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Review

Plastic-on-Plastic: Underestimated Contamination Risks in Micro- and Nanoplastic Research

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Abstract: Micro- and nanoplastics (MNPs) are detected in nearly every environmental and biological sample, yet most experiments evaluating their presence and effects take place in laboratories already contaminated with MNPs. Pipette tips, plastic tubes, culture plates, filters, water systems and indoor air all shed MNPs, and, as a result, the lab itself becomes an unaccounted exposure source. At low environmentally relevant doses, this hidden background can reach or even exceed the intended experimental concentrations. This can bias exposure estimates and experimental readouts. This review synthesizes evidence on laboratory plastic contamination in MNP research. Here, we outline the major sources, airborne fibres, purified water and storage containers, plastic consumables and media preparation. We then use these data to construct scenarios of cumulative contamination for typical biological and environmental workflows, highlighting conditions under which laboratory-derived particles can approach or exceed nominal experimental doses. Finally, we analyse how this background inflates and underestimates experimental doses, generates unintended mixed-polymer exposures, and interferes with particle identification and dose–response analysis.

Keywords: nanoplastics; microplastics; Laboratory contamination; plastics consumables; airborne microplastics; background contamination; quality control; exposure assessment

1. Introduction

Microplastics (MPs, <5 mm) and nanoplastics (NPs, <1 μm) have transitioned into a major focus in environmental and toxicological research [1]. They have now been detected in the majority of samples analysed, for example surface waters, deep sea sediments, soils, air, food, and human tissues, and are increasingly studied for their potential to contribute to ecosystem disruption and adverse health effects in humans [2–4]. This rapid expansion of the field has been driven by a legitimate sense of urgency, where the scale of plastic production continues to rise, and the fragments generated through degradation or abrasion are long-lasting and dispersive [5,6].

At the same time, current experimental and laboratory set ups are deeply dependent on plastics. Microtubes, pipette tips, cell culture plates, filtration units, tubing, and plastic bottles are embedded into almost every step of most environmental and biological experimental workflows. Micro and nanoplastics (MNPs) research are, as a result, frequently carried out in laboratories where the contaminant of interest is also structurally present in the infrastructure and reagents used to study it. Reviews on MNPs occurrence, analytics, and effects increasingly acknowledge contamination issues, but routine laboratory practice still often assumes that the laboratory, samples and reagents themselves are free of MNPs [7,8].

This means that MNPs research is often conducted in an environment where the target contaminant is both the analyte of interest and an ever-present substance. While this would already be problematic for an advanced field of study, for a field that is new with many unknowns, which operates in micro to nano detection limits and requires a focus on environmentally relevant low concentrations, such contamination is not only a minor nuisance but a potential source of systematic bias. It threatens to inflate reported concentrations, distort apparent dose–response relationships, and erode confidence in the evidence base used for environmental management and health risk assessment [9,10]. Despite a growing awareness of "blanks and bias", there is still no comprehensive, quantitative framework that links contamination from different laboratory sources into an integrated view of the total plastic load introduced during a typical workflow [11].

In this review, we aim to demonstrate that hidden plastics in the laboratory setting represent an underestimated source of error in MNPs research. Robust conclusions about environmental occurrence, biological effects, and human health risks require consideration of laboratory plastic contamination and its quantitative and cumulative contribution to total MNP burden [12]. Additionally, we highlight a critical gap, which is the lack of integrated, workflow-spanning quantification of plastic contamination across entire experiments, which prevents the field from moving toward contamination-corrected doses and comparable, high-quality evidence, and propose a contamination-corrected framework for MNPs exposure studies [13,14].

1.1. Scope of Microplastics and Nanoplastics in Experimental Research

MPs are conventionally defined as plastic particles with at least one dimension below 5 mm, while NPs are generally considered smaller than 1 μm [15,16]. The size difference between particles can lead to major shifts in their behaviour, since it underlies sharp changes in transport, partitioning, adsorption of co-contaminants, biological uptake pathways, and the analytical approaches required to detect them [17]. A central challenge is that NPs occupy a conceptual and methodological space that partly overlaps with larger MPs. This overlap has prompted calls to treat NPs as a distinct class rather than as a simple extension of the MP size range [18,19].

Currently, research on MNPs is focused on the detection, characterization, and quantification of particles in environmental and biological samples, such as surface and drinking waters, wastewater, sediments, soils, atmospheric fallout, and biota [20,21]. These studies vary with extreme heterogeneity not only in particle size and shape (fragments, fibres, films, beads) but also in polymer type and degree of weathering [22–24]. Each analytical workflow typically combines density separation, filtration, or sieving with enzymatic or chemical digestion and instrumental techniques including micro-FTIR, Raman microscopy, Pyrolysis Gas Chromatography Mass Spectrometry (Pyr-GC/MS), or nanoparticle tracking analysis (NTA), which have specific biases and limits of detection [25].

On the other hand, experimental outcome and effects studies include controlled exposures in in vitro systems, organoid or organ-on-chip models, and aquatic and terrestrial organisms [26]. Such studies probe how MNPs are transported in the environment, interact with natural organic matter and co-contaminants and elicit adverse endpoints on the cellular and organism level such as oxidative stress, inflammation, genotoxicity, metabolic disruption and altered behaviour [3,27]. As the field grows, there is also more interest in relating particle properties to specific modes of action and addressing human-relevant exposures by inhalation, ingestion and possibly translocation over barriers [28,29].

In parallel, there is also more interest by experimentalists in sources, environmental fate and transformation, including how consumer products, textiles and larger plastic debris produce MNPs by mechanical wear, UV weathering or thermal degradation, and how such particles further fragment or agglomerate [30]. Recent reviews highlighted that harmonized terminology, inter-laboratory comparability and method validation are required as a matter of urgency if experimental data are to support sound risk assessment and regulatory decision-making [31]. All these efforts share one critical dependency: they are based on laboratory workflows where plastics are simultaneously object of study and integral part of the experimental infrastructure [32]. The same methodological sophistication that enables trace-level detection and fine-grained effect characterization also heightens sensitivity to background contamination, making the integrity of laboratory conditions a central determinant of data quality in MNPs research [33,34].

