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Research Article

Microplastics in the indoor laboratory environment: A study on deposition rate, size, shape, polymer, and source

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Abstract

This study examined the abundance, size, shape, color, and polymer composition of microplastics (MPs) in a laboratory environment. The deposition rate of MPs ranged from 178 to 427 particles per square meter per day (p/m²/d), with an average of 286 ± 104 p/m²/d. Fragments were the most common shape, followed by pellets and fibers. Most MPs were found in the 53-300 µm and 300-500 µm size ranges. Micro-Fourier Transform Infrared Spectroscopy (FTIR) analysis identified polyethylene (PE), cellulose nitrate (CN), polypropylene (PP), and cellulose, with PE being the most prevalent polymer. The color analysis revealed that white and black were the most common colors, with PE mostly white, PP brown, and cellulose and CN appearing in various colors. The identified polymers suggest that the MPs found in the laboratory likely originate from routine laboratory activities, including handling of reagent bottles and caps, as well as from materials used in laboratory infrastructure such as walls, furniture, air conditioner (AC), and curtains.

Keywords: Deposition rate, fragments, laboratory environments, micro-FTIR, microplastics, polymers

Introduction

Plastic particles less than 5 mm in diameter are MPs (Arthur et al., 2009). MPs are divided into two categories, namely primary and secondary, based on the method of formation. Primary MPs are intentionally shaped into microscopic sizes as plastic microbeads, pellets, or resins (Cole et al., 2011; Filella, 2015). Personal care products, facial products, and air-blast cleansing are the major sources of primary MPs (Cole et al., 2011). Secondary MPs are formed through the degradation of larger plastic items into fibers and fragments (Filella, 2015). Similarly, based on morphology, MPs can be categorized into five major classes (Anderson et al., 2017). They are fragments, pellets, fibers, films, and foams. MPs vary in chemical composition, shape, size, color, and density, and these features strongly influence the abundance and concentration of MPs in the environment (Amato-Lourenco et al., 2020; Rocha-Santos & Duarte, 2015). Most commonly observed shapes around the globe are fibers, fragments, and films (Wagner & Lambert, 2018). Among them, micro-fibers are the dominant shapes coming from textiles, garments, fragmented nets, fishing lines, and other plastic materials (Barrows et al., 2018; Cole et al., 2011; De Falco et al., 2020). PP, PE, polyamide (PA), and polyethylene terephthalate (PET) are the most observed polymer types around the globe (Carr, 2017).

In Wenzhou, China, indoor airborne MPs concentrations were found to be more than eight times higher than those outdoors (Liao et al., 2021). This suggests that indoor environments may have significantly higher levels of MPs, and future research should focus on studying MPs in such settings. The literature indicates that both the composition (polymer type and shape) and abundance of MPs in indoor environments vary across studies. In China, indoor MPs deposition rates as high as 9.3772 E^4 to 3.11040 E^5 p/m²/d have been reported, whereas Brazil recorded a much lower deposition rate of 309.40 ± 214.71 p/m²/d (Amato-Lourenço et al., 2022; Cui et al., 2022). Fibers are the most reported form of MPs found in indoor environments (Abad-López et al., 2024; Dris et al., 2017; Jenner et al., 2021; Soltani et al., 2021; Zhang et al., 2020b). PP, PE, PA, polyester (PES), and PET are the most common indoor polymers (Abad-López et al., 2024; Jenner et al., 2021; Liao et al., 2021; Soltani et al., 2021; Yao et al., 2022; Zhang et al., 2020b). Many factors may influence the abundance and properties of MPs indoors. Human activities have been shown to influence the number of MPs in indoor environments (Dris et al., 2017; Jenner et al., 2021; Zhang et al., 2020b). While Liao et al. (2021) and Jenner et al. (2021) found no link between indoor and outdoor MP abundance, Soltani et al. (2021) observed an association between the number of MPs and the ventilation rate of the room. Furthermore, a study showed that MPs may be efficiently released into interior air through AC filters lined with PE and PET, spreading them at different distances from the AC units (Chen et al., 2022). In another study, higher concentrations of MPs were observed in households with greater use of electronic devices (Bhat, 2024). Similarly, a study by Dahal and Babel (2024) detected polymers and copolymers commonly used in paints and coatings indoors. The same authors proposed that potential sources of indoor MPs include the deterioration of polymer-based paints and coatings used on walls, furniture, and electronic devices such as cellphones, desktops, and tablets. Thus, a variety of factors may influence the abundance of MPs in indoor environments.

