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Cross-contamination pathways in the analysis of plastics and related chemical compounds: Good laboratory practices and tips

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ABSTRACT

The analysis of plastics and related chemical compounds, such as plasticizers, flame retardants, and micro- or nanoplastics, often requires working at trace levels, where even minimal contamination can significantly affect results. However, many of these target analytes are also present in common laboratory materials and environments, increasing the risk of cross-contamination. We identified six major cross-contamination pathways frequently found in analytical workflows: (I) laboratory materials, (II) environmental contamination, (III) human handling and manipulation, (IV) solvents and reagents, (V) cleaning and sample preparation, and (VI) instrumental and system-related contamination. For each of these, preventive measures and good laboratory practices are suggested based on both experimental experience and examples in the literature. As a general recommendation, procedural blanks should be included throughout the analytical process, and contamination risks should be anticipated as early as the experimental design stage. This work provides a structured reference to support more reliable and reproducible data generation in the analysis of plastic-related contaminants. Researchers are further encouraged to evaluate contamination risks throughout the workflow and to report them transparently in their publications.

1. Introduction

Plastics have revolutionized modern life due to their versatility, low cost, durability, and adaptability to a wide variety of applications from food packaging and medical devices, to electronics, textiles, and automotive components [1]. Their widespread utility stems from the ability to engineer them with tailored properties, often achieved through the incorporation of chemical additives such as plasticizers, stabilizers, flame retardants, and colorants [2]. These compounds are often referred to as “everywhere chemicals” due to their extensive use in modern society and their pervasive presence in the environment, wildlife, and the human body [3]. They enhance flexibility, longevity, color, transparency, and resistance to heat or ultraviolet radiation. However, the very features that make plastics attractive, such as their chemical stability and resistance to degradation, are also what make them persistent environmental pollutants. In 2022, global plastic production reached approximately 400 million tons, with a significant proportion ending up in landfills, oceans, and other ecosystems, where plastics slowly degrade into microplastics and nanoplastics [4]. These small particles, together with the associated additives they release, pose a serious risk to environmental and human health.

Among the most concerning groups of these additives are plasticizers, particularly phthalate esters, adipates, and structurally similar compounds [4]. These substances are not covalently bound to the plastic polymer chains, which allows them to migrate easily into surrounding media, including food, water, and the human body. Plasticizers are widely used to enhance the elasticity, color, and durability of polymers, especially in food contact materials [5]. Their uncontrolled migration has raised concern due to their identification as endocrine-disrupting chemicals capable of interfering with hormonal systems and causing potential developmental, reproductive, and metabolic disorders in humans and wildlife [6]. These compounds have been detected in various matrices such as foods [7,8], water [9], dust [10,11], soil [12], and marine environments [13]. Due to their ubiquity and potential impact, plasticizers and related compounds have become a hot topic in analytical chemistry as mentioned by the European Chemical Agency,

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with numerous studies focusing on their quantification [6], migration behavior [14], and toxicological impact [15].

Nevertheless, the accurate determination of these compounds remains a challenge, especially when working at trace levels in complex matrices such as food samples [16,17]. While significant progress has been made in developing analytical methods, risk assessment, and regulatory frameworks, an important methodological issue remains insufficiently addressed: the risk of cross-contamination during sample handling and analysis. Plastic-related compounds are so pervasive in laboratory environments that they can unintentionally contaminate samples leading to false positives [18].

Although some studies have identified specific sources of contamination, such as laboratory air, consumables, solvents, or instrumentation [19–23], these works mainly focus on detecting or quantifying contamination sources rather than providing practical strategies to prevent or minimize them. As a result, clear guidance on how to mitigate these contamination pathways during analytical workflows is still limited. The aim of this work is therefore to highlight the main cross-contamination pathways that may occur during the analysis of plastics and related chemical compounds and to compile good laboratory practices and simple, actionable recommendations to minimize them. Through the classification of contamination sources, the suggestion of preventive actions, and the presentation of selected examples from the literature, we intend to provide a practical framework to recognize, control, and reduce contamination risks in the analysis of plastics and related compounds.

2. Cross-contamination pathways in the analysis of plastics and related compounds

The accurate determination of plastic-related compounds in analytical chemistry is susceptible to cross-contamination due to the ubiquitous presence of plastics and their additives in laboratory environments [16,24]. Given the trace-level concentrations often involved and the structural similarity between analytes and potential contaminants, even minor interferences can significantly affect analytical outcomes [25]. Cross-contamination may arise from multiple sources throughout the workflow, including laboratory materials, environmental exposure, human handling, solvents, cleaning procedures, and instrumentation. Understanding these pathways is important to ensure the reliability of results, particularly when analyzing phthalates, microplastics, flame retardants, or other additives prone to migration [4]. In this section, we categorize and describe the most relevant cross-contamination pathways (see Table 1), providing examples of how these sources can compromise analytical integrity when working with plastics and related compounds in the laboratory.

2.1. Laboratory materials

A significant number of cross-contamination incidents in the analysis of plastics and related compounds originate from the materials used during laboratory procedures [24]. Items such as plastic packaging, glassware, vial caps, filters, pipette tips, gloves, and tubing are routinely employed in sample handling and preparation, yet they may themselves be sources of the same analytes under investigation [26]. Plastic packaging and storage devices often contain phthalates or other additives that can leach into samples during storage, especially under conditions of heat, solvent exposure, or prolonged contact. Glassware, though often considered inert, can also contribute to contamination if not properly cleaned, particularly when residues from previous experiments persist on surfaces. Septa and caps made from rubber or low-grade polymers may release compounds such as siloxanes or plasticizers, which can be detected during chromatography-mass spectrometry analysis and misidentified as target analytes. Filters and membranes used for sample clarification may introduce low levels of polymer residues if not certified as plasticizer-free. Besides cellulose acetate filters, which are susceptible

Table 1
Cross-contamination pathways and preventive measures in the analysis of plastics and related compounds.