1.2. Why Laboratory Plastic Contamination Matters for Environmental and Biological Studies

MNPs are often present as contaminants at low concentrations, and the signal-to-noise ratio is intrinsically unfavourable, for this reason even modest laboratory contamination can have overestimated consequences [35]. Multiple studies have now shown that microplastics and fibres derived from laboratory air, dust, water, reagents, and consumables can be readily introduced into samples during collection, processing, and analysis [12,36]. If unrecognized or poorly controlled, this contamination can inflate MNP count, alter apparent polymer distribution, and generate false positives, particularly for matrices in which genuine environmental loads are low or highly variable [37].

For environmental monitoring and exposure assessment, this means that reported concentrations may partly reflect the laboratory environment rather than the sampled ecosystem. Background fibres from airborne fallout or from clothing can be misclassified as environmental particles, while MNPs released from laboratory tubing, filtration units, or storage containers can be indistinguishable from those present in the original sample. In such scenarios, the contamination behaves not as random noise that averages out across replicates, but as a systematic upward bias that can distort spatial and temporal trends, compromise source apportionment, and misinform models of environmental transport and fate [38,39].

The stakes are equally high for biological and toxicological studies. When MNPs are detected in tissues, fluids, or cell culture systems, there is a strong temptation to interpret them as evidence of exposure and internalization. However, if the same polymer types and size classes are also being shed from culture plates, centrifuge tubes, pipette tips, or even from plastic caps used during sample processing, attribution becomes uncertain [40–42]. In vivo or in vitro experiments that do not rigorously characterize and correct for laboratory-derived particles run the risk of misestimating actual doses, conflating environmentally-derived MNPs with artifacts, and drawing erroneous conclusions about dose–response relationships, thresholds, or mechanisms of toxicity [9,14].

Moreover, the lack of standardized contamination-control protocols across laboratories exacerbates these problems. Recent reviews and guidance documents on sampling and quality assurance and control in MPs analytics highlight substantial heterogeneity in the use of procedural blanks, clean-air facilities, filtered reagents, and reporting practices [43,44]. This heterogeneity undermines reproducibility and the comparability of results across studies, complicating meta-analyses and weight-of-evidence approaches for hazard and risk assessment. In short, laboratory plastic contamination is not a local technical inconvenience; it is a field-level threat to the validity, interpretability, and relevance of MNPs research [45,46].

1.3. Lack of Integrated Quantification across Laboratory Workflows

Despite a growing recognition that laboratory contamination is pervasive and significant, the evidence base is inconsistent. Individual studies have characterized MNPs released from specific sources, for example, airborne deposition in laboratories, contamination of commonly used reagents, or particle shedding from plastic consumables, and have proposed mitigation strategies such as standardized blanks [47,48]. These efforts have been instrumental in demonstrating that laboratories are not plastic-free spaces and in motivating the adoption of basic quality assurance practices.

However, most existing work addresses contamination at isolated stages of the workflow or focuses on single components in relative isolation. One study may quantify fibres settling in laboratories, another might document MNPs present in commercially available salts, while a third quantifies particles released from laboratory tubing or filters [49–51]. What is largely missing is an integrated, quantitative perspective that tracks how these contributions accumulate from sample intake through pre-treatment, extraction, concentration, and final analysis, and how the resulting contamination compares to the nominal doses or environmental concentrations being measured [52].

Interlaboratory comparisons and methodological reviews have repeatedly called for harmonized quality criteria and for more systematic reporting of blanks and contamination controls, especially in the context of risk assessment [53]. Yet even where blanks are measured, their interpretation is often limited to subtracting average background counts, without embedding them in a broader framework that distinguishes between contamination arising from air, water, reagents, and plasticware, or that explicitly propagates contamination-related uncertainty into final exposure or effect estimates [33,39]. As a result, the field lacks an actual control group, common standards and set of metrics for comparing contamination burdens across laboratories, methods, and study types [54,55].

This gap in research has several consequences. Firstly, without workflow-spanning quantification, it is difficult to know under which laboratory contaminations are negligible relative to true sample loads, and when they represent a dominant fraction of the measured signal [56,57]. Secondly, the absence of a formalized concept of contamination-corrected dose means that toxicological studies may inadvertently misclassify exposures, for example by attributing effects to nominal doses that in fact differ substantially from the particles actually reaching cells or tissues [58]. Thirdly, without integrated frameworks, it is challenging to prioritize mitigation, where laboratories lack quantitative guidance on which sources or procedural steps contribute most to contamination and therefore deserve the greatest focus in redesign or control [59].

Addressing this gap requires moving beyond ad hoc descriptions of contamination toward workflow-level cumulations and dose accounting, grounded in empirical measurements but structured in a way that can be generalized and adapted across systems [60,61]. Providing such a framework is necessary to understand where, how, and to what extent hidden plastics in the lab shape the evidence base on MNPs.

2. Sources and Quantification of Plastic Contamination in Laboratories

Laboratories contain multiple sources of MNPs contamination (Figure 1). These sources differ between environmental MNPs analysis and biological effect studies, but the underlying mechanisms are similar. Workflows involving open handling, filtration, pipetting, or repeated transfers are especially vulnerable to unintended MNP contamination. Laboratory-based studies of MNPs are inherently vulnerable to contamination originating from the lab environment itself. Several recent investigations have systematically assessed how air, water, reagents, and consumables contribute to MNP contamination, often at levels that risk confounding experimental results [12,62]. The main analysed sources of contamination in common laboratory workflows are outlined in Table 1.

Table 1. Quantitative summary of laboratory micro- and nanoplastic contamination.

