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Rinse, Reuse, Repeat: Sustainable Solutions For Lab Plastics

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Abbreviations

ACC	Automated Cell Culture
AUC	Area Under the Curve
BSA	Bovine Serum Albumin
CO_2e	Carbon Dioxide Equivalent
dBA	A-weighted decibel values
DI	Deionized
ECHO	Echocardiogram
GHG	Greenhouse Gases
PE	Polyethylene
PP	Polypropylene
PS	Polystyrene
ROI	Return On Investment
UVC	Ultraviolet C

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Abstract

Single-use plastics in cell culture laboratories represent a major source of plastic waste and greenhouse gas emissions, contributing to Scope 3 emissions in healthcare and research sectors. Pipette tips and microwell plates, predominantly made of polypropylene and polystyrene, are widely used in high-throughput workflows and are typically incinerated after one use, generating substantial CO_2 emissions. This thesis investigates the feasibility of implementing automated washing systems (TipNovus and PlateNovus) to enable controlled reuse of these consumables, thereby reducing environmental impact without compromising data integrity.

A comprehensive evaluation was performed, including gravimetric calibration, microscopic inspection, sealing-rim adhesion tests and cell-based assays on Greiner 384-well plates and impedance-based E-Plates. Washed pipette tips retained volumetric accuracy within ISO 8655 limits and showed no detectable morphological changes after up to 12 washing cycles. Five times washed microwell plates supported cell growth and pharmacological responses comparable to new plates, with EC_{50} values remaining within acceptable ranges. Cleanliness assays (Bradford, Fluorescein, Neutral Red) confirmed effective removal of protein and chemical residues, and sterility monitoring demonstrated that scheduled decontamination eliminated microbial growth inside washers.

Environmental and economic analyses revealed significant benefits. Washing reduced CO_2 emissions by 98% for pipette tips and 65% for plates, corresponding to an annual reduction of ~1.53 metric tons carbon dioxide equivalent (CO_2e) at the tested throughput. Cost modeling indicated a wash cost of €2.48 per tip box and €2.60 per plate, compared to purchase prices of €15 and €7, resulting in annual savings of ~€63,000. With a combined capital investment of €200,000, the estimated return on investment (ROI) is 3.2 years, after which the systems generate recurring cost savings alongside measurable sustainability gains.

These findings demonstrate that automated washing workflows can be integrated into regulated laboratory environments without compromising sterility or assay performance. Implemented under validated protocols and hygiene controls, reuse strategies offer a scalable solution to reduce plastic waste, lower operational costs, and align laboratory practices with global sustainability objectives.

Zusammenfassung

Einwegkunststoffe in Zellkulturlaboren tragen erheblich zu Plastikabfall und Treibhausgasemissionen bei und stellen einen relevanten Anteil der Scope 3 Emissionen im Gesundheits- und Forschungssektor dar. Pipettenspitzen und Wellplatten aus Polypropylen bzw. Polystyrol werden in Hochdurchsatz-Workflows in großen Mengen eingesetzt und nach einmaligem Gebrauch üblicherweise verbrannt, was die Umweltbelastung zusätzlich erhöht. Ziel dieser Arbeit ist, die Machbarkeit und Leistungsfähigkeit automatisierter Waschsysteme (TipNovus und PlateNovus) als nachhaltige Alternative zu Einwegmaterialien zu untersuchen.

Die Bewertung umfasst gravimetrische Kalibrierungen, mikroskopische Analysen, Haftungstests, sowie zellbasierte Assays mit Greiner-384-Well-Platten und Impedanz-basierten E-Platten. Gewaschene Pipettenspitzen erfüllten die ISO-8655-Toleranzen für Volumengenauigkeit und zeigten keine strukturellen Veränderungen nach bis zu zwölf Waschzyklen. Gewaschene Platten unterstützen Zellwachstum und pharmakologische Reaktionen in vergleichbarer Qualität zu neuen Platten. EC_{50}-Werte bleiben innerhalb akzeptabler Grenzen, auch nach bis zu fünf Waschläufen. Rückstandsanalysen (Bradford-, Fluorescein- und Neutralrot-Assays) bestätigen die effektive Entfernung biologischer und chemischer Kontaminationen und die Sterilitätsüberwachung zeigt, dass Dekontaminationsprotokolle mikrobielle Belastungen vollständig beseitigen.