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The sampling method should be robust and scientifically valid to ensure reliable results. Before sampling, the sampling site, height, duration, and frequency should be clearly defined (Chen et al., 2020; Dahal & Babel, 2026). Sampling should be conducted at least three times to ensure data reliability (Allen et al., 2019; Dahal & Babel, 2026) and to enable statistical analysis for assessing data variability and significance. Moreover, a treatment and extraction method with a high recovery rate should be employed to maximize the reliability of MPs enumeration and sorting. In addition, using spectroscopic techniques for polymer identification would improve data accuracy (Chen et al., 2020). To ensure the accuracy and reliability of the study, possible contamination during experimental procedures should also be prevented. The results showed that laminar flow is the most effective choice for MPs analysis, as it reduces blank levels better than a fume hood or a standard laboratory environment (Bhat et al., 2024). However, background contamination from ubiquitous MPs cannot be ignored during sample handling and processing in the laboratory environment. Therefore, procedural blanks should be processed in parallel with samples to ensure reliable estimation of environmental contamination levels.

Although some studies have included blank samples, most were either omitted or inadequately reported blank corrections in their final analyses (Bhat, 2023). Consequently, concerns arise regarding potential contamination from atmospheric deposition of MPs in laboratory environments, particularly due to the extensive use of plastic-based consumables. As a result, a methodological gap persists in adequately addressing laboratory-derived background contamination in MPs research. Furthermore, MPs present in laboratory environments may also pose occupational health and safety risks, as laboratory personnel can be exposed through inhalation of airborne particles or dermal contact during prolonged work. This study was therefore designed to address these gaps by assessing the abundance and characteristics of MPs in a laboratory environment. The objectives of this study were: (1) to examine the deposition rate of MPs within the laboratory, (2) to characterize the identified MPs in terms of shape, size, color, and polymeric composition, and (3) to identify possible sources of MPs in the laboratory environment. The findings are expected to highlight the importance of incorporating quality control measures, such as laboratory blanks, to improve data reliability and support the inclusion of MPs in laboratory occupational health and safety assessments.

Materials and Methods

Study area

To understand the MPs deposition rate in the laboratory setting, the laboratory at Sirindhorn International Institute of Technology, Thammasat University, Thailand, was selected as a sampling site for this research. The laboratory expanded in an area of approximately 5 m × 20 m and was one of the busiest environmental research laboratories. During the sampling period, it was used by around 5-7 people daily, with working hours from 9:00 AM to 4:00 PM. The laboratory was air-conditioned throughout the day, and no manual ventilation was used. It had separate collection bins for the disposal of hazardous and non-hazardous chemical bottles.

Sample collection

This study adopted the methodology developed by Dahal and Babel (2024) for analyzing atmospheric MPs. The laboratory deposition was assessed using a passive sampling technique. A sampler consisting of a 10-cm glass funnel connected to a 3 L glass jar was used for collecting atmospheric deposition. The sampler was placed on the working table at approximately 1.15 m above the floor to evaluate the MPs deposition rate during sample processing and handling. Duplicate samples were collected monthly over three months. One set was designated for Nile Red (NR) staining, and the other for micro-FTIR examination. In total, 6 samples were collected from the laboratory between February 13 and May 30, 2022, resulting in a sampling duration of 101 days. On each sampling occasion, 1 L of deionized water (DI) was used to rinse the funnel, transferring the deposited particles into the glass jar. The jar containing the rinsed sample was immediately replaced with a new one. It was sealed with aluminum foil and stored in the laboratory for subsequent analysis.

Sample extraction and treatment

The samples were filtered using a 53 µm sieve. The particles retained on the sieve were rinsed into a 250 mL beaker using DI water and oven-dried at 60 °C. After drying, the dried samples were subjected to digestion using Fenton's reagent (40 mL of hydrogen peroxide (H_2O_2) and 1 mL of Fe (II)) following a two-step process: a 30-minute heated reaction at 40 °C to remove organic matter, followed by 24 hours of further digestion at room temperature. The digested samples were re-sieved through a 53 µm sieve and thoroughly rinsed with DI water to remove residual H_2O_2. The final residue was transferred to a 250 mL beaker and oven-dried at 60 °C for further analysis.

For density separation, 40 mL of sodium iodide (NaI) solution was added to the beaker. The mixture was transferred to a 50 mL glass cylinder, and the beaker was rinsed with 15 mL and 10 mL of NaI solution to recover any remaining particles. The suspension was left undisturbed for 24 hours to allow density separation. Afterward, the floating particles were carefully decanted into a 250 mL beaker for further examination.