Article (Author, Year)	Study Context	Polymer Types Identified	Particle Amount/Size	Contamination Source or Material	Quantitative Findings & Implications
Bhat et al., 2024 [63]	Airborne MPs in university indoor spaces	PA66, PTFE, PP, HDPE, PE + 20 others	13.88–18.51 MPs/m^3; 2.5–336.89 µm size	Airborne indoor dust, fibres from textiles, ventilation	Exposure to airborne MPs with diverse morphologies; irregular fragments dominant; human activity and airflow increase MP resuspension
Zhai et al., 2023 [64]	MP deposition in offices, labs, dorms	Not specified	Dormitories: 14,088.05 pcs/m^3; Labs: 7512.55 pcs/m^3; Mean size 66.15 µm (lab MPs smaller)	Indoor air turbulence, human activity	Higher contamination in dormitories; smaller MPs in labs potentially more hazardous; airflow promotes MP redistribution indoors
Rindelaub & Miskelly, 2025 [62]	Airborne MPs in chemical labs (mass-based)	PC, PVC, PE	PM2.5: 0.51 µg/m^3; PM10: 1.14 µg/m^3	Resuspended dust, ventilation systems in labs	Detected seven polymers and plasticizers in inhalable fractions; important for assessing workplace inhalation exposure
Kutralam-Muniasamy, 2023 [65]	MPs in lab water, salts, reagents	PE, PP, polyester, nylon, acrylic, paint polymers, cellophane, viscose	Water: 30.21 ± 30.40 particles/L; salts: 24 ± 19 particles/10g; chemicals: 187 ± 45/L	Contaminated lab consumables (water, reagents)	Fibres dominant (81%); majority MPs <500 µm; wide polymer diversity in routine lab liquids and salts
Mason et al., 2018 [66]	Bottled water MP contamination globally	PP dominant (54%), industrial lubricants	10.4 particles/L (>100 µm size); 325 particles/L including 6.5–100 µm fraction	Bottled water, packaging materials	93% samples contaminated; fragment-shaped MPs dominate; smaller MPs prevalent but less chemically characterized
Jones et al., 2024 [12]	MP contamination in lab water & consumables	Not fully specified	Milli-Q water: 21.7 particles/mL; tap water higher at 151.7 particles/mL	Lab water sources (Milli-Q to tap), plastic & glassware	Water contamination unavoidable even in ultrapure sources; glassware shows higher contamination than plasticware due to handling/storage
Hermabessiere et al., 2020 [67]	Plastic additive leaching from reagent bottles	Irgafos® 168 (antioxidant in PE, PP)	Up to 5493 ng/mg (plastics), 3000 ng/mg (powders)	Reagents stored in PE and PP bottles	Additives leach into reagents, biasing ecotoxicology and MNP leaching studies
Gwinnett & Miller, 2021 [68]	Contamination from plastic vs. glassware	Not specified	Plasticware: ~6.9 particles/mL; glassware: ~1356.9 particles/mL	Plastic & glass lab consumables	Plastic consumables have unexpectedly lower contamination, probably due to sterile packaging and manufacturing standards
Fang et al., 2024 [69]	Plastic syringe contamination (PDMS lubricant)	PDMS (solid fragments and liquid droplets)	Thousands of MPs & millions of NPs introduced per 1 mL syringe	Syringe stopper lubricant (PDMS)	Injection introduces significant MPs and NPs, affecting contamination in experiments using plastic syringes

ABS, acrylonitrile butadiene styrene; PA66, polyamide 66; PTFE, polytetrafluoroethylene; PP, polypropylene; HDPE, high density polyethylene; PDMS, polydimethylsiloxane; PE, polyethylene; PC, polycarbonate; PVC, polyvinyl chloride; PM2.5, particulate matter ≤ 2.5 µm; PM10, particulate matter ≤ 10 µm; PDMS, polydimethylsiloxane; MPs, microplastics; NPs, nanoplastics; MNPs, micro and nanoplastics.

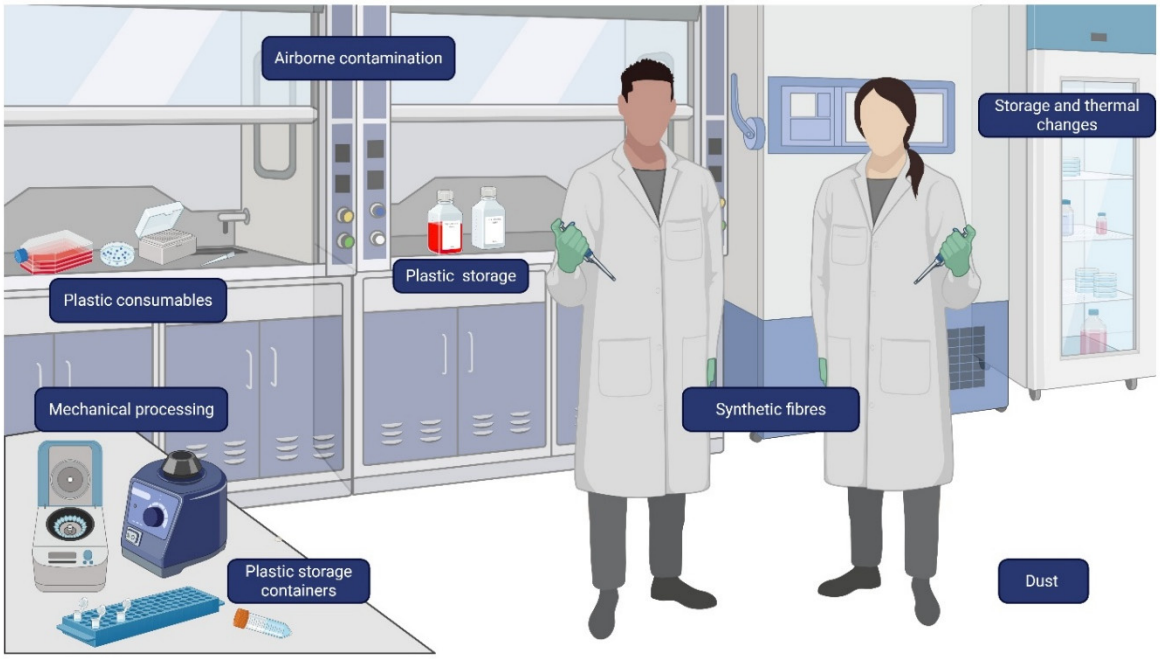

Figure 1. Possible sources of micro- and nanoplastic contamination in laboratory environments.

2.1. Airborne Plastic Contamination

Airborne MNPs and synthetic particles have gained recognition as a pervasive source of contamination in indoor environments [63,64]. To investigate the amount of airborne plastic contamination, a recent study examined the occurrence, morphology, and chemical composition of MPs in multiple indoor environments [63]. Optical microscopy revealed a wide range of particle morphologies, including fibres, fragments, pellets, foams, films, and line-shaped particles. Fragments were the dominant MP type, though their accurate quantification was limited by high particle density. Many MPs exhibited irregular, rough margins indicative of secondary fragmentation [63].