Die Umwelt- und Wirtschaftsanalyse verdeutlicht den Nutzen dieser Strategie: Das Waschen reduziert die CO_2 Emissionen um 98 % bei Pipettenspitzen und 65 % bei Platten, was einer jährlichen Einsparung von rund 1,53 Tonnen Kohlendioxyd äquivalent (CO_2e) entspricht. Die Waschkosten liegen bei etwa 2,48 € pro Spitzenbox und 2,60 € pro Platte, verglichen gegenüber Anschaffungskosten von 15 € pro Box bzw. 7 € pro Platte. Daraus ergeben sich jährliche Einsparungen von etwa 63.000 €. Bei einer Gesamtinvestition von 200.000 € beträgt die Amortisationszeit (ROI) rund 3,2 Jahre. Nach diesem Zeitraum generieren die Systeme fortlaufende Kostenvorteile bei gleichzeitiger Reduktion von Plastikabfall und Emissionen.

Die Ergebnisse zeigen, dass automatisierte Waschprozesse unter validierten Protokollen und Hygieneauflagen in regulierte Laborumgebungen integriert werden können, ohne die Datenintegrität zu beeinträchtigen. Wiederverwendungsstrategien bieten damit eine skalierbare Lösung, um ökologische Nachhaltigkeit mit ökonomischer Effizienz in der biomedizinischen Forschung zu verbinden.

1 Introduction

1.1 Plastic pollution as a driver of climate change

Climate change refers to long-term alterations in temperature and weather patterns, primarily driven by anthropogenic activities such as the combustion of fossil fuels, deforestation, and industrial operations. These processes elevate the concentration of greenhouse gases (GHGs), including carbon dioxide and methane, in the atmosphere, thereby trapping heat and disrupting the earth's climate systems[1]. The consequences of climate change are increasingly severe, contributing to natural disasters and premature mortality worldwide. In 2015, pollution was likely responsible for approximately nine million premature deaths globally, accounting for 16% of all deaths[2].

Commercial GHG emissions are commonly grouped into three categories: Scope 1, Scope 2, and Scope 3 emissions[3]. Scope 1 emissions are direct emissions from sources owned or controlled by an organization, such as on-site fuel combustion. Scope 2 emissions are indirect emissions resulting from the consumption of purchased energy, including electricity, steam, and district heating. Scope 3 emissions, which represent the largest share, encompass all other indirect emissions across the value chain. In the U.S. healthcare sector Scope 3 emissions account for approximately 83% of total emissions [4] (**Fig. *1***a). These include emissions from the production and transportation of goods, as well as downstream processes such as recycling and waste management.

An often-overlooked contributor to Scope 3 emissions is plastic pollution. Plastics, derived from fossil fuels, release substantial amounts of GHGs throughout their life cycle from raw material extraction to end of life incineration[1]. Climate change and plastic pollution are closely interconnected. Extreme weather events, intensified by climate change, can disperse plastic waste into natural ecosystems, while marine environments already stressed by rising temperatures are further degraded by plastic debris and microplastic particles. These microscopic fragments persist in soil and water, infiltrating food chains and amplifying ecological and health risks. Addressing these dual crises requires integrated strategies that simultaneously reduce plastic consumption and GHG emissions, while promoting circular economy principles to mitigate the environmental impact of plastic materials[1].

