANOVA test ($p > 0.05$). The abundance of microplastics and polymers in environmental samples was expressed in percentage (%). Statistical tests were performed using Minitab 19 software.

3 Results and discussion

3.1 Microplastic extraction

The proposed method aimed to minimize particle loss during sample processing by reducing transfers of samples between containers. It also uses low-cost, easy-to-obtain, and eco-friendly reagents, such as NaCl and H_2O_2. The protocol comprised five key steps: (1) drying sediment samples, (2) density separation using saline solution, (3) filtering, (4) digestion of organic matter using H_2O_2, and (5) visual analysis. The compliance of these steps ensures high efficiency of microplastic recovery, as demonstrated by the validation results described in the present study.

Commonly, the environmental sediment samples are dried in an oven until complete humidity removal (Gerolin *et al.*, 2020; Zhang *et al.*, 2024). However, with organic-rich clayey sediment, this process must be adapted. Drying clayey sediment often results in a rigid mass (Fig. S5) that requires high mechanical force to break apart, which can fragment microplastic particles, reduce their original size, and generate new particles. This damage on microplastics can be more evident in weathered particles found in environmental samples which can be more fragile than pristine particles. Such outcomes can overestimate microplastic concentrations. Here, drying sediment in thin layers was a critical step to preserve plastic particles, facilitate sample processing, and ensure that particles were not trapped within sediment clumps.

3.2 Microplastic recovery

The density separation using a NaCl-saturated solution associated with centrifugation promoted high recovery rates for plastic particles (Fig. 2). The organic content of the

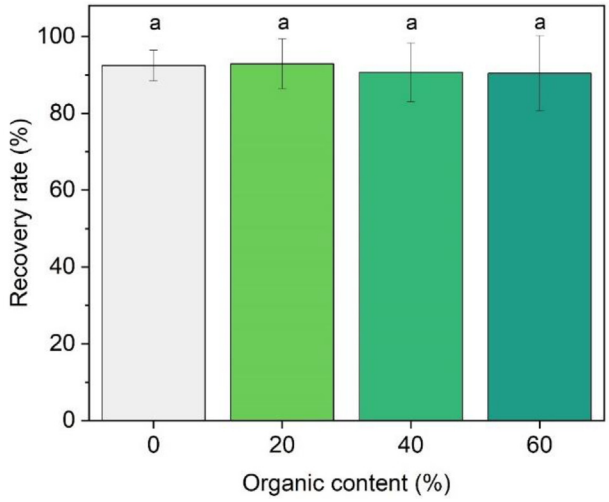

Fig. 2. Microplastic recovery rate (%) for different organic contents (0, 20, 40, and 60%). Equal letters indicate no significant difference between organic treatments (0, 20, 40, and 60%) (Fisher's test, $p > 0.05$). The analysis was performed in triplicate ($n = 3$).

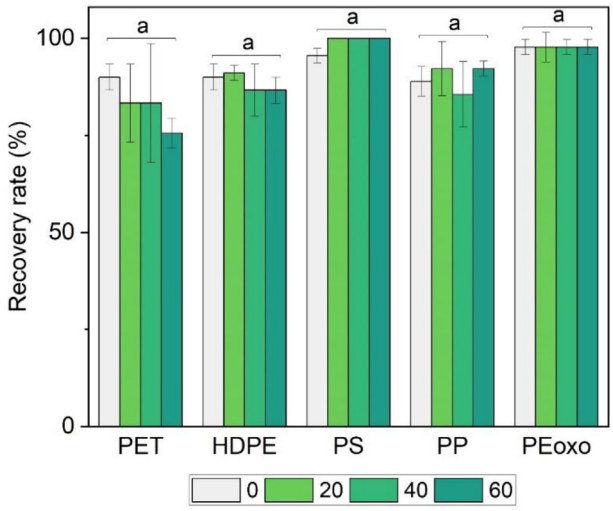

Fig. 3. Recovery efficiency (%) for each polymer tested (PET, HDPE, PS, PP, and PE oxo-biodegradable) in different organic contents. Equal letters indicate no significant difference between organic treatments (0, 20, 40, and 60%) for each polymer (Fisher's test, $p > 0.05$). The analysis was performed in triplicate ($n = 3$).

sediment did not significantly affect the microplastic recovery using the proposed method. Recovery rates in organic clayey matrices ranged from 90.44 ± 9.80 to 92.44 ± 3.96% (Fig. 2). No significant differences in microplastic recovery were observed among the treatments with varying organic content ($p > 0.05$).