They further carried out polymer identification, by micro-Raman spectroscopy and SEM-ED, which revealed 25 different plastic types, with polyamide 66 (PA66), polytetrafluoroethylene (PTFE), polypropylene (PP), high-density polyethylene (HDPE), and polyethylene (PE) among the most abundant [63]. University occupants were estimated to be exposed to airborne MPs in the size range of 2.5 to 336.89 μm, at concentrations of 13.88–18.51 MPs/m^3 and corresponding daily inhalation rates of 180–240 MPs. Particle size distributions and abundances varied across indoor environments. Elemental analysis consistently detected carbon and oxygen as major components, along with fluorine, sodium, chlorine, aluminum, silicon, and other trace elements [63].

In another study that monitored MP deposition across offices, laboratories, dining halls, and dormitories, the highest average MP abundance was detected in dormitories (14,088.05 pcs/m^3), followed by offices (13,097.13 pcs/m^3), laboratories (7512.55 pcs/m^3), and dining halls (4308.26 pcs/m^3) [64]. Across all sites, the mean particle size was 66.15 μm, although MPs in laboratory environments were reported to be smaller and potentially more hazardous [64]. Airflow experiments using air conditioning systems demonstrated that turbulence enhances the resuspension of MPs. In addition, human activity levels emerged as a major driver of temporal variability in indoor MP concentrations, while airflow-induced turbulence promoted their redistribution within indoor spaces [64].

A targeted study in chemical laboratories addressed the lack of mass-based data on airborne MPs and associated chemical additives, which is critical for evaluating inhalation exposure in indoor workplaces such as chemical laboratories [62]. Using a Pyr-GC/MS approach, the authors quantified both polymeric particles and low-molecular-weight plastic additives in the inhalable air fraction from two indoor locations. Seven polymer types were detected in PM2.5 and PM10, with mean plastic mass concentrations of 0.51 μg/m^{-3} and 1.14 μg/m^{-3}, respectively [62]. Polycarbonate (PC), PVC, and PE were the most abundant polymers in inhalable air. In parallel, phthalate-based plasticizers were quantified at an average concentration of 334 ng m^{-3} [62].

These data indicate that routine lab air, even under clean conditions, carries a measurable load of MPs and associated additives. Sources likely include resuspended dust, fibres shed from clothing or textiles, abrasion of plastic surfaces, and operation of ventilation/air-handling systems that stir up settled particles [63,70,71].

Thus, work done in open benches, fume hoods, or laminar flow cabinets without strict contamination control can lead to deposition of MPs onto samples, culture media, reagents, or open containers, possibly exposing experiments with unintended plastic particles [63,70–72].

2.2. Water Systems and Reagents

Water and reagents used in MNP analysis and environmental/biological experiments are themselves non-negligible sources of contamination [65]. To investigate this, researchers examined the presence and characteristics of MPs in commonly used laboratory consumables, including distilled, deionized, and Milli-Q water, salts (NaCl and $CaCl_2$), chemical reagents (H_2O_2, KOH, and NaOH), and ethanol obtained from both academic laboratories and commercial sources [65]. MP concentrations averaged 30.21 ± 30.40 particles L^{-1} in water samples, 24.00 ± 19.00 particles per 10 g in salts, 187.00 ± 45.00 particles L^{-1} in chemical solutions, and 27.63 ± 9.53 particles L^{-1} in ethanol, with statistically significant differences observed among the different substances. Fibrous particles were predominant, representing 81% of all detected MPs, followed by fragments (16%) and films (3%) [65]. Most particles were smaller than 500 μm (95 percent), with observed sizes spanning from 26 μm up to 2.30 mm. Polymer analysis identified a diverse mixture of materials, including PE, PP, polyester, nylon, acrylic, paint-derived polymers, cellophane, and viscose [65].

Although particles smaller than 100 μm could not be chemically characterized by spectroscopy, their positive Nile Red staining strongly suggested a plastic origin, as Nile Red stains plastics effectively as it's a lipophilic dye that binds to the hydrophobic surfaces of polymers [66,73]. When these smaller particles (6.5–100 μm) were included, the overall mean concentration increased to 325 MNP particles per litre. Across all samples, MP abundances ranged from non-detectable to more than 10,000 particles per litre, with 95% of the detected particles falling within the 6.5–100 μm size fraction [66].

These findings show that even "ultrapure" water (e.g., Milli-Q, reverse osmosis) often used in labs is not absolutely free of MNP particles. In another study, all evaluated water sources contained detectable plastic and non-plastic particles [12]. Milli-Q water exhibited the lowest level of plastic contamination at 21.7 particles mL^{-1} (95% CI: 4.7–64.4), followed by reverse osmosis water at 29.9 particles mL^{-1} (95% CI: 9.6–77.3), whereas tap water showed substantially higher concentrations at 151.7 particles mL^{-1} (95% CI: 62.2–387.0) [12]. Tap water contained significantly greater numbers of both plastic and non-plastic particles compared with Milli-Q and reverse osmosis water ($p < 0.05$). Although Milli-Q represented the cleanest source, these findings highlight that background water contamination must be carefully considered when establishing procedural and experimental blanks [12].

While it stands to reason that the storage of reagents and chemicals in plastic bottles and containers may contribute to NMP contamination in their contents, very little has actually been measured between these different reagents, for example cell culture media or PBS [68]. The process of media or buffer preparation itself may introduce MNPs, via plastic media bottles (commonly HDPE or PP), through shedding during handling (pouring, decanting, mixing), or via filtration through plastic units (e.g., syringe filters, plastic-tip filtration devices). In the laboratory, every step involving reagent and sample preparation, storage, and transfer may contribute to cumulative MNP contamination [12,74].