1.2 Cell culture laboratories as major contributors to plastic waste

The healthcare sector alone generates approximately 5.5 million tons of plastic waste annually, significantly contributing to environmental degradation[5]. The climate footprint of the healthcare industry, including the biotechnology and pharmaceutical sectors, is substantial, with emissions totaling two gigatons of CO_2 equivalent (CO_2e), representing 4.4% of global emissions[6]. For instance, each 96-tip pipette rack is associated with approximately 296g of CO_2e emissions[7]. When extrapolated to the total number of racks used globally in one year, this corresponds to an estimated 7.7 million metric tons

of CO_2e emissions annually[8]. To offset this amount, approximately 127 trees would need to grow over a ten-year period[9]. This considerable environmental impact highlights the urgent need for sustainable practices in laboratory environments, where plastic waste is particularly prevalent. Laboratories, especially those utilizing tissue and cell culture techniques, are among the largest contributors to plastic waste within the health care sector[10]. Approximately 72% of this waste consists of polystyrene (PS) and polypropylene (PP), while the remaining portion includes polyethylene (PE), nitrile, and other plastics such as packaging materials commonly used in cell culture environments (**Fig. *1***b).

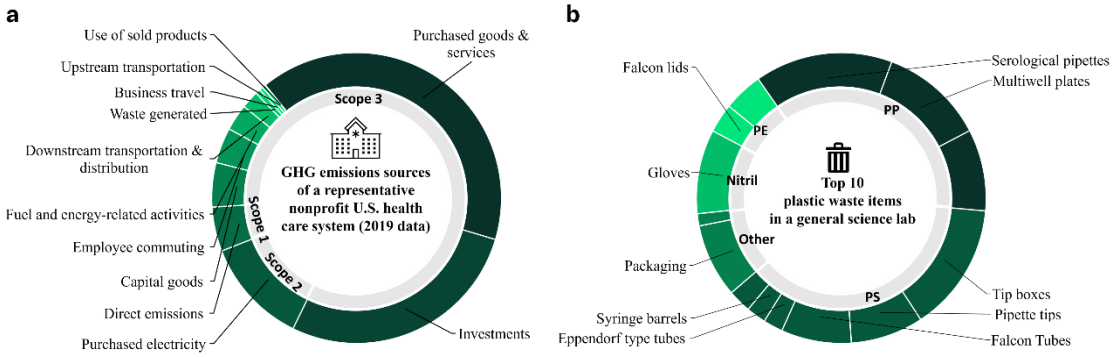

Fig. 1 | Laboratory contributions to waste and emissions.

a, Breakdown of GHG emissions for a representative nonprofit U.S. healthcare system in 2019: Scope 1 (direct emissions) = 5%, Scope 2 (purchased electricity) = 12%, and Scope 3 (indirect emissions) = 83%. Laboratory consumables such as single-use plastics fall under Scope 3 (Zigmund 2024). **b,** Material composition of laboratory plastic waste: polypropylene (PP) = 39% (mainly serological pipettes 16.1%, multiwell plates 12.2%), polystyrene (PS) = 33% (tip boxes 15.0%, pipette tips 8.5%, Falcon tubes 8.2%), polyethylene (PE) = 8%, nitrile = 10% (mostly gloves 9.8%), and other plastics = 10% (primarily packaging 8.6%) (Weber 2025).

PS is produced through free-radical polymerization, in which styrene monomers are linked into long polymer chains. PS is favored in laboratory settings due to its high optical clarity, ease of manufacturing, and low production cost. Its relatively high hardness also contributes to its robustness and durability[11]. In cell culture laboratories, PS is commonly used for manufacturing tissue culture well plates and serological pipettes (**Fig. *2***a). PP, on the other hand, is synthesized from propylene, a byproduct of crude oil processing, using various polymerization techniques[12]. PP is widely used due to its excellent processability, chemical resistance, and moisture barrier properties. In laboratory applications, PP is employed in the production of durable and chemically resistant equipment such as pipettes, rack boxes, and tubes (**Fig. *2***b). These materials are central to routine operations, including liquid handling with pipette tips and the use of microwell plates for a wide range of experiments. Pipette tips and microwell plates are highly standardized consumables in these contexts and are widely used at high throughput in daily laboratory practice.