Among the tested polymers, PS particles showed the highest recovery rate (98.89 ± 2.22%) across all treatments. PE oxo-biodegradable particles had a recovery rate of 97.78%. The average recovery rates for PP and HDPE were 89.72 ± 3.19% and 88.61 ± 2.29%, respectively. The lowest recovery rate was observed for PET particles (83.06 ± 5.91%). While no significant differences were found, PET recovery tended to decrease as organic matter concentration increased, possibly due to interactions between the particles and the organic matter (Fig. 3).

Considering all polymers, the method achieved an average recovery rate of 91.61 ± 6.97%. No significant differences were observed in any treatment (Fisher's test, $p > 0.05$).

Numerous methods have been proposed for extracting microplastics from sediments, each with varying recovery efficiencies. For instance, Semmouri *et al.* (2023), reported a recovery efficiency of 82 ± 4% for PE (90–106 μm) using a multi-step method that requires large reagent volumes and sodium iodide (NaI), which poses environmental and health risks. Similarly, Constant *et al.* (2021) identified NaI as effective for extracting PET, polyethersulfone (PES), and polyamide (PA) (71.17% recovery). Zinc chloride ($ZnCl_2$) has also been reported as a good extractor, with a recovery efficiency of 96.84% (Rodrigues *et al.*, 2018). Here, we reported an average recovery above 91.61% for 5 polymers (PET, HDPE, PS, PP, and PEoxo). Although NaI and $ZnCl_2$ have been used for microplastic extraction, these salts are toxic, and their use carries safety risks and represents a problem for discharge of the residuals generated along the study. Also, these salts are more expensive representing an issue for the development of wide studies. Our protocol offers a simplified procedure with fewer steps, smaller reagent volumes, and a saturated NaCl solution, which is safer, low-cost, and more environmentally friendly (Cutroneo *et al.*, 2021).

We observed that the proposed protocol shows a similar recovery compared with other methods using NaCl solution for extraction. Monteiro and Costa (2022) summarized findings in the literature about microplastic recovery from solid matrices and determined an average recovery rate by NaCl of 81%. These authors highlight that NaI and $ZnCl_2$ show higher recoveries for high-density polymers but also indicate that these reagents are harmful to the environment.

3.3 Organic matter reduction

Removing organic matter is a crucial step for microplastic analysis of environmental sediment samples (Monteiro *et al.*, 2022). High organic content can hinder visual analysis due to the darker coloration of the sample, making microplastic detection and extraction more challenging. Organic matter can also form a layer on top of them altering the surface characteristics. This layer compromises polymer identification generating fluorescence signals during Raman spectroscopy. Despite its importance, the choice of organic matter removal method requires careful consideration. Reagents used for digestion have limitations, as prolonged exposure can degrade plastic particles. When combined with high temperatures, the impact on particle structure becomes even more pronounced (Monteiro *et al.*, 2022; Pfohl *et al.*, 2021).

Potassium hydroxide (KOH) may not be ideal for clayey samples with high organic content. Digestion with KOH often increases turbidity and darker coloration compared to untreated samples (Fig. S6). This occurs because alkaline hydrolysis is inefficient in removing humins, the most common organic portion in soils, that have a dark color