One study examining laboratory contamination risks revealed that the plastic additive Irgafos® 168, an antioxidant used in PE and PP packaging, leaches ubiquitously from high-density PE bottles and PP caps into common reagents such as citric acid, sodium phosphate dibasic, sodium cholate hydrate, calcium chloride, Tris-HCl, pepsin, trypsin, and pancreatin [67]. Both the reduced form (tris(2,4-di-tert-butylphenyl)phosphite) and its oxidized form (tris(2,4-di-tert-butylphenyl)phosphate) were quantified via pyrolysis-GC/MS, with at least one form detected in all tested caps, bottles, and reagent powders, showing significantly higher concentrations in plastics (up to 5493 ng/mg in pepsin bottles for reduced form) than in powders (up to 3000 ng/mg in citric acid). This leaching was evidenced by matching concentration profiles and ratios between bottles and powders across reagents, particularly elevated in acidic ones, compounded by detection in lab deionized water containers, underscoring how such MNPs and additives bias ecotoxicological experiments on MNP leaching [67].

2.3. Plastic Consumables

Plastic laboratory consumables may commonly be viewed as a major source of secondary MNP contamination during sampling and analysis, leading to frequent recommendations to replace them with glass alternatives [8,68]. In contrast to this assumption, a landmark study demonstrated a markedly lower level of contamination when plasticware was used compared with glassware ($p < 0.0001$) [12]. Experiments performed with glass instruments exhibited the highest contamination levels, with an estimated concentration of 1356.9 plastic particles mL^{-1} (95% CI: 975.3–1861.1). By comparison, experiments conducted using plastic consumables showed only 6.9 particles mL^{-1} (95% CI: −0.7–19.2), a value similar to the Milli-Q procedural control measured immediately after collection (6.7% mL^{-1}; 95% CI: −0.8–19.0) [12].

This discrepancy is likely linked to differences in manufacturing and handling conditions. Plastic laboratory consumables are produced, transported, and stored under medical and analytical quality standards, including ISO 13485 and ISO 9001, ensuring high levels of cleanliness and sterility [12]. All plastic items used in the experiments were sterile, RNase-free, endotoxin-free, and individually sealed in airtight packaging. In contrast, glassware was supplied in cardboard packaging, often with minimal plastic covering and without individual wrapping, which may facilitate particle deposition during transport and storage [12].

Notably, the few particles detected in plasticware experiments were smaller than their standard 3.2 μm detection threshold, making them unlikely to be identified by μ-FTIR. This emphasizes the need to carefully consider the interaction between potential contamination sources and the size limits of the analytical techniques used for MNP detection [12].

Plastic syringes, which typically have a stopper located at the end of the plunger, typically composed of polydimethylsiloxane (PDMS) and associated additives, with low-molecular-weight liquid PDMS applied as a lubricant to reduce friction and ensure sealing between the stopper and barrel [69]. During injection, both solid PDMS fragments derived from mechanical abrasion and liquid PDMS droplets originating from the lubricant can be released, representing solid and liquid MNPs. Raman hyperspectral imaging was used to visualize PDMS MNPs, with SEM providing high-resolution morphological validation. Weak Raman signals from NPs were enhanced using image deconvolution, and multiple syringes and randomly selected areas were analysed to ensure statistical reliability and reduce quantification bias[69]. Based on these measurements, the use of a single 1 mL plastic syringe was estimated to introduce thousands of PDMS MPs and millions of PDMS NPs in both solid and liquid form [69].

Altogether, while plastic consumables, such as pipette tips, tubes, syringes, and culture plates shed particles during use, especially under thermal stress, friction, or freeze-thaw cycles, glassware as an alternative does not always significantly reduce unintended plastic contamination in laboratory settings, both capable of undermining the integrity of experiments especially in MNP research or other assays sensitive to particulate contaminants [8,12,68]

2.4. Lab Processes That Create MNPs

Laboratory-generated MNPs may arise from routine handling of plastic consumables, chemical treatments, or thermal stress applied during experimental procedures. These MNPs may confound experimental outcomes, particularly in studies examining cellular or molecular responses to nanomaterials [68,75]. Mechanical, chemical, and thermal processes generate MNPs with distinct characteristics. Overall, mechanical fragmentation produces irregular, polydisperse particles with high surface area, chemical degradation generates smaller particles with altered surface chemistry, and thermal processes yield oxidized, rough-surfaced particles [76–78]. These mechanistic differences are important when considering the potential biological activity of MNPs, as particle size, morphology, and surface chemistry influence interactions with cells, proteins, and various substances, which is an essential context for minimizing unintended contamination and interpreting results accurately [79].

2.4.1. Mechanical Processes and MNP Generation

Mechanical fragmentation is a primary route through which laboratory plastics can release MNPs [75,80–82]. Activities such as stirring, vortexing, pipetting, cutting, opening and closing containers, or nanoscratching surfaces, subject polymers to friction, shear, and abrasion forces [78,80–82]. Brittle polymers, including PS, are particularly susceptible to fragmentation, with repeated stress producing irregular, polydisperse nanoscale particles [75]. Laboratory simulations have demonstrated that repeated mechanical stress accelerates MNP formation, producing particle abundance and size distributions comparable to those observed in environmental weathering studies [78,81]. Similarly, studies on synthetic textiles indicate that mechanical abrasion during washing releases nanoplastics, highlighting the general susceptibility of polymers to mechanical degradation [82]. These findings suggest that routine lab manipulations, even when considered low impact, may represent a continuous source of MNP shedding.

2.4.2. Chemical Degradation and Surface Modification

Chemical processes in the laboratory can also generate MNPs and alter their surface properties. Exposure of plastics to oxidative reagents, hydrolytic enzymes, or hydrogen peroxide induces polymer chain scission, producing nanoscale fragments enriched with functional groups such as carbonyls [29,76]. Enzymatic and oxidative treatments mimic environmental weathering, accelerating MNP formation while modifying physicochemical properties, including particle size, surface chemistry, and colloidal stability [29,78]. Such chemical modifications can increase MNP reactivity, alter protein adsorption, and potentially influence cellular

uptake and toxicity [83,84]. Laboratory protocols employing sterilizing agents, oxidative cleaning, or enzymatic reactions should therefore be recognized as potential contributors to MNP contamination.

2.4.3. Thermal Processes and Oxidative Aging

Thermal stress, including heating in the presence of oxidants or UV exposure, promotes polymer chain scission and surface oxidation, possibly resulting in the production of MNPs [76–78]. Experimental systems combining heat, UV radiation, drastic temperature changes, and mechanical shear, such as the accelerated plastic aging in suspension (APAS) method, generate MNPs that closely mimic environmental particles in terms of morphology and surface chemistry [77]. Thermal degradation not only reduces particle size but also modifies chemical functionality, which can influence interactions with biological systems and may complicate interpretation of experimental results [5,76,85].