Section	Cross-contamination pathways	Suggested solutions
I. Laboratory materials	Plastic packaging	Use certified plasticizer-free packaging; prefer glass containers
	Glassware (flasks, beakers, tubes)	Clean with a specific protocol, and dry with foil cover
	Vial caps, septa, and sample containers	Use septa and caps made of Teflon or other inert materials
	Filters and membranes	Use pre-cleaned, low-binding, plasticizer-free filters
	Pipette tips and syringes	Use certified contaminant-free tips; pre-rinse before use
	Gloves	Use powder-free gloves and change frequently to avoid leaching
	Plastic tubing, connectors, and valves in instruments	Replace with inert materials when possible
	Storage devices	Use glass or certified contaminant-free plastic containers; avoid long-term storage in standard plastics
II. Environmental contamination	Laboratory air	Work under laminar flow hoods or clean benches; limit exposure time
	Ambient dust	Wipe surfaces regularly with lint-free cloths; keep sample containers closed
	Fibers from clothing	Wear lab coats made of cotton or use disposable cleanroom garments
	Cabinets, fume hoods, and benches	Clean all surfaces before use; use foil or paper liners if needed
	Airflow systems	Install HEPA filters and maintain airflow systems routinely
III. Human handling and manipulation	Direct contact from operator (skin, breath, hair)	Wear specific gloves, masks, and hairnets; avoid leaning over open containers
	Cross-use of tools or instruments without proper cleaning	Use dedicated tools for each sample or clean thoroughly between uses
	Carry-over between samples due to poor handling	Implement strict sample handling protocols; use procedural blanks
	Manual transfers without intermediate cleaning or protection	Use pre-cleaned or single-use materials; avoid touching sample contact surfaces
IV. Solvents and reagents	Contaminated organic solvents	Use high-purity (e.g., LC-grade) solvents; test with blanks before use
	Reagents stored in plastic containers that may leach additives	Store sensitive reagents in glass containers with proper sealing
	Ultrapure water not verified for microplastic contamination	Regularly test water with blank runs; consider filtration prior to use
V. Cleaning and sample preparation processes	Improper rinsing of glassware or instruments	Follow validated rinsing procedures using ultrapure water and acid washes
	Inadequate covering materials (e.g., plastic wrap instead of aluminum foil)	Use aluminum foil or glass lids to cover containers

(continued on next page)

Table 1 (*continued*)

Section	Cross-contamination pathways	Suggested solutions
	Contamination during drying (e.g., dusty ovens or shared equipment)	Use dedicated clean ovens; cover items with aluminum foil during drying
VI. Instrumental and system-related contamination	Contaminated injector ports in GC or LC systems	Regularly clean or replace liners and injector components
	Carry-over from internal system parts (liners, tubing, valves)	Flush with solvent blanks; use dedicated paths when possible
	Contaminated chromatographic columns	Use guard columns; monitor and replace columns regularly
	Plastic internal components in instruments (e.g., pump tubing, filter housings)	Replace with inert materials like stainless steel when possible

to degradation, glass fiber filters have been widely used for microplastic sampling because of their durability and affordability [27]. However, glass fiber filters may also become contaminated with fibers or plastic residues, since they are often manufactured in uncontrolled environments and packaged using plastic materials.

Similarly, pipette tips and syringes made from polypropylene or polyethylene can carry residual additives or absorb analytes, resulting in inconsistent recovery values. For example, Marega et al. [20] demonstrated that the absorption of phthalates such as, diisobutyl phthalate, dibutyl phthalate and di(2-ethylhexyl) phthalate, from the laboratory air on the outer wall of the syringe needle, is an important contribution to the blank problems.

Even gloves, particularly those made from vinyl or nitrile containing plasticizers, can transfer contaminants during manual handling [5]. Additionally, plastic tubing and connectors in instrumentation may slowly leach additives into the mobile phase or sample stream, while long-term storage in conventional plastic boxes or Eppendorf tubes may lead to background signals [6]. All these factors highlight the importance of scrutinizing the materials in contact with samples, especially when working at trace levels.

2.2. Environmental conditions

Environmental contamination poses a silent but substantial risk in the analytical determination of plastics and their related compounds, especially in laboratories that lack stringent air quality controls. Airborne particles present in the laboratory atmosphere can settle on open containers or exposed surfaces, leading to unintended introduction of microplastics or additive residues into the samples [28]. For example, microfibers are recognized as the most frequent particles contributing to cross-contamination. They are commonly present in the ambient air of sampling sites and laboratory environments, where they can adhere to equipment and materials, and potentially interfere with the analysis of microscopic plastics [29].

Ambient dust is often composed of fragmented synthetic materials originating from lab coats, packaging materials, or degraded equipment, which can mimic target analytes such as polyester or polyethylene fibers in microplastic analyses [24]. This issue was investigated by Fankhauser-Noti and co-workers who found that di(2-ethylhexyl) phthalate and dibutyl phthalate present in the air were easily absorbed onto various surfaces, including instruments, solvents, glassware, and septa [21]. Their study identified laboratory air as the primary source of contamination, which ultimately led to the abandonment of plastic materials containing phthalate esters in their procedures. Clothing fibers, particularly from garments made of synthetic textiles like acrylic or polyester, are another frequent contributor to background contamination [30]. These fibers may be released during normal laboratory activity and become electrostatically attracted to sample surfaces or filters. In addition, particle deposition can occur during the analysis of microplastic filters, since airborne fibers and particles can settle on the filter surface while it is exposed to laboratory air during microscopic or spectroscopic examination. Moreover, fume hoods may act as reservoirs for previously deposited contaminants that become resuspended during sample processing. They are ineffective at preventing airborne contamination because their primary function is to extract air, which draws unfiltered laboratory air in through the front opening [27]. Even the laboratory's ventilation system, when lacking proper filtration, may distribute fine airborne debris across the workspace. Similarly, open windows and doors, as well as high levels of human circulation in the laboratory, can further contribute to the introduction and redistribution of airborne particles.

However, the magnitude and composition of environmental contamination vary between laboratories, depending on infrastructure, ventilation systems, and implemented cleanroom practices. Facilities equipped with HEPA-filtered air and strict clothing policies generally exhibit lower airborne particle counts than conventional analytical laboratories [27]. In contrast, shared spaces or laboratories without controlled air exchange are more prone to fiber and additive deposition. Therefore, these environmental differences should be considered when comparing analytical data between laboratories or establishing harmonized quality-control protocols for microplastic analysis.