NR tagging for microscopic analysis and enumeration

Following density separation, the samples were sieved using a stacked series of 500, 300, and 53 µm sieves. The particles retained on each sieve were rinsed separately into a 250 mL glass beaker using DI water, ensuring independent collection of each size fraction. Each fraction was then individually filtered onto cellulose nitrate membrane filters (0.45 µm pore size, 47 mm diameter). NR staining was performed to identify and count MPs. NR, a dye that adsorbs onto plastics and fluoresces under ultraviolet (UV) light, facilitates the differentiation of plastics from non-plastics. A mixture of NR and chloroform (1:1) was prepared as the staining solution. Approximately 1 mL of this solution was evenly applied to the filter surface using a micropipette. The filter was then left undisturbed for 30 minutes to allow adsorption. The staining procedure was repeated to enhance NR adsorption and maximize MPs recovery.

The stained filters were examined using a fluorescence microscope. The MPs were manually counted, and blank corrections were applied. Deposition rates were calculated and normalized to a monthly basis, and reported as p/m^2/d. MPs were categorized based on shape as fibers, fragments, or pellets. Fibers were long and thin, pellets were round/spherical, and fragments were irregular in shape. MPs were further categorized into three size classes: 53-300 μm, 300-500 μm, and >500 μm.

Polymeric identification

From each filter, 10 visually suspected MPs were randomly selected for polymer identification, following the method of Dahal and Babel (2024), which has also been applied in a recent study by Pandit et al. (2025) and recommended by Dahal and Babel (2026). This identification was conducted using micro-FTIR (Nicolet iN10, Thermo Scientific, USA) coupled with OMNIC™ Picta™ software. The instrument was equipped with a cooled MCT-A detector to enhance sensitivity. Spectral acquisition was conducted over the range of 500-4000 cm^{-1} with 128 scans at a resolution of 8 cm^{-1} and an acquisition time of 25 seconds per measurement. An adjustable aperture (50-250 μm) in both height and width was used to optimize microscopic analysis.

The resulting FTIR spectra were recorded and compared with reference libraries to identify the polymer type. A similarity threshold of 70% or higher was used to classify particles as a synthetic material (Dahal & Babel, 2024, 2026). A total of 30 particles were analyzed to identify the polymeric composition of the MPs.

Quality control

All the equipment was thoroughly washed with DI water before use. The use of plastic items was minimized to prevent contamination. Samples were handled and processed using a cotton coat and latex-free gloves. To further minimize background contamination, all sample processing was conducted within a fume hood. The use of a fume hood has been reported to reduce contamination by 61.3% (Bhat et al., 2024). Three procedural blanks were prepared to assess background contamination in the laboratory. Each procedural blank consisted of 1 L of DI water processed in the same manner as the samples. These accounted for potential sources such as atmospheric deposition during sample handling and fluorescence analysis, DI water, and reagents including NaI, H_2O_2, and NR. Blank values were subtracted from the sample results.

Results and Discussion

Abundance of MPs

The laboratory deposition rates were 178, 427, and 254 p/m^2/d during the first, second, and third sampling periods, respectively, with an average of 286 ± 104 p/m^2/d. Fragments dominated all other shapes during all sampling periods (Fig. 1). Pellets were the second most repeatedly observed shapes after fragments. The average abundances of fragments, pellets, and fibers were 70%, 20%, and 10%, respectively.