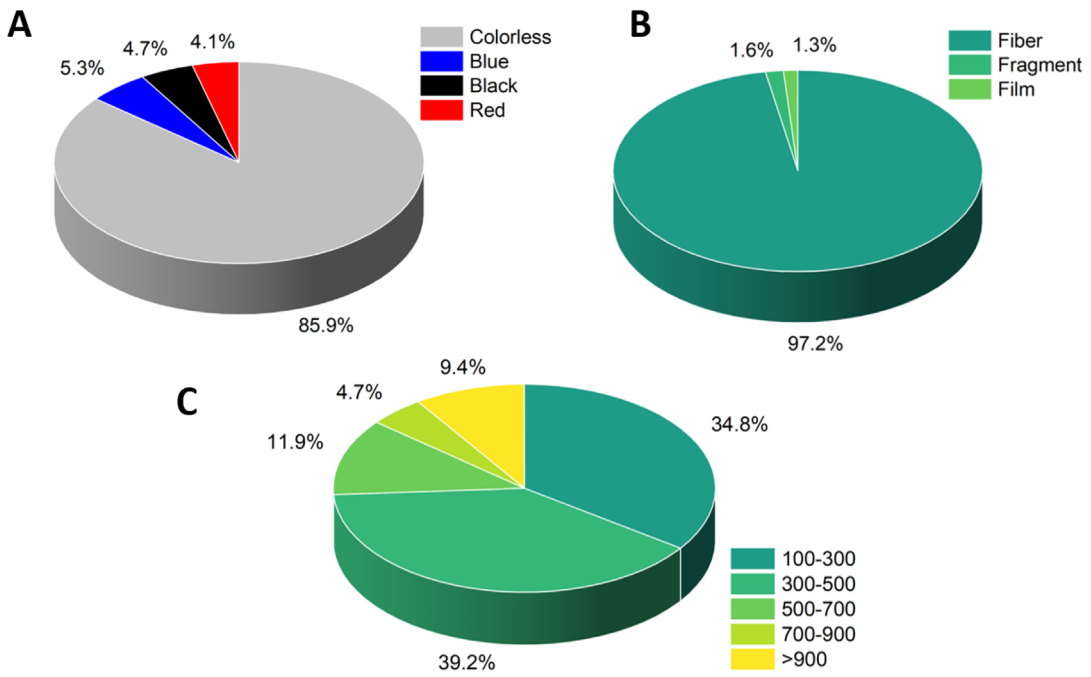

Fig. 4. Relative abundance (%) of plastic particles detected in environmental samples. Microplastics were categorized by color (A), shape (B), and size (C). All detected plastic particles (319) were considered.

(Schnitzer, 1982, Almendros and González-Vila, 1987). After filtration, such samples produce filters with intense dark coloration, making it difficult to detect plastic particles under a microscope, particularly black particles. Consequently, digestion before filtration can significantly hinder visual analysis. Based on these characteristics, some studies have indicated alkaline digestion as more suitable for biological samples (Blasing and Amelung, 2018).

Our protocol addresses these challenges by eliminating issues associated with sample turbidity. Centrifugation combined with a saline solution precipitates organic matter. Following this, we use H_2O_2 at the end of the process. The filter containing plastic particles with reduced organic content is treated with H_2O_2 and maintained at 60 °C for 24 h. Some sediment samples may contain a high prevalence of insect and plant fragments, which naturally separate from the sediment during density extraction due to differences in density. However, if their presence persists and complicates microplastic detection on the filter, an additional H_2O_2 digestion step can be performed. This approach ensures mild digestion, reducing the potential damage to polymers while simplifying microscopy analysis. Additionally, this protocol significantly reduces reagent consumption, making it cost-effective.

3.4 Microplastic extracted from environmental samples

The proposed method described in Section 2.3 was applied to environmental clayey sediment samples collected from a tropical reservoir. The microplastic concentration in each sample was determined at 11800 and 20100 particles kg^{-1}. A total of 319 particles were detected in the two environmental samples (10 g per sample) collected from the tropical reservoir. Fibers corresponded to 80.4% of the total extracted particles. Colorless particles were predominant (85.9%) as well as microplastics with sizes varying between 300 and 500 µm (39.2%) (Fig. 4). Our findings align with the literature which also reports a predominance of fibers and particle sizes between 100 and 500 µm in environmental samples (Akdogan *et al.*, 2023, Salikova *et al.*, 2024, Zakiah *et al.*, 2024). These results demonstrate that the proposed protocol successfully extracts a wide range of microplastic shapes and sizes.

Raman microscopy identified a wide variety of polymers in the sediment samples. The most representative polymer was PP (31.0%) followed by PE, PC, Nitrocellulose, PET, Polyester, PA, ABS, and viscose (Fig. 5). Notably, the detection of six additional polymers (PC, Nitrocellulose, Polyester, PA, ABS, and viscose) not explicitly targeted in this study highlights the protocol's efficacy in recovering microplastics with diverse polymeric compositions.