2.3. Human handling and manipulation

Human handling is an underestimated source of contamination in analytical workflows. Direct contact from the operator, via skin, breath, or hair, can unintentionally introduce interfering substances into the sample, especially in the absence of gloves, masks, or hairnets. In analyses involving volatile plasticizers or microplastics, these interactions become problematic due to the ease with which airborne contaminants can settle on uncovered containers or sample supports. The cross-use of tools or instruments without adequate cleaning further compounds this issue. Pipettes, spatulas, or weighing boats used across different sample sets without intermediate rinsing or solvent blanks can facilitate carry-over of analytes [25]. Manual transfers performed without protective barriers or cleaning steps may also result in cross-contamination between containers or matrices. These subtle yet impactful errors often go unnoticed unless strict procedural blanks and handling protocols are implemented.

2.4. Solvents and reagents

Solvents and reagents, though assumed to be chemically pure, can themselves become unexpected sources of cross-contamination [4]. Typical organic solvents such as methanol, acetonitrile, or dichloromethane may contain trace levels of plasticizers or other additives, either from the manufacturing process or due to storage in plastic containers [25]. Even high-purity organic solvents can contain phthalate ester at approximately 1-ng/mL level and may become further compromised if stored for extended periods in polyethylene or polypropylene bottles, as leaching of additives can occur gradually over time [19]. This may represent a significant source of contamination when preparing calibration curves, extraction solutions, or mobile phases, where background signals can considerably affect analyte concentrations [24]. Similarly, other reagents stored in plastic containers may contribute to contamination if the containers are not certified contaminant-free.

Water (ultrapure, distilled, or tap) is one of the most frequently used reagents in analytical laboratories, employed for cleaning materials, preparing solutions, and even directly adding to samples. Consequently, its contamination can substantially contribute to microplastic overestimation in analytical results. Researchers typically rinse laboratory materials several times with water of varying purity grades, from distilled and Milli-Q® to filtered or HPLC-grade water. Prata et al. [27] demonstrated that tap water contained the highest microplastic concentration (10 particles L^{-1}), followed by distilled water (4 particles L^{-1}) and ultrapure water (1 particle L^{-1}). Contamination can originate directly from the water source, from contact with laboratory equipment and containers, or through the deposition of airborne fibers. Differences among studies may also arise from variations in methodology, particularly in the filter pore size used for microplastic detection. For instance, ultrapure water showed twenty times more microplastics when filtered through a 0.7 μm membrane compared to a 1.2 μm one [27]. These observations highlight that water contamination becomes increasingly relevant as particle size decreases, given the higher abundance of smaller microplastics. Moreover, in some laboratories, unexpected plastic-related signals have been traced back to unfiltered ultrapure water systems or water stored in standard plastic carboys. In fact, the analysis of trace contamination of phthalate esters in ultrapure water using a modified solid-phase extraction procedure and automated thermal desorption–gas chromatography/mass spectrometry was investigated [28]. The study revealed that di-*n*-butyl phthalate and di (2-ethylhexyl)phthalate were the most common contaminants detected, highlighting the potential for ultrapure water to act as a hidden source of plastic-related interference.

Although it is difficult to quantify the relative contribution of each potential pathway to total analytical contamination, recent evidence offers a general perspective on this issue [23]. Laboratory water and handling-related sources, including consumables such as pipette tips and tubes, appear to be the main contributors, while glassware and laboratory dust can also introduce significant contamination. Even materials commonly used to prevent airborne exposure, such as aluminum foil, may add measurable amounts of plastic particles. In contrast, short-term experiments suggest that airborne particles are generally less influential than water and consumables in contributing to overall contamination.

2.5. *Cleaning and sample preparation processes*

Even with appropriate materials and reagents, ineffective cleaning and preparation procedures can affect the analytical results. Residual plasticizers, monomers, or polymer fragments may remain adsorbed on glassware or instrument components if not thoroughly rinsed between uses. Standard laboratory detergents are often insufficient for eliminating trace contaminants, and when rinsing is skipped or performed inadequately, carry-over into subsequent samples becomes inevitable. Another subtle but relevant source of contamination is the use of inappropriate covering materials during drying or storage. Wrapping clean glassware with plastic film, for instance, can introduce compounds such as phthalates through passive transfer, especially if the material is exposed to heat or prolonged contact. A more suitable approach is to use aluminum foil, which acts as a physical barrier without contributing organic residues [27]. However, Jones et al. [23] reported that fresh aluminum foil may represent a potential source of contamination. The foil itself may already contain particulate residues originating from manufacturing processes, transfer during packaging (e.g., contact with plastic packaging materials), or environmental deposition prior to use, as its large surface area can easily accumulate airborne particles. Therefore, although aluminum foil has traditionally been used to reduce airborne contamination, researchers should consider that it may also introduce particles into samples.

Drying ovens and shared equipment can also contribute to background contamination, particularly if dust particles or volatile residues accumulate inside. When glassware is placed in such conditions without protective wrapping, contaminants can adsorb onto surfaces and later be released during sample processing. These often-invisible missteps may lead to elevated blanks or unexpected chromatographic signals.

2.6. *Instrumental and system-related contamination*

Contamination coming from the analytical instruments themselves is more common than often assumed. Gas chromatography (GC) and liquid chromatography (LC) coupled with mass spectrometry are the most widely employed techniques for the analysis of plastic substances [1, 25]. Compared to GC, LC has been less reported in the analysis of plasticizers, possibly because of multiple contamination sources, such as filters, pipes and mobile phases [16,31]. In this context, some researchers have proposed strategies to minimize phthalate contamination in LC analysis. For instance, Vavrouš et al. [32] addressed phthalate contamination by incorporating a contamination trap into the system, using a 50 mm reversed-phase chromatographic column. Moreover, in GC or LC systems, injector ports can accumulate residues over time, especially when working with low-volatility compounds or high-concentration standards. Even with proper purging, small amounts of plasticizers or analytes can stick to liners, column head, needles, or valve components, slowly releasing across multiple runs [33,34]. Internal parts like tubing, seals, or solvent filters can also retain traces of compounds due to incomplete flushing or adsorption, making it difficult to pinpoint the source when unexpected peaks appear [35]. Chromatographic columns, too, are a known weak spot, certain analytes may interact strongly with the stationary phase and linger between injections, even after routine conditioning. And when plastic components are part of the instrument's flow path, such as degasser membranes or pump tubing, they can leach additives into the mobile phase over time, especially when exposed to solvents or elevated temperatures.