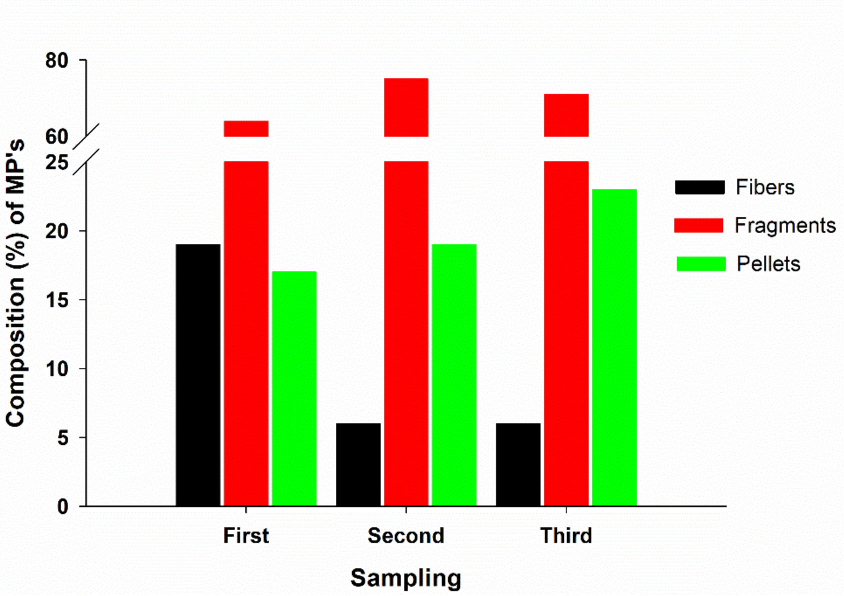

Figure 1 Abundance of fragments, fibers, and pellets during each sampling event

The average laboratory deposition was approximately 12 particles per square meter per hour (p/m^2/h) (converted from daily value), ranging from 7 to 18 p/m^2/h. This rate indicates that laboratory workspaces can be heavily contaminated with MPs even during short-term exposure. Therefore, maintaining clean workspaces and laboratory utensils is essential to reducing background contamination. Laboratory deposition may contaminate samples during sieving, digestion, and density separation if these processes are not conducted in a fume hood. In addition, airborne MPs concentrations may vary temporally and spatially within laboratory environments, increasing the risk of inconsistent exposure and cross-contamination between samples.

However, previous studies (Table 1) did not simultaneously implement key quality assurance and quality control (QA/QC) measures, including laboratory blanks, field blanks, and controlled processing conditions such as the use of a fume hood. This indicates a lack of standardized QA/QC protocols across studies, which limits the comparability of MPs deposition data between studies. As a result, reported deposition rates in those studies may not fully represent true sample contamination and may include background contributions from the laboratory environment. This raises concerns about the scientific robustness of such studies. In this regard, conducting blank analysis, distinguishing between procedural and field blanks, using pre-cleaned utensils and workspaces, and operating under clean-air or fume hood conditions should be considered essential methodological practices in MPs studies.

Table 1 Quality control measures adopted in studies analyzing indoor and outdoor MPs

Blank Samples	Blank type	Blank value	Fume hood	References
2	Laboratory	0	No	(Abbasi et al., 2019)
2		0	No	(Abbasi & Turner, 2021)
6	L & P	3 ± 1 fibers, 1 ± 1 films, 8 ± 1 fragments/filter	No	(Allen et al., 2019)
No	No	No	No	(Cai et al., 2017)
No	Laboratory	0	No	(Ding et al., 2021)
N0	L & P	0-2 fibers/filter	No	(Dris et al., 2015)
No	L & P	No	No	(Gonzalez-Pleiter et al., 2021)
4	L & P	0	No	(Huang et al., 2021)
12	L & P	7.3 ± 4.8 particles/filter	Yes	(Klein & Fischer, 2019)
No	Procedural	0	Yes (partially)	(Liu et al., 2019a)
No	Procedural	0	No	(Liu et al., 2019b)
No	No	No	No	(Narmadha et al., 2020)
No	Procedural	0	No	(Patchaiyappan et al., 2021)
15	L & P	0.9 fibers per hour	No	(Roblin et al., 2020)
32	L & P	5.6 MPs/kg	No	(Su et al., 2020)
5	Laboratory	1-3 fibers/filter	No	(Szewc et al., 2021)
No	Laboratory	No	No	(Truong et al., 2021)
42	Procedural (site only)	No	No	(Wang et al., 2020)
1	L & P	3 fibers/filter	Yes	(Wright et al., 2020)
No	No	No	No	(Dris et al., 2017)
144	Laboratory	0.2 ± 0.4 particles/sample	No	(Jenner et al., 2021)
30	L & P	3.3 ± 1.8 particles/filter	No	(Liao et al., 2021)
9	L & P	0-3 fibers/filter	No	(Soltani et al., 2021)

Note: laboratory blanks represent blanks taking account of airborne MPs in the lab. Procedural blanks represent blanks that have undergone the same chemical treatment processes as samples to identify MPs originating from these processes. L&P represents blanks, considering both air-borne MPs and MPs originating from chemical treatment processes/blanks samples treated in the same way as samples in the lab. "No" means information not available or steps not considered.

Table 2 shows the abundance and characteristics of indoor MPs reported in previous studies. The characteristics of MPs observed in the laboratory, including their count, shape, polymer type, color, and size, are comparable to those reported in indoor environments using passive sampling. However, the deposition rate observed in this study was higher than that reported for indoor air in Bangkok, Thailand (Dahal & Babel, 2024), but slightly lower than that reported in São Paulo, Brazil (Amato-Lourenço et al., 2022). Several studies have reported MPs deposition rates exceeding 1,000 p/m^2/d, which may be attributed to differences in detection limits, as well as the greater diversity and intensity of indoor MPs sources compared to a controlled laboratory environment.