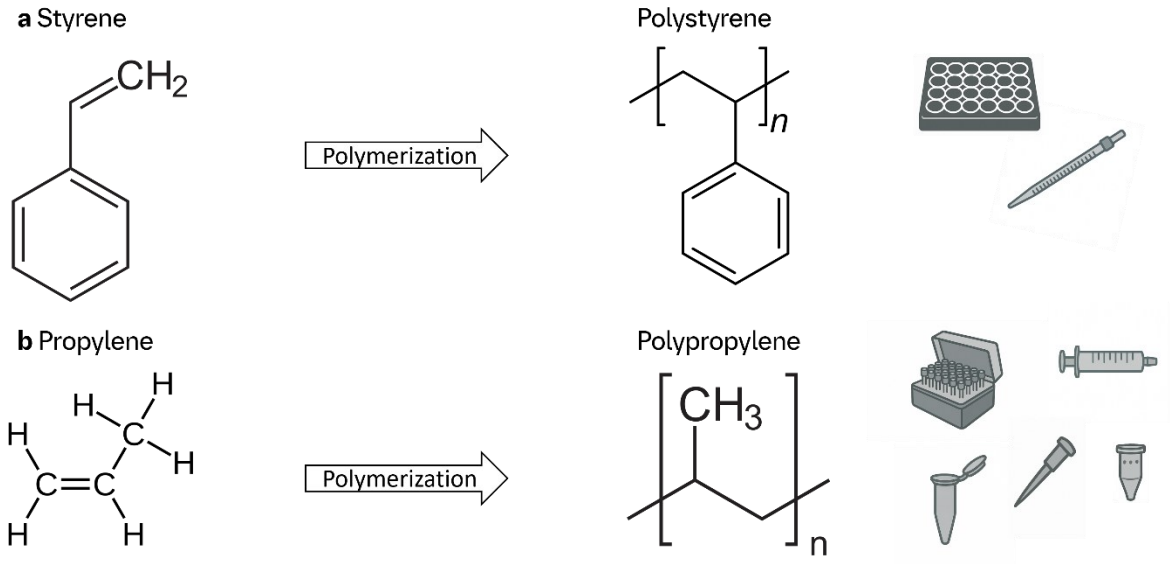

Fig. 2 | Polymerization of Styrene and Propylene.

a, Polymerization of styrene (C_8H_8) into PS. Laboratory items commonly manufactured from PS, including a multi-well plate and a pipette. **b,** Polymerization of propylene (C_3H_6) into PP. Laboratory items made from PP are displayed next to the polymer structure, including pipette tip racks, syringes, tubes, and pipette tips. The corresponding laboratory equipment icons were created using Copilot.

Although both PS and PP are technically recyclable, practical implementation in laboratory settings faces specific challenges. PP is considered highly recyclable but depends on uncontaminated, segregated collection streams to maintain material quality[12]. PS, while recyclable, is hindered by low density, which makes transportation and storage inefficient because large volumes of lightweight material occupy significant space while contributing little weight. This increases logistical costs and complicate collection and processing[13]. A major barrier in laboratories is contamination with biological or chemical substances, which places items under hazardous-waste handling and limits suitability for mechanical recycling within circular economy schemes. Consequently, sizable fractions of laboratory plastics are incinerated rather than recycled, releasing additional CO_2 emissions even when base polymers are in principle recyclable.