Importantly, the extent of instrumental contamination may be different between laboratories and instrument models. Different system designs, materials of construction, and maintenance histories can result in distinct contamination profiles even when identical analytical protocols are followed. Instruments that employ PEEK or PTFE-based flow paths may exhibit different leaching behaviors compared to all-metal systems. Additionally, the frequency and thoroughness of maintenance influence the persistence of residual contaminants. Regular preventive maintenance, combined with system blanks and dedicated contamination checks after servicing, is important to ensure reliable and reproducible chromatographic performance.

3. Analytical techniques for the characterization of plastics and microplastics

It is important to highlight that many of the instrumental systems, despite their contamination risks, are also at the core of advanced analytical strategies for characterizing plastics and microplastics. Among the most used methods are Fourier transform infrared (FTIR) spectroscopy and Raman spectroscopy, both of which provide non-destructive, chemically specific analysis. FTIR is especially effective for detecting microplastics larger than 20 μm and can identify the chemical structure of polymers based on their infrared absorption spectra. It offers modes such as transmission, reflectance, and attenuated total reflectance (ATR), each with different sample preparation requirements. While transmission mode requires thin and transparent particles, ATR and reflectance modes allow the analysis of thicker or more opaque particles without extensive preparation [36]. Micro-FTIR imaging further enhances the ability to identify particles both visually and spectroscopically on a single platform, although challenges such as particle adhesion, low spectral quality for small particles, and high equipment cost persist [37,38]. Raman spectroscopy, on the other hand, is better suited for detecting smaller particles, down to a few micrometers, using laser light to generate distinct spectra based on molecular composition. Although it preserves the sample and offers high spatial resolution, Raman analysis can be complicated by fluorescence interference, particle curvature, and the influence of additives or pigments that alter the spectral response [39]. Both FTIR and Raman methods depend heavily on robust spectral libraries to minimize misidentification, especially when microplastics are weathered or mixed with other substances such as surfactants, microbes, or heavy metals [40].

In addition to spectroscopic techniques, mass spectrometry-based methods, such as gas chromatography–mass spectrometry (GC-MS) and liquid chromatography–mass spectrometry (LC-MS), are gaining attention for their ability to identify and quantify microplastics, particularly nanoplastics, through the detection of pyrolysis or hydrolysis byproducts. These methods allow polymer identification even in complex matrices [1]. Thermal analysis techniques such as thermogravimetric analysis (TGA) [41,42] and differential scanning calorimetry (DSC) [43] are also powerful tools, in which microplastics are heated to monitor changes in physical state and thermal behavior. The appearance of endothermic peaks and specific thermogram patterns provides insights into polymer type, composition, and the presence of additives.

Each of these techniques, while offering advantages, also presents limitations such as high cost, analytical complexity, and the need for expert interpretation [44]. Also, the performance and operating conditions of these instruments may influence analytical outcomes [45]. Contaminants on optical components, residues in sampling cells, or poor baseline correction may introduce false peaks or mask polymer-specific bands [46]. For example, non-plastic materials such as organic matter (e.g., lipids or biogenic residues) might generate similar analytical signals, particularly in pyrolysis-based techniques, complicating polymer identification. In Raman spectroscopy, laser-induced heating can modify or degrade thermoplastic materials, while fluorescence from additives or pigments often obscures the target signal [47]. In FTIR, the selection of measurement mode (ATR, transmission, or reflectance) affects penetration depth and detection sensitivity, and improper contact between the crystal and particle surface may produce distorted spectra [48]. A major limitation of chromatography-based methods is the effective removal of interferences, since many compounds produce the same pyrolysis products as plastics, potentially leading to false positives and overestimation [44]. Calibration quality, environmental humidity, and background subtraction are critical parameters to control, as they determine the reproducibility and comparability of spectral data across laboratories. Nevertheless, with the development of hybrid systems and automated data processing, these advanced methods are increasingly used for the accurate and comprehensive analysis of plastics in samples.

4. Impacts of plastic contamination generated during analytical processes

The increasing ubiquity of plastic particles has led to extensive research on their effects on ecosystems, food safety, and human health. However, their influence within the analytical workflow itself remains insufficiently explored. Although several studies have described its environmental and toxicological consequences [49,50], few have discussed how contamination originating during analytical processes can compromise data reliability and interpretation. Contamination generated throughout sampling, sample handling, or instrumental analysis can affect the accuracy of results. The most frequent impacts include:

a) False positives, particularly in analyses of small particles or fibers, where distinguishing genuine signals from laboratory-derived contamination is difficult.
b) Overestimation of microplastic concentrations caused by the introduction of external particles during analytical procedures [27]. However, microplastic losses during sampling and analysis may lead to underestimation, which in some cases can outweigh contamination effects.
c) Distortion of quantitative results, reducing accuracy and precision, and leading to unreliable data interpretation [27].
d) Reduced interlaboratory comparability, since uncontrolled contamination introduces random variability that obscures real differences between samples [51].
e) Carry-over from injector ports, columns, or tubing, which can persist across multiple injections, causing ghost peaks and unstable baselines. In complex matrices, such interference may be mistaken for real analytes, especially when retention times or spectra overlap.
f) Incorrect quantification of plasticizers or microplastics, leading to biased environmental monitoring data, misleading risk assessments, and unreliable food safety evaluations. Overestimation may trigger unnecessary regulatory concern, whereas underestimation can conceal real exposure levels.
g) Invalidation of experiments or sampling campaigns when contamination levels in blanks equal or exceed those in samples [52].
h) Loss of scientific credibility and challenges in standardization, as contamination undermines confidence in analytical outcomes and complicates the establishment of quality benchmarks [27].

i) Waste of analytical resources and time, since contaminated analyses often require repetition or exclusion from datasets.
ii) Inability to establish realistic detection limits, as background contamination elevates the analytical noise and conceals true signals.
iii) Increased indirect environmental footprint, due to additional waste and solvent consumption during control and reanalysis steps.