Most previous studies (Table 2) have reported either fibers or fragments as the dominant shape. This variation is likely due to methodology differences, such as visual counting versus NR staining techniques (Dahal & Babel, 2024). In this study, fragments were the dominant shape. Fragments are generally formed through the breakdown of larger plastic items (Browne, 2015; Koelmans et al., 2015) and may also originate from single-use plastics or thicker recyclable materials (Cai et al., 2017). However, fragmentation of larger plastics items in the indoor laboratory setting is unlikely. The dominance of fragments in this study, therefore, suggests alternative sources. For instance, handling processes such as opening and closing reagent bottles, whether plastic or glass, may generate MPs due to friction between the bottle caps and necks. Fragments may also originate from the paints and coatings on walls and furniture due to aging (Dahal & Babel, 2024). In addition, indoor environments are known to contain MPs released from household materials such as textiles, carpets, and curtains, which shed fibers through abrasion and surface wear (Dahal & Babel, 2024). Accordingly, laboratory curtains may represent a potential source of fibrous MPs in the present study. Overall, these findings highlight the potential for MPs contamination arising from routine laboratory practices and infrastructure.

Regarding size distribution, most particles (40%) were observed in the 53-300 μm range. A nearly equal proportion (38%) was found in the 300-500 μm range, while particles ≥ 500 μm accounted for 22 % of the total MPs. The relatively comparable contribution of small and larger size fractions may be influenced by the passive sampling technique used in this study. In general, smaller particles remain more abundant in the air due to their higher suspension and lower settling velocity, whereas larger particles are more strongly influenced by gravity and thus have a higher tendency to deposit. Since passive sampling relies on gravitational settling, larger particles are more efficiently deposited, which may increase their representation in the collected samples. The size distribution of MPs for each sampling event is shown on Fig 2.

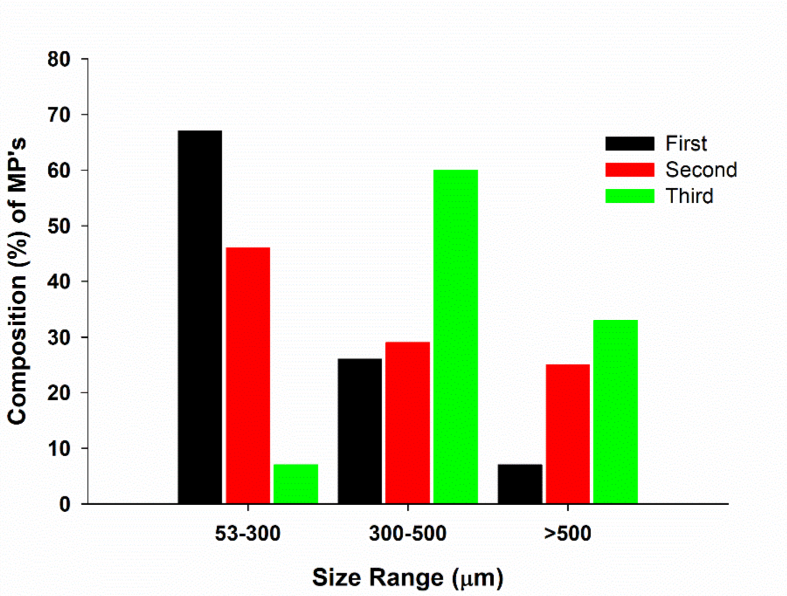

Figure 2 Size distribution of MPs during each sampling event

Size distribution of shapes and polymeric composition of MPs

Fragments also dominated other shapes across all the size ranges. Overall, fragments were more dominant in the lower size range, comprising 68% of the total MPs in both 53-300 and 300-500 μm ranges. However, their abundance decreased to 48% in the > 500 μm range. Pellets accounted for 46% of the total MPs in the > 500 μm range. Specifically, pellets comprised 26% and 18% of total MPs in the 53-300 μm and 300-00 μm size ranges, respectively. The distribution of fragments, fibers, and pellets across different size ranges for each sampling event is shown in Fig. 3.

The size of MPs observed in the laboratory is consistent with findings from previous indoor studies (Table 2), suggesting that laboratory deposition may contribute to the overestimation of MPs in samples if not properly controlled. The dominance of smaller MPs in this study further indicates that distinguishing between true environmental MPs and laboratory-derived contamination can be challenging, as both may occur in similar size ranges.