2.5. Cumulative Contamination during Typical Lab Workflows: A Rough Estimate

Rather than acting as isolated sources, airborne deposition, reagents, consumables, handling steps, and analytical workflows may form a continuous contamination pathway in which plastic particles accumulate progressively across the full experimental pipeline (Figure 2) [86,87]. Airborne MNPs and laboratory dust establish the primary background burden before any experimental manipulation occurs, driven by ventilation systems, resuspension from surfaces, and synthetic textile fibres from clothing [12,63,88]. This initial background is then carried forward into experimental solutions through laboratory water and chemical reagents, which consistently contain measurable MNP loads even after high-purity treatment [12,56]. Because these solutions are used at every stage of biological experimentation, reagent-derived plastics act as persistent vectors rather than isolated contamination events.

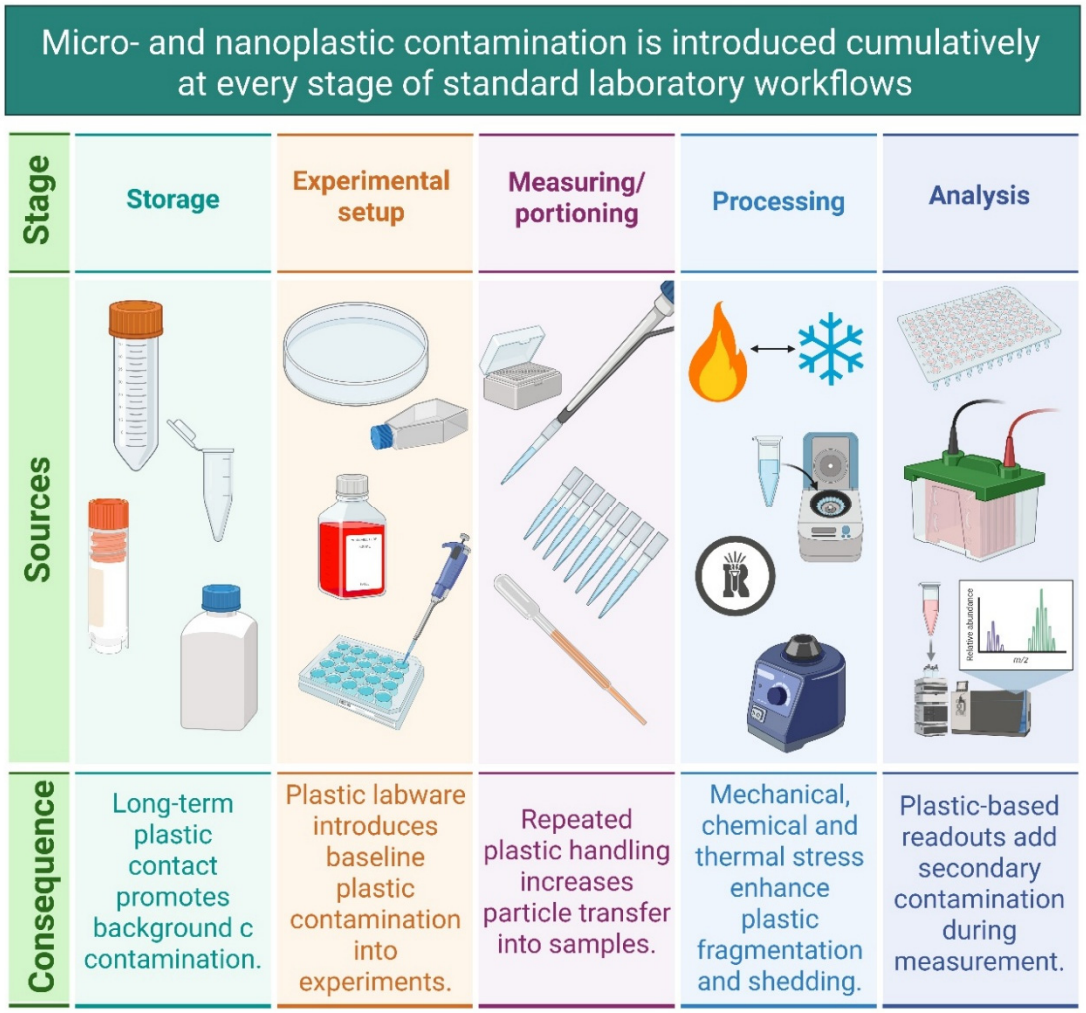

Figure 2. Representation of possible sources contributing to cumulative micro- and nanoplastic contamination across experimental pipelines.

During sample preparation, consumables and bench materials amplify this background rather than replacing it. Both plasticware and glassware contribute comparably to MNP introduction through abrasion, surface shedding, and repeated handling [12,63]. Critically, contamination introduced at this stage is not reset but compounded as samples transition into biological processing. Each downstream step, including pipetting, centrifugation, digestion, resuspension, and filtration, may simultaneously introduces new particles and selectively removes others in a size-dependent manner, with NPs being preferentially lost and underestimated [86]. This creates a structural asymmetry in which background contamination accumulates while true exposure becomes progressively harder to resolve.

Finally, analytical detection does not eliminate this accumulation but instead imposes an additional layer of uncertainty. Commonly applied techniques such as flow cytometry and filtration-based quantification operate near their sensitivity limits for NPs and are affected by both procedural contamination and incomplete recovery [48,89,90]. As a result, the final reported particle concentrations reflect not a single contamination event but the integrated sum of inputs originating from air, water, consumables, handling, and detection itself. This cumulative pathway explains why background burdens in untreated controls frequently approach, and in some cases rival, nominal low-dose experimental exposures [39,88].

The cumulative nature of this contamination pathway has direct implications for experimental bias in MNP research. When background burdens from air, water, reagents, consumables, and workflows are not rigorously quantified and corrected, measured particle counts effectively conflate true exposure with laboratory-derived artefacts, leading to systematic overestimation of environmental or biological loads, especially at low doses [12,65,88,91,92]. High and variable blank values can dominate signal in low-concentration samples, inflating apparent contamination and distorting limits of detection and quantification [39,88,92]. In toxicity testing, additional artefacts, such as particle adsorption to vessel walls, sedimentation, flotation, and interactions with test reagents, can generate both false positives and false negatives if not controlled with appropriate spike and zero-hour controls [93].