5. Preventive measures and good laboratory practices

Identifying the sources of contamination is only the first step. In the analysis of plastic substances and related chemical compounds, rigorous contamination control measures must be applied throughout the entire analytical process to obtain reliable results [1]. The following section presents a set of practical tips and good laboratory habits that can help reduce or eliminate such issues [24]. The recommendations outlined are not intended to serve as a strict protocol, but rather as a practical guide which, based on our experience (see Table 1) and previous publications [21,22,27,53,54] (see Table 2), has proven useful for researchers working in this field. A recent study reported that the most widely adopted measures to control cross-contamination in microplastics sampling are the use of non-plastic containers, followed by non-plastic collection utensils and non-plastic samplers [55]. In contrast, more specific preventive measures such as rinsing laboratory materials, performing blank tests, or wearing cotton clothing and gloves are mentioned less frequently. However, during sample processing, the most reported practices include the use of blank tests, cotton lab coats, and material rinsing [55].

Proper management of laboratory glassware is undeniably important (see Fig. 1). Glassware should be thoroughly washed by multiple rinses with ultrapure water, acids (e.g., nitric acid, sulfuric acid) and organic solvents (e.g., acetone, methanol, hexane, dichloromethane, and isooctane) (see Table 3) [16]. These steps help remove residual compounds from previous analyses. Once cleaned, it is recommended to dry it in dedicated ovens and protected with aluminum foil or glass lids rather than plastic film, which may release contaminants upon contact [56]. Keeping cleaned items stored in closed or covered conditions further reduces their exposure to airborne particles during storage or preparation. Placing all clean laboratory consumables in a desiccator containing aluminum oxide is also highly recommended [16]. Aluminum oxide is commonly used as a desiccant and adsorbent because of its high surface area, porosity, and strong affinity for polar compounds including those potentially released from polymeric materials or laboratory consumables. These properties make Al_2O_3 effective in trapping low-molecular-weight contaminants and residual moisture, thus maintaining a clean atmosphere inside desiccators [57]. Similar adsorptive approaches may be achieved using other porous materials such as activated silica gel, zeolites, or metal–organic frameworks [58].

In injection systems such as gas chromatography–mass spectrometry, cleaning the needle by immersing approximately 5 cm in a cleaning

Table 2
Summary of main strategies to avoid cross-contamination in the analysis of plastics and related compounds.

Analytes	Main cross-contamination control strategies	Ref.
Plasticizers	Glassware cleaning procedure; plastic consumables (micropipettes tips) and chemical reagents checked; run procedural blanks; compound blank response subtracted	[4]
Plasticizers	Glassware cleaning procedure; no gloves during samples preparation and analysis; blanks were run between sets of samples	[5]
Plasticizers	Glassware cleaning procedure; laminar flow cabinet use; no gloves during sample weighing; routine inclusion of procedural blanks; pre-check of vial caps, filters, and organic solvents	[6]
Phthalate esters, alkylphenols	Glassware cleaning procedure; blanks were run between sets of samples; cellulose filters	[14]
Non-phthalate plasticizers	Glassware cleaning procedure; use glass material instead of plastic material; wear nitrile gloves by the analyst; avoid the use of personal care products like creams or cosmetics; do not share solvents with colleagues to prevent cross-contamination, as this could lead to potential contamination issues; use Teflon filters instead of nylon filters; run analytical blanks	[18]
Phthalate esters	Glassware cleaning procedure; install a charcoal filter into the gas supply; heat out the injector at high temperature; purify solvent by adding aluminum oxide; add aluminum oxide to the wash solvent on the autosampler; store clean laboratory ware in a desiccator containing aluminum oxide	[21]
Phthalate esters	Replacement of filters and solvent tubing; washing the flow path of the mobile phase from the glass bottles to the column with organic solvents (isopropanol, isohexane, acetonitrile and dichloromethane); shortening the re-equilibration time between runs; install an additional hold-back HPLC column packed with the same material as the analytical column upstream of the injection valve to retain interferents	[22]
Microplastics	Glassware cleaning procedure; solvents tested	[25]
Phthalate esters	Glassware cleaning procedure; run analytical blanks	[33]
Plasticizers	Glassware cleaning procedure; cover materials with aluminum foil; sample preparation was performed in clean surroundings; solvents and reagents were avoided to approach plastics; run analytical blanks	[34]
Plasticizers	Glassware cleaning procedure; solvents checked; run analytical blanks	[56]
Phthalate esters	Glassware cleaning procedure; organic solvents were treated with AlO_3	[60]
Phthalate esters	Glassware cleaning procedure; plastic materials used (gloves or pipette tips) were free of phthalates; solvents checked; run procedural blanks	[67–69]
Phthalate esters	Glassware cleaning procedure; plastic or rubber materials were not used to avoid contact with samples or solvents	[70]
Phthalate esters	Glassware cleaning procedure; solvents checked; run analytical blanks	[71]
Phthalate esters, adipates	Material cleaning procedure; run analytical blanks	[72]
Phthalate esters	Glass or enamel labware were used in order to avoid contamination from plastic labware	[73]

1 INITIAL CLEANING
Wash glassware with a suitable laboratory detergent using non-plastic tools when possible (e.g., brushes or metal supports)

2 ULTRAPURE WATER RINSE
Rinse thoroughly (e.g., three times) with high-purity water (Grade 1 or equivalent)

3 SOLVENT RINSE
Rinse with a high-purity, pre-filtered organic solvent (e.g., methanol or acetone) to eliminate remaining residues

4 DRYING
Dry in a clean, controlled environment (e.g., dedicated oven or laminar flow cabinet) free from polymeric materials

5 IMMEDIATE PROTECTION
Cover immediately using pre-cleaned, inert materials (e.g., aluminum foil or glass lids)

6 CONTROLLED STORAGE
Store in a clean, closed environment (e.g., glass desiccator or sealed cabinet) to prevent airborne contamination

Fig. 1. Plastic-free glassware cleaning and handling workflow for contamination control.