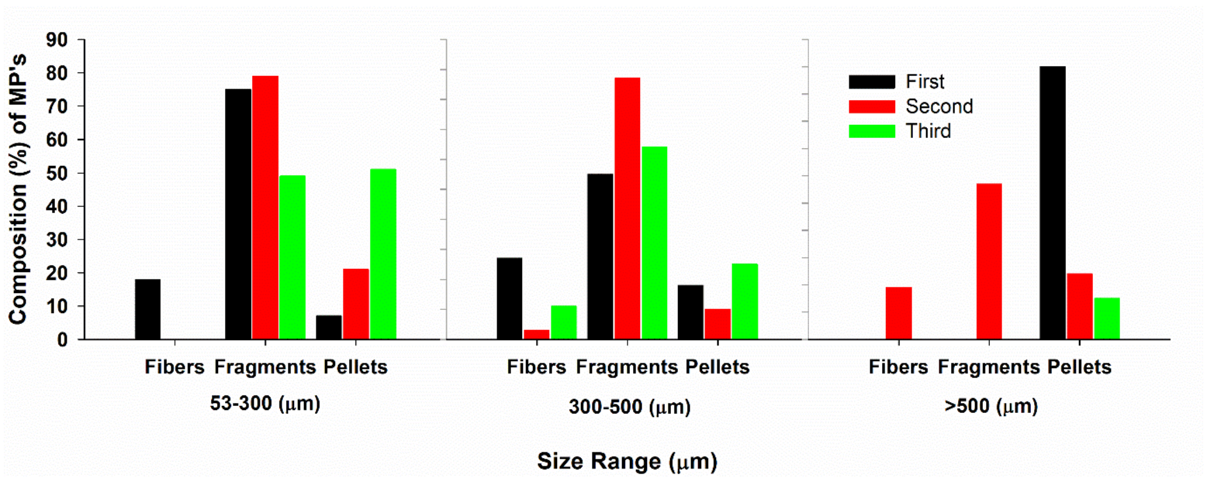

Figure 3 Abundance of MPs shapes across size ranges and sampling events

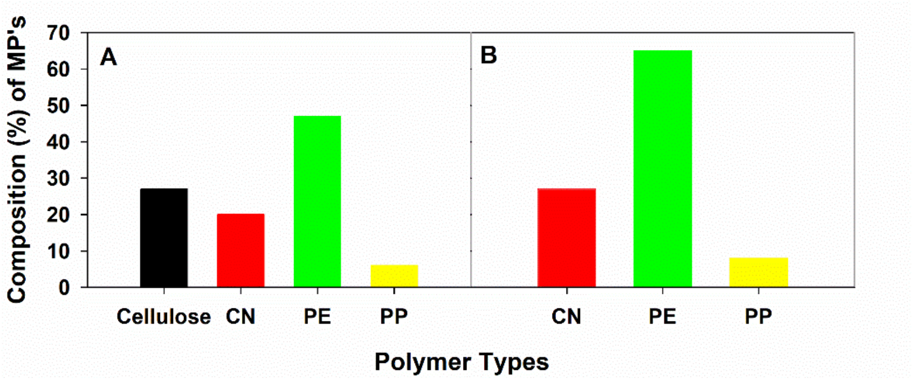

Figure 4 a) Composition of MPs including cellulose, **b)** Composition of MPs excluding cellulose

Micro-FTIR analysis identified four polymers in the laboratory samples. PE (64%) was the most frequently observed polymer, followed by CN (27%) and PP (9%). Although not included in the reported deposition rate, cellulose (a natural polymer) was also abundant in the samples. The abundance of cellulose was higher than that of CN and PP, suggesting it may represent a potential source of natural polymer contamination in the laboratory environment. The polymer composition, including and excluding cellulose, is shown in Fig. 4.

The use of fume hoods and high-efficiency particulate air (HEPA) filtration systems should be encouraged to reduce airborne MPs. In addition, routine monitoring of indoor air quality in laboratory environments is recommended.

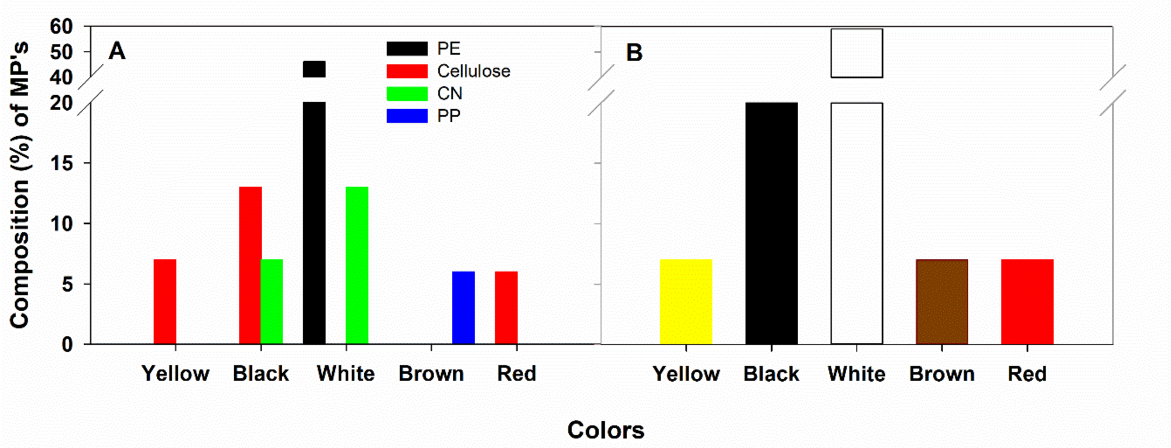

Figure 5 a) Color composition by polymer types**, b)** overall color composition of MPs