At the same time, many exposure studies rely on supraphysiological concentrations and simplified particle types, further decoupling nominal doses from environmentally realistic internal burdens and biasing inferences about risk [94–96]. Omnipresent procedural contamination of key reagents and solvents means that even well-intentioned quality-control measures (e.g., switching to glassware, using recommended digestion chemistries) can silently reintroduce plastics [12,65,92,97]. Collectively, this MNP accumulation during experimental work flows creates a structural tendency for reported MNP concentrations and effects to be skewed away from real-world exposure scenarios unless rigorous, matrix-specific QA/QC, blank design, and data correction strategies are integrated from the protocol design stage onward [8,37,39,83,91,98].

3. Impact on Micro- and Nanoplastics Research: Why This Matters

3.1. Inflation of Nominal Dose and Distortion of True Exposure

Possibly, one of the most fundamental consequences of laboratory-derived MNP contamination is the systematic inflation of the nominal dose relative to the true intended experimental dose [1,37].

Given that background particles are already present in laboratory water, reagents, consumables, and air prior to any intentional spiking, the total plastic burden experienced by cells, organisms, or environmental samples may exceed the concentration assumed by the investigator [12,65]. This becomes especially problematic in low-dose and environmentally relevant exposure studies, where intended concentrations often fall within the same order of magnitude as background contamination. Under these conditions, the experimental system is effectively pre-exposed before treatment begins. As a result, dose–response relationships can be artificially shifted, producing exaggerated toxicity signals or false-positive effects. This issue is amplified for NPs, where recovery efficiencies frequently drop below 50% for particles near 100 nm, while detection itself approaches instrumental sensitivity limits [81,86,89]. Together, these factors create a scenario in which true exposure is simultaneously underestimated analytically and overestimated biologically, leading to profound uncertainty in quantitative toxicological interpretation.

3.2. Mixed-Polymer Exposures and Loss of Experimental Specificity

A second major consequence of laboratory contamination is the unintended conversion of single-polymer experiments into mixed-polymer exposure systems. Background MNPs originating from pipette tips, tubes, gloves, air, and reagents routinely include PE, PP, polyester, nylon, and acrylic polymers [12,65]. When experiments are designed to test the biological effects of a specific material, such as PS nanospheres, this polymeric background introduces uncontrolled heterogeneity in size, shape, surface chemistry, and degradation state [7,14,99]. Because different polymers exhibit distinct cellular uptake kinetics, oxidative reactivity, protein corona formation, and toxicological profiles, even small contributions from unintended plastics can confound mechanistic attribution of

observed effects [40,53,100]. This loss of experimental specificity directly undermines comparability across studies. Two laboratories using identical nominal particles and doses may in practice expose cells or organisms to fundamentally different plastic mixtures, which likely contributes to the high variability and poor reproducibility that characterize much of the current MNPs toxicology.

3.3. Analytical Misidentification and Measurement Bias

Laboratory-derived contamination also directly interferes with particle detection and polymer identification. A clear example is provided by glove-derived leachates, which contain long-chain hydrocarbons and fatty acid derivatives that can be misclassified as PE by μ-Raman, μ-FTIR, and pyrolysis-GC/MS [101]. This demonstrates that contamination introduced purely through handling may generate chemical signatures indistinguishable from true plastic polymers [101,102]. At the same time, commonly applied spectroscopic techniques show declining accuracy for particles below approximately 20 to 30 μm, with an increased risk of misclassifying natural organic matter, mineral fragments, or laboratory dust as plastics [48]. These analytical limitations are particularly relevant for NPs, where reliable particle-specific identification and polymer discrimination remain major technical challenges [35].

3.4. Misleading Biological and Environmental Conclusions

The combined effects of dose inflation, mixed-polymer exposure, and analytical misidentification can ultimately lead to misleading scientific conclusions. Toxic effects attributed to a specific plastic type may in fact arise from background polymers unintentionally introduced before exposure [15,37,54]. Apparent dose–response relationships may reflect cumulative background plus experimental particles rather than the intended treatment alone. Inter-laboratory variation in airflow, reagent batches, consumable lots, and handling practices further amplifies experimental noise, contributing to the emerging concerns about reproducibility of MNP toxicological studies [74]. These same mechanisms also affect exposure assessment studies, including drinking water, food, and biological tissue surveys [12,27,81]. When blank controls are absent or inadequately reported, laboratory-derived plastics can be misinterpreted as environmentally derived contamination, leading to overestimation of environmental plastic burdens and inaccurate polymer source attribution.

4. Limitations, Knowledge Gaps, and Variability across Studies

4.1. Methodological Heterogeneity and Poor Cross-Study Comparability

Despite growing awareness of contamination, quantitative estimates remain highly variable across studies. Large standard deviations in reported reagent and blank contamination are common, reflecting differences in sampling protocols, filtration thresholds, staining procedures, and detection platforms [73,74]. These methodological differences make direct cross-study comparisons inherently unreliable. In particular, most studies still prioritize MPs above 1 to 10 μm, while the nano-size fraction remains largely understudied, due to instrumental detection limits and low recovery efficiency [10,103]. This is a critical gap for nanotoxicology, where the smallest particles are expected to show the highest biological reactivity.

4.2. Absence of Standardized Per-Item Release Metrics

At present, there is no widely accepted quantitative framework describing how many particles are released per defined laboratory action, such as a single pipetting cycle, thermal changes, vortexing step, or centrifugation step. As a result, consumable-derived contamination is often reported qualitatively rather than quantitatively [12]. This severely limits the ability to build predictive contamination studies for experimental workflows and prevents meaningful standardization across laboratories.

4.3. End-to-End Workflow Quantification Remains Rare

Most contamination studies examine isolated sources, such as water, air, or consumables, rather than tracking total particle accumulation across an entire experimental pipeline. To date, only a small number of comprehensive assessments have evaluated contamination from multiple sources simultaneously within a single workflow [12,59,63,65]. Even these do not yet provide universally transferable contamination amounts applicable to diverse experimental systems such as cell culture, in vivo toxicology, or environmental analysis.