4 Conclusions

The present study introduces a straightforward protocol for extracting microplastics from freshwater sediment samples with high organic matter content. The results contribute to discussions on strategies for predicting the occurrence and transport of microplastics in environments influenced by organic matter. The five-step method developed in this study demonstrated efficiency in extracting microplastic particles from clayey organic sediments, achieving recovery rates above 83.1%. The processing preserved plastic particles from mechanical damage. The combination of centrifugation and saline solution effectively removed organic content, simplifying or even eliminating the need for digestion steps. The high efficiency of the proposed method is attributed to reduced particle loss through minimized sample transfers, lower reagent consumption, cost-effectiveness, high recovery rates, and straightforward execution. This protocol represents a

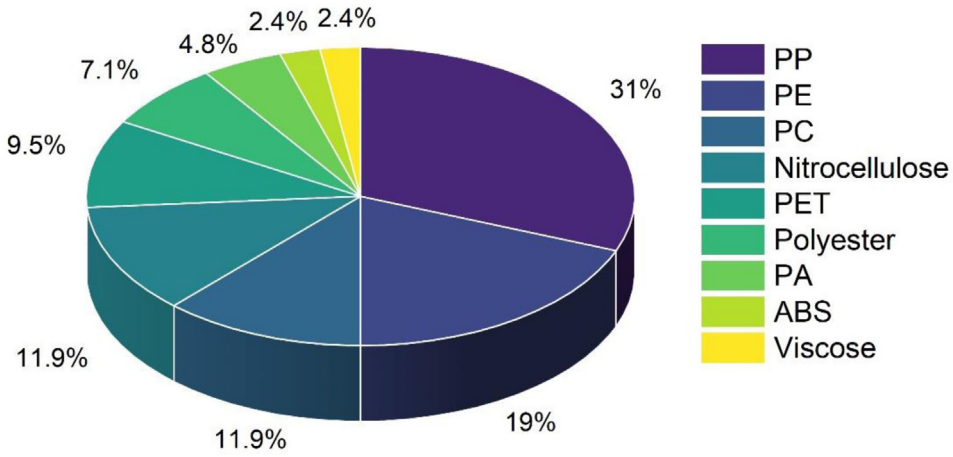

Fig. 5. Polymer abundance (%) of microplastics detected in environmental sediment samples from a tropical reservoir. Fourty two particles ($n = 42$) were analyzed for chemical characterization.

valuable tool for advancing research on microplastic pollution in complex environmental matrices.

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Conflicts of interest

The authors declare that they have no known competing financial interests or personal relationships that could have appeared to influence the work reported in this paper.

Data availability statement

The data that support our findings are available from the corresponding author upon reasonable request.

Author contribution statement

Lucas Gonçalves Queiroz: Conceptualization, Methodology, Formal analysis, Data curation, Investigation, Visualization, Writing – original draft, Funding acquisition. **Luana de Oliveira Hallai:** Formal analysis, Investigation, Data curation. **Beatriz Moraes:** Formal analysis. **Rômulo Augusto Ando:** Formal analysis, Funding acquisition. **Marcelo Pompêo:** Supervision, Writing – review and editing, Funding acquisition. **Bárbara Rani-Borges:** Supervision, Methodology, Writing – review and editing.

Supplementary material

Figure S1. Microplastics used in the proposed extraction method. (A) Polystyrene (PS), (B) Polyethylene terephthalate (PET), (C) Polypropylene (PP), (D) Oxo-biodegradable polyethylene (PE-oxo), (E) High-density polyethylene (HDPE). Scale bars correspond to 1 mm.

Figure S2. The visual aspect of the sediment sample during the drying process and formation of fine flake sediment following the proposed method.

Figure S3. The visual aspect of the sediment sample during the density separation process and organic matter digestion following the proposed method.

Figure S4. Simple technique to analyze the entire filter. Petri dish containing lines to guide the filter analysis (A). Scheme demonstrating the path taken on the filter surface (B). Region of the filter containing the guide-lines (arrows) and a microplastic fiber (circle).

Figure S5. Sediment samples after the proposed drying process (left) and conventional drying process forming a solid mass (right).

Figure S6. Different characteristics of sediment sample after 24h of digestion using KOH (left) and H_2O_2 (right).

Table S1. Categories of microplastics used to describe particles identified in the environmental sample.

The Supplementary Material is available at https://www.kmae-journal.org/10.1051/kmae/2025001/olm.

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