Most MPs were white, followed by black. Other detected colors included yellow, brown, and red. Regarding polymer-specific coloration, PE was observed only in white, while PP appeared in brown. Cellulose and CN exhibited multiple colors; cellulose was identified in yellow, black, and red forms, whereas CN was observed in white and black forms. The overall color distribution and polymer-specific composition are shown in Fig. 5.

Similarly, the color and polymeric composition of MPs reported in previous indoor studies are summarized in Table 2.

As observed in this study, PE and PP are the dominant polymers in many indoor environments, as reported by several researchers (Abad-López et al., 2024; Amato-Lourenço et al., 2022; Dahal & Babel, 2024; Dris et al., 2017; Soltani et al., 2021; Yao et al., 2022). In their study, Dris et al. (2017) and Zhang et al. (2020a) identified textiles and household items like carpets, curtains, and clothing as major sources of PP and PET fibers. Soltani et al. (2021) highlighted that carpets are a significant source of MPs, particularly PE, PES, PS, and polyacrylic, with homes containing carpets having twice as many polymers as those without. Similarly, Cui et al. (2022) linked MPs to the abrasion of clothing and upholstery made from polymers such as PA, polyurethane (PU), PE, and PET. Although household items such as carpets and clothing have been reported as potential sources of PE and PP, the absence of such materials in the laboratory and dominance of fragmented MPs suggest that PE and PP were more likely derived from plastic bottles and caps. PE is commonly used in plastic bottles, bags, and food containers (Hee et al., 2023), and is also present in some laboratory items and handling materials due to its excellent chemical resistance. Moreover, PE is also used in AC filter linings, which have been identified as a source of PE MPs in indoor air (Chen et al., 2022). Similarly, PP is a widely used thermoplastic resin found in packaging bags, bottle caps, and plastic films (Jia et al., 2022). It is frequently used in the laboratory for items like centrifuge tubes, pipette tips, trays, beakers, and cylinders. Although fibers were present in a relatively small proportion compared to fragments, a possible source of fibrous PP may be laboratory curtains. Overall, the presence of MPs in the laboratory is likely associated with routine laboratory activities, including handling of reagent bottles and caps, as well as laboratory materials such as curtains.

Cellulose is a natural fiber derived from materials such as cotton and wood (Liu et al., 2019a) and is commonly used in the production of paper and textiles (Lavanya et al., 2011). Therefore, the cellulose fibers found in the laboratory may have originated from the cotton laboratory coats worn by researchers. CN is a flammable and versatile polymer that is used in various applications, including explosives, rocket propellants, plastics, membranes, biosensors, quick-drying lacquers, medical treatments for minor cuts, and in the wood and paint industries (Adekunle, 2010; Gismatulina & Budaeva, 2024). Therefore, the CN detected in the laboratory may be associated with lacquers and paints used on laboratory walls and furniture.

Table 2 Abundance and characteristics of MPs observed in different indoor environments.