Table 3
Reported cleaning protocols for glassware used in the analysis of plastics and related compounds.

Cleaning procedure (step-by-step)	Ref.
Soaked with ultrapure water, nitric acid, and ultrapure water again, followed by a final wash with a mixture of acetone:methanol (1:1, v/ v), dried at 150 °C (1 h)	[4]
Heated at 400 °C for at least 4 h and was covered with aluminum foil prior to use	[5]
Soaked in ultrapure water, washed with 0.2 M nitric acid, and sonicated (20 min). Rinsed with ultrapure water and sonicated again. Then washed with acetone:methanol (1:1, v/v), dried at 150 °C (≥1 h), and covered with aluminum foil	[6]
Soaked with distilled water and then with acetone before drying in an oven (60 °C)	[14]
Heated at 400 °C for 2 h and rinse with hexane	[21]
Soaked and washed in acetone, dried at 140 °C for at least 4 h	[25]
Immersed in methanol overnight, then dried at 140 °C for 4 h and rinsed with methanol	[33]
Soaked with ultrapure water, acetone and n-hexane, then heated at 80 °C for 1 h	[34]
Soaked acetone, rinsed with hexane, dried at 400 °C for at least 4 h and wrapped with aluminum foil until analysis	[56]
Soaked in acetone for 30 min, rinsed with n-hexane, and then dried at 120 °C for 5 h	[60]
Nonvolumetric glassware was kept at 550 °C for 4–5 h in a muffle; volumetric glassware was cleaned for 24 h with a sulfuric acid solution (95% w/w)	[67–69]
Soaked in acetone for at least 30 min, then washed with acetone, rinsed with hexane, and dried at 120 °C for overnight	[74]
Soaked in 10% nitric acid, then rinsed three times with distilled water and then with pure acetone	[75]
Soaked with n-hexane, acetone and dichloromethane followed by drying at 4 h, 120 °C	[70]
Soaked using detergent, rinsed with distilled water, acetone, hexane, dichloromethane, isooctane followed by heating at 350 °C for 10 h	[76]
Soaked in concentrated sulfuric acid for 24 h and rinsed with ultrapure water, methanol, hexane and dried	[77]
Cleaned with detergent and rinsed with ultrapure water, immersed in acetone, sonicated for 30 min, rinsed with ultrapure water, and dried in oven at 100 °C for at least 3 h	[71]
Soaked with LC-grade hexane and then heated at 350 °C for 4 h	[72]

solvent prior to each injection may help reduce blank problems [20]. To the best of our knowledge, the most frequent source of contamination leading to errors during analysis is the inadequate cleaning of laboratory materials. In this way, various authors in the literature routinely apply specific cleaning practices to minimize background interference (see Table 3). As a rule, it is assumed that volumetric glassware should be pre-cleaned using oxidizing agents [17], while non-volumetric glassware can be calcined at 450–550 °C after being washed with ultrapure water and organic solvents, a process that significantly reduces contamination [21,26].

Moreover, one of the most effective strategies to limit cross-contamination during the analysis of plastics and related chemical compounds is to carefully select the materials that will come into contact with the sample throughout the analytical process. Although rinsing plastic materials with the aforementioned organic solvents has been shown to reduce contaminant levels, the use of appropriate materials is still recommended [19]. This includes using plasticizer-free consumables, such as filters, pipette tips, gloves, and vials, and prioritizing inert alternatives like glass, aluminum, Teflon, or stainless steel for components that interact with solvents or sample extracts [4,26]. To not overestimate plastic concentration, Prata et al. [27] propose to use thermally treated glass fiber filters, then store them in clean glass Petri dishes, which should be wrapped with aluminum foil.

Regarding gloves, if safety permits, critical items should ideally be handled without gloves after a rigorous hand-washing protocol, and contamination should always be verified using control blanks. Otherwise, it is recommended to use cleanroom-grade nitrile gloves that are powder-free, low-residue, free of accelerators, and without polymer coatings. The exterior of the gloves should be rinsed with ultrapure water, air-dried in a hood, and the glove brand, lot, and corresponding control blank documented for each batch. Vinyl, powdered, and polymer-coated gloves should be avoided [59], and neoprene or latex gloves used with caution, as stearates and other additives can leach and be mistaken for polymers. Polyethylene gloves should never be used when measuring polyethylene. Contact between gloves and analytical substrates should be minimized (using pre-cleaned stainless-steel tweezers), and the glove material or lot should be changed if contamination levels increase [29].

Moreover, replacing standard plastic tubing or storage containers with contaminant-free options reduces the likelihood of additive leaching, particularly under warm conditions or with prolonged contact [60]. Therefore, minimizing plastic packaging and avoiding long-term storage in conventional plastic containers is also advisable when working with compounds.

Working under laminar flow hoods, using lint-free cloths to clean surfaces, and wearing cotton lab coats or cleanroom garments helps limit the introduction of airborne fibers or dust [59]. For example, Wesch et al. [61] reported that air contamination by fibers was reduced by 50% when using fume hoods and by 97% when working under a laminar flow hood. Commonly used workspaces must be cleaned thoroughly before use, especially in shared environments. Ventilation systems should be equipped with high efficiency particulate air filters and maintained regularly to ensure they do not redistribute particles that could compromise sample integrity. Air quality should be monitored at defined intervals using particle counters or passive deposition plates to quantify background contamination. Laboratories operating under cleanroom or controlled-environment conditions typically follow ISO 14644 or equivalent standards, which specify acceptable particle concentrations and inspection frequencies. Concerning laboratory clothing, many researchers opt for cotton lab coats to minimize fiber contamination [62,63]. Some studies instead employ cotton test suit [64] or protective garments made from natural cotton and leather fibers [65]. However, even these natural fabrics may shed fibers, potentially contributing to background contamination [27].