4.4. Implications for Data Interpretation

Together, these limitations mean that background contamination must be treated not as a minor experimental disturbance, but as a structurally embedded component of MNP research. Without rigorous blank correction, delivered-dose verification, and polymer-specific validation, both biological and environmental studies remain vulnerable to inflated effect sizes, false mechanistic attribution, and poor reproducibility [33,39,88]. This is particularly critical for environmentally relevant exposure studies, where background contamination may represent a dominant fraction of the total particle burden and fundamentally compromise causal inference.

5. Future Research Directions

Future progress in the field depends on shifting from nominal-dose toward real-dose quantification, supported by systematic recovery experiments and blank subtraction [12,74]. Greater emphasis must be placed on analytical methods capable of resolving the nanometer size fraction with polymer-specific identification, as this currently represents a major blind spot for both exposure science and nanotoxicology. Standardization of airborne deposition metrics, development of per-item release coefficients for consumables, and end-to-end contamination mapping across full experimental workflows are needed to enable cross-laboratory comparability. This will aid the field to overcome inconsistent results, uncertainty in exposure quantification, and limited mechanistic resolution.

Quantitative and Standardized QA/QC Frameworks in MNP Research

A key priority for the field is the transition from largely descriptive assessments of contamination toward structured, quantitative, and reproducible QA/QC frameworks that can be realistically implemented across laboratories. Recent reviews and harmonization studies highlight that inconsistencies in sampling strategies, analytical resolution, polymer identification techniques, reporting units, and metadata availability remain major barriers to inter-study comparability [104–106]. Rather than pursuing rigid universal models that may not be adaptable across matrices and methodologies, future work should focus on flexible harmonization strategies based on minimum reporting standards, transparent QA/QC documentation, and FAIR-compatible data structures [105,107].

A practical direction forward is the systematic integration of tiered blank controls into all experimental workflows. Recent literature emphasizes that contamination can originate from multiple sources—including airborne deposition, reagents, laboratory materials, and handling procedures, and that incomplete blank control remains one of the most critical sources of bias [39,108,109]. Accordingly, future studies should routinely include field blanks, procedural blanks, reagent blanks, and instrument blanks, processed in parallel with experimental samples under identical conditions [108]. Replicate blanks should also be incorporated to estimate variability, as blank-associated uncertainty is an integral component of quantitative MNP analysis [39].

Contamination should be treated as a quantitative variable rather than a qualitative limitation. Recent advances in analytical frameworks emphasize the need to report contamination in terms of particle number concentrations, size distributions, and polymer composition, alongside method-specific detection limits [106,110]. This is particularly critical for NP-scale analyses, where procedural contamination can significantly influence measured signals and compromise interpretability if not explicitly accounted for [108].

Future methodological development should also prioritize the implementation of contamination-corrected dose calculations. In vitro and in vivo exposure studies should normalize corrected particle counts to biologically relevant units, such as cell number, tissue mass, or exposure surface area, enabling more meaningful comparisons across experiments [106]. In parallel, uncertainty associated with blank variability should be propagated into reported values. This is especially important given that contamination levels may approach or exceed true experimental signals, particularly in low-dose studies [39,108].

Equally important is the translation of contamination awareness into actionable mitigation strategies. Recent experimental and review studies consistently identify airborne deposition, laboratory consumables, reagents, and synthetic clothing as major contamination sources [109,111,112]. Airborne contamination can be significantly reduced through laminar flow systems and minimized exposure times, while filtration of reagents and the use of low-shedding or non-plastic materials can further limit background contamination [109,111]. Instrument-related contamination and memory effects should also be addressed through routine cleaning and the inclusion of instrument blanks [110]. Despite growing awareness, these mitigation strategies are still inconsistently implemented across studies [104].

In this context, a structured QA/QC workflow should become a routine component of experimental design. Such workflows should include pre-experimental baseline contamination assessment, parallel processing of multiple blank types, post-acquisition correction of measured values, and transparent reporting of both raw and

corrected data [105,107]. Recent guidance documents emphasize that reproducibility depends not only on analytical methods but also on standardized reporting and accessible metadata [105].

Future studies should also define explicit decision criteria for data reliability. Emerging approaches suggest that datasets heavily influenced by blank contamination or characterized by high variability should be flagged or excluded from downstream analyses [113]. In this context, artificial intelligence tools have recently been proposed as scalable approaches for QA/QC evaluation, demonstrating the ability to replicate expert-based quality assessments and support data screening in large datasets [113].

Finally, future efforts should aim to consolidate these practices into community-endorsed guidelines that balance rigor with feasibility. Interlaboratory comparison studies demonstrate that even standardized protocols still exhibit variability due to differences in sample preparation and analytical workflows [114]. Therefore, rather than enforcing uniform methodologies, future guidelines should define essential QA/QC elements, including contamination quantification, correction procedures, and transparent reporting [8,105,109]. Collectively, these advances will improve contamination-corrected dose estimation, enhance data comparability, and strengthen the interpretability of MNP toxicity and exposure studies [104,106].

6. Conclusions

Laboratory contamination by MNPs is not incidental but systematic, biasing all experimental studies investigating MNPs. Quantitative studies have shown that air, dust, reagents, and consumables carry detectable MNP levels, and that these sources together can contribute background levels comparable to or exceeding the intended experimental exposures [12,65]. As a result, the interpretation of MNP experiments requires explicit contamination control and transparent reporting.

By incorporating blanks and contamination-corrected dose calculations, researchers in MNP research can ensure that observed effects reflect the intended exposure rather than those derived from laboratory contamination, as well as considering the extent to which MNPs may be affecting research in other fields. Adoption of these practices will improve reproducibility across studies and support more reliable risk assessment, environmental monitoring, and mechanistic toxicology research.

Author Contributions

Conceptualization, A.B., C.M. and E.T.; writing—original draft preparation, C.M.; writing—review and editing, E.T.; visualization, A.B., C.M., E.R. and S.P.; supervision, E.T. All authors have read and agreed to the published version of the manuscript.

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Conflicts of Interest

The authors declare no conflict of interest.

Use of AI and AI-Assisted Technologies

No AI tools were utilized for this paper.

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