Country	**Limit (μm)**	**Deposition Rate (p/m^2/d) and Major Shape**	**Major Size and Color**	**Major Polymers**	**References**
Thailand	53	286 ± 104, Fibers, fragments*	(40% = 53-300 μm) White*, black, yellow, brown, red	PP, PE*, CN, Cellulose	This study
Bangkok, Thailand	53	154 ± 62, Fibers, fragments*	(72% = < 500 μm)	PP*, PE, PET, PA, CN	(Dahal & Babel, 2024)
Paris, France	50	1.6E^3-1.1E^4, Fibers*		PP*, PA, copolymers of PP and PE	(Dris et al., 2017)
United Kingdom	5	1,414 ± 1,022, Fibers*, films fragments, foam, spheres	(52% = <50 μm)	PET*, PE, PA, PP, PAN, PMMA, acrylates, copolymer blends	(Jenner et al., 2021)
Australia	50	3,095, Fibers*, films fragments	(19% = 200-400 μm) Black*, green, blue, red, grey, brown, and transparent.	PE*, PES, PET, PA, PVC	(Soltani et al., 2021)
China	50	2.1E^3-2.9E^4 (Dormitory), 620-4,500 (office), 500-600 (corridor), Fibers*, fragments	(80% = 50-2,000 μm) Transparent*, blue*, red, black, yellow, green, purple	PET*, PP, PES, PS, PA, rayon*, acrylic cellophane	(Zhang et al., 2020b)
New Jersey, USA	1	1.29E^4-2.5E^4, Fibers, films, fragments*	Blue*, red, yellow, brown, grey*, black	PE*, PS, PVC*, PET, PP	(Yao et al., 2022)
Sao Paulo, Brazil	50	309.40 ± 214.71, Fibers, fragments*	(Most = 100-200 μm)	PE*, PP, PET, PES, PVC, copolymers	(Amato-Lourenço et al., 2022)
Yangzhou, Jiangsu, China	20	9.3772E^4 -3.11040E^5 Fibers, fragments*	(39.16-43.58% = 20-30 μm)	PA*, PU, PE, PET, PU, PMMA, PVC, PTFE	(Cui et al., 2022)
Wenzhou, China	5.9	(7.6 ± 3.9) E^5, Fibers, fragments*	(85% = < 100 μm)	PES/PET*, PE, PA, PE, PS, PP	(Fang et al., 2022)
Colombia	10	1.9E^3 - 1.1E^4 Apartment, 1,200-6,100 work areas, Fibers*, fragments	Fibers (1,000-5,000 μm), fragments (< 50 μm), Transparent*, blue*	PES*, PA, PP, PE*, PET, PU, PVC	(Abad-López et al., 2024)

Note: * = Dominant type, PAN = Polyacrylonitrile, PMMA = Polymethyl Methacrylate, PS = Polystyrene, PTFE = Polytetrafluoroethylene, PU = Polyurethane, PVC = Polyvinyl Chloride

Exposure risk

The MPs deposition rate in the laboratory (286 ± 104 p/m^2/d) is approximately two times higher than that reported for the household indoor environment (154 ± 62 p/m^2/d) using the same methodology for MPs identification (Dahal & Babel, 2024). This suggests that laboratory environments may present higher potential exposure to MPs compared to typical indoor settings such as offices or homes. Given that laboratory personnel spend extended periods in such environments, there is a potential for increased inhalation exposure.

Previous studies have reported that inhaled MPs can reach the lower respiratory tract, where they may induce inflammatory responses or act as carriers for associated contaminants. For example, fibers longer than 250 μm with widths of approximately 50 μm were detected in human lung tissue (Pauly et al., 1998), indicating that MPs can penetrate deep into the respiratory system (Gasperi et al., 2018). In addition, indoor exposure assessments have estimated daily inhalation of approximately 704 ± 254 airborne MPs (Geng et al., 2023). Moreover, MPs may also act as carriers of persistent organic pollutants (POPs), metals, pharmaceuticals, and microorganisms, potentially contributing to respiratory health risks (Emenike et al., 2023; Tumwesigye et al., 2023). Deposition of MPs onto laboratory surfaces, gloves, and equipment may also lead to incidental dermal contact; however, the extent of dermal absorption remains uncertain and requires further investigation. Nonetheless, prolonged exposure to MPs and associated additives may raise potential occupational health concerns, including irritation and sensitization (Emenike et al., 2023).

This study indicates that MPs detected in the laboratory likely originate from routine laboratory activities, including handling reagent bottles and caps, as well as from materials used in laboratory infrastructure such as walls, furniture, and curtains. This highlights the need for strengthened occupational health and safety (OHS) measures, including improved air filtration, reduced reliance on plastic-based laboratory consumables, and regular surface cleaning to minimize potential exposure to MPs. Furthermore, laboratory safety guidelines should consider MPs as emerging contaminants of concern. It is recommended to adopt non-plastic alternatives for laboratory ware wherever feasible.

Conclusions

MPs in the laboratory were mainly found as fragments, with PE, CN, and PP as the major polymers. The identified polymers suggest that MPs in the laboratory likely originate from routine laboratory activities, including handling of reagent bottles and caps, as well as from laboratory infrastructure materials such as walls, furniture, AC filter linings, and curtains. Therefore, strict quality control measures are required to minimize background contamination during sample handling and processing in MPs studies. In addition, the persistent presence of MPs in laboratory environments may expose laboratory personnel through inhalation and dermal pathways. This highlights the importance of considering occupational health risks and incorporating MPs into laboratory safety guidelines.

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Author contributions: YD: Conceptualization, acquisition and analysis of data, methodology, writing-original draft; SB: Conceptualization, supervision, writing-review & editing.

Conflict of interests: The authors declare no conflict of interest.

Data availability statement: The data can be provided by the corresponding author upon reasonable request.

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