Operator handling also requires attention, as direct contact through skin, breath, or clothing can easily introduce extraneous compounds into samples. Wearing gloves, masks, and hairnets, along with avoiding leaning over open containers, helps reduce this risk. It is advisable to use dedicated tools for each sample or implement cleaning protocols between uses to prevent cross-contact. Transfers should be carried out with pre-cleaned materials in a controlled work area, and procedural blanks should be included as part of routine good practice, particularly when handling multiple samples [6,24].

The selection and management of solvents and reagents must not be overlooked. Solvents should be of analytical grade or higher, tested using blank runs before use and purified with aluminum oxide [16,25]. In our view, these preliminary tests are extreme important, as even solvents purchased with high-purity specifications may still contain detectable residues, such as plasticizers. Reagents should ideally be stored in glass containers, as plastic bottles may release additives over time, particularly when containing alcohols or organic solvents. Ultrapure water, though widely used in laboratories, can also be a source of contamination if not regularly tested. To minimize water-related contamination, regular monitoring and maintenance of ultrapure water systems are essential. Filtration units should be periodically replaced, and water purity should be checked. When possible, freshly produced water should be used immediately rather than stored for extended periods, as storage in plastic carboys may promote leaching of polymer additives. Glass, aluminum or stainless-steel containers are preferred for short-term storage [66]. Filtration through membranes with pore sizes ≤ 0.2 μm prior to use can further reduce microplastic content, and dedicated blanks using the same water batch should be analyzed alongside samples to detect possible background contributions.

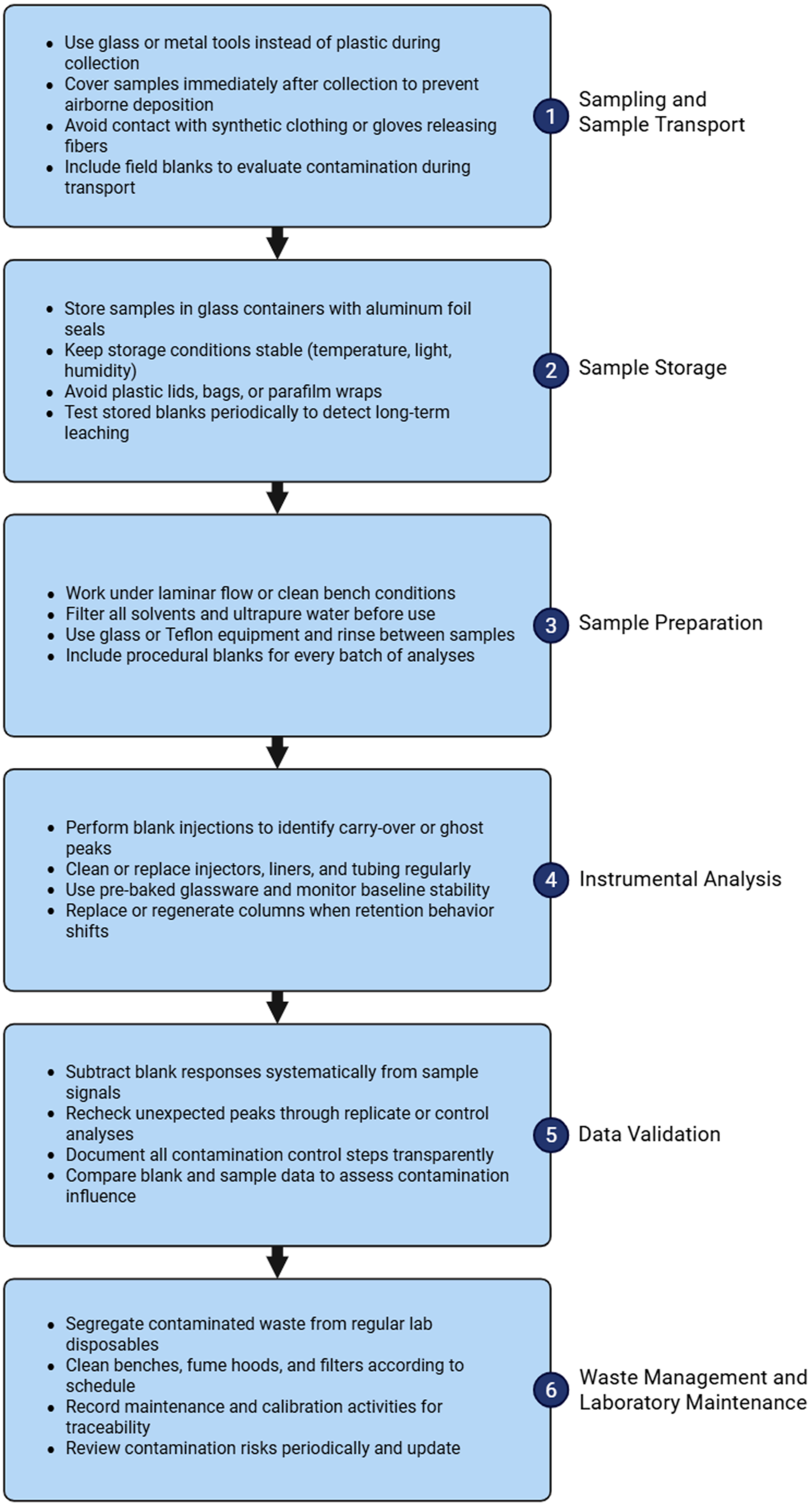
Regarding chromatography instruments injection ports, liners, inlets, and internal tubing can retain residues from previous runs if not properly maintained [16]. These components should be cleaned or replaced routinely, and contact surfaces flushed with solvent blanks when switching between sample types. Chromatographic columns also require attention, as strongly retained analytes may elute unpredictably across subsequent injections. Guard columns, routine monitoring of retention behavior, and replacement scheduling are all measures that contribute to cleaner and more consistent performance across runs. In cases where, despite following the aforementioned good laboratory practices, consistent signals of certain analytes are still observed in all procedural blanks, the blank response of the target compound(s) should be systematically subtracted from every analysis [4].

Minimizing cross-contamination is not only a matter of cleaning well, but of thinking carefully about contamination risks before starting any experiment. Researchers should incorporate contamination control into their experimental design from the beginning, evaluating all possible sources and weak points based on the analytes, solvents, containers, and instrumentation involved. This mindset, rooted in prevention rather than correction, not only enhances the analytical workflow and reduces the risk of errors, but also saves time and prevents unnecessary setbacks when dealing with such complex analytes.

To summarize the preventive key actions described above, Fig. 2 presents a practical workflow for minimizing cross-contamination during the analysis of plastics and related compounds. However, given the limited information available on the effectiveness of these measures, it remains challenging to quantify the reduction in contamination achieved by each individual action, or to estimate an overall percentage of pollution reduction resulting from their combined implementation.

6. Perspectives

Looking ahead, future efforts should focus on promoting more consistent contamination control strategies in laboratories, particularly in studies involving microplastics, plastic additives, and environmental or food-related matrices. Despite the growing attention to plastic-related pollution, there remains a lack of standardized protocols for addressing cross-contamination in routine analytical workflows. Moreover, the absence of standardized methodologies for sampling and analysis means that results are often not comparable due to variations in the number of replicates, the size range of the studied microplastics, the reported concentration units, and the sampling depth. The development and broader adoption of guidelines that explicitly detail cleaning procedures, solvent testing, and selection of low-contamination-risk materials could significantly enhance data comparability and reduce analytical uncertainty. In addition, investment in the design of new types of labware such as low-adsorption consumables, solvent-resistant glassware coatings, or contamination-proof storage solutions, would be valuable to mitigate the limitations of current materials. Beyond technical improvements, contamination control should not be treated as a secondary consideration or post-analysis troubleshooting step, but as a part of



1 Sampling and Sample Transport

- Use glass or metal tools instead of plastic during collection
- Cover samples immediately after collection to prevent airborne deposition
- Avoid contact with synthetic clothing or gloves releasing fibers
- Include field blanks to evaluate contamination during transport

2 Sample Storage

- Store samples in glass containers with aluminum foil seals
- Keep storage conditions stable (temperature, light, humidity)
- Avoid plastic lids, bags, or parafilm wraps
- Test stored blanks periodically to detect long-term leaching

3 Sample Preparation

- Work under laminar flow or clean bench conditions
- Filter all solvents and ultrapure water before use
- Use glass or Teflon equipment and rinse between samples
- Include procedural blanks for every batch of analyses

4 Instrumental Analysis

- Perform blank injections to identify carry-over or ghost peaks
- Clean or replace injectors, liners, and tubing regularly
- Use pre-baked glassware and monitor baseline stability
- Replace or regenerate columns when retention behavior shifts

5 Data Validation

- Subtract blank responses systematically from sample signals
- Recheck unexpected peaks through replicate or control analyses
- Document all contamination control steps transparently
- Compare blank and sample data to assess contamination influence

6 Waste Management and Laboratory Maintenance

- Segregate contaminated waste from regular lab disposables
- Clean benches, fume hoods, and filters according to schedule
- Record maintenance and calibration activities for traceability
- Review contamination risks periodically and update

Fig. 2. Workflow for cross-contamination prevention during plastic-related analytical processes.

experimental design.

7. Conclusions

The widespread use of plastics and plastic-derived chemicals in laboratories, as well as in daily life, makes the complete avoidance of contamination an unrealistic goal. However, sample contamination by these compounds cannot be ignored. This work provides an overview of the main cross-contamination pathways during the analysis of plastics and related compounds. From the materials used for sample handling to environmental exposure, human interaction, reagent purity, and instrument maintenance, every step of the analytical process holds potential for introducing artifacts that may interfere with the accuracy of results. While many contamination sources can be mitigated through proper planning, meticulous cleaning, and conscious material selection, others remain difficult to eliminate due to the pervasive presence of plastic additives in research settings. Even when best practices are implemented, background levels may persist. Therefore, it is recommended to include the routine analysis of procedural blanks to monitor contamination and assess its effect on method sensitivity. When a contaminant is detected in the blanks, the average signal from the replicates should be subtracted from the corresponding sample signal to ensure reliability.

Ensuring reliable and comparable analytical results requires the consistent implementation of contamination control measures across laboratories. Good practices include wearing clean cotton lab coats and sterile gloves, conducting work in controlled environments such as laminar flow hoods, and exclusively using glass or metal materials thoroughly cleaned between samples. All working solutions should be pre-filtered and stored in closed glass containers, while high-quality filters must be pre-heated or properly decontaminated before use. Moreover, materials and samples should remain covered to prevent airborne contamination, and the inclusion of field and procedural blanks is crucial to identify potential contamination sources.

Moreover, the good laboratory practices and preventive recommendations discussed in this work are not limited to studies on microplastics or plasticizers. They are equally relevant to other types of analytes and experimental designs, such as biological or toxicological assays, where unintended contamination may compromise data reliability, obscure true dose–response relationships, or lead to misleading conclusions. We encourage researchers to reflect on the potential contamination risks at every stage of their workflow and to document them transparently in their publications.

Abbreviations

ATR, attenuated total reflectance; **DSC**, differential scanning calorimetry; **FTIR**, Fourier transform infrared; **GC**, gas chromatography; **GC-MS**, gas chromatography–mass spectrometry; **HEPA**, high efficiency particulate air; **LC**, liquid chromatography; **LC-MS**, liquid chromatography–mass spectrometry; **TGA**, thermogravimetric analysis.

CRediT authorship contribution statement

Adrián Fuente-Ballesteros: Writing – review & editing, Writing – original draft, Visualization, Supervision, Methodology, Investigation, Conceptualization. **Reem H. Obaydo:** Writing – review & editing. **Samar H. Elagamy:** Writing – review & editing. **Ana M. Ares:** Writing – review & editing, Visualization. **José Bernal:** Writing – review & editing, Visualization, Supervision.

Declaration of Competing 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.

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Data availability

No data was used for the research described in the article.

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