1.3 Sustainability in cell culture laboratories

In response to the challenges posed by single-use plastics and high resource consumption, major pharmaceutical companies have implemented Green Lab initiatives to reduce the environmental footprint of research operations. AstraZeneca's Green-Labs-Program emphasizes energy optimization, waste reduction, and sustainable procurement[14] while Roche integrates sustainability into laboratory practices through energy efficiency, waste management, and circular economy principles[15]. Novartis has advanced similar strategies, including greener clinical trials[16]. Manufacturers are also innovating, Eppendorf introduced BioBased polypropylene for pipette tips and tubes, using recycled vegetable oil

to replace fossil-based raw materials, which reportedly cuts CO_2 emissions by up to 68%[17]. Other suppliers, such as Wildcat Laboratory Solutions in collaboration with PulpFixin, offer compostable paper racks as alternatives to plastic[18]. Many of these efforts align with My Green Lab, a globally recognized non-profit organization that certifies laboratories and partners with the UN's Race to Zero campaign[19]. Collectively, these initiatives illustrate an industry-wide commitment to embedding sustainability into laboratory workflows.

Among the emerging technology providers, Grenova, a U.S.-based company specializing in sustainable laboratory solutions, has developed automated washing systems for pipette tips and microwell plates[20]. These systems integrate high-pressure rinsing, UV sanitation, sonication, and proprietary cleaning agents to enable multiple reuse cycles without compromising sterility or functional integrity. In contrast, IonField employs a different approach based on low-temperature plasma technology[21]. This method supports both pipette tips and microwell plates and operates as a dry, chemical-free alternative to conventional washing. For pipette tips, the process involves sequential aspiration and dispensing of plasma within the tips. For microplates, the workflow combines micro-dispensing, high-pressure rinsing, centrifugation, and plasma exposure to achieve molecular-level removal of contaminants. Both technologies exemplify practical implementations of circular economy principles in laboratory environments by mitigating single-use plastic waste and associated greenhouse gas emissions.

1.4 Automated washing technologies for reusable laboratory consumables

Building on these sustainability principles, Grenova's TipNovus and PlateNovus system exemplifies an applied approach to automated washing technology designed for high-throughput laboratory environments.

1.4.1 TipNovus

The TipNovus system is designed to wash up to four racks of manual or automated tips in 24, 48, 96, or 384 formats in a single cycle. Washing protocols are customizable, using only distilled water and an eco-friendly washing reagent called GrenoClean[22]. Depending on specific laboratory requirements, alternative reagents may also be used.

GrenoClean is formulated to meet stringent safety standards and is not classified as hazardous under OSHA (29 CFR 1910.1200) or WHMIS 2015 criteria. It is prepared in Type II deionized water (DI) and contains sodium percarbonate and sodium bisulfate. The solution maintains a pH of 9.1 ± 0.1 and remains stable for up to twelve hours under appropriate conditions[23].

TipNovus is certified under ISO 9001 and ISO 13485, ensuring compliance with high-quality management systems and regulatory standards for medical devices[24]. The system comprises three main components: the control box, the TipNovus-washer and the TipNovus-dryer. The control box integrates

electrical, pneumatic, and kinetic systems to manage compressed air, reagent delivery, and waste disposal. The dryer facilitates rapid drying of pipette tips through a combination of directed airflow and mechanical agitation. The washer is divided into an upper section for liquid dispensing, a middle section containing the sealed wash chamber, and a lower section housing the electrical supply for sonication, UV illumination, and drawer actuation.

1.4.2 PlateNovus

The PlateNovus system is engineered for cleaning and reuse of standard SLAS-format 96- and 384-well microplates. It supports routine laboratory workflows by integrating multiple cleaning modalities that ensure effective decontamination. The system can process up to four microplates simultaneously, with a maximum plate height of 50 mm. It is compatible with a wide range of plate types and is suitable for use across various scientific disciplines.

1.4.3 TipLumis

TipLumis is a controlled storage unit for maintaining the sterility of washed pipette tips and microplates. It holds up to 45 rack carriers across nine compartments and ensures cleanliness through active heating, UV irradiation, and HEPA filtration. When a new carrier is inserted or the system is started manually, a sanitation cycle that includes brief UV exposure from top and bottom sources, followed by a heating phase at 40 °C, is initiated. This sequence is repeated to complete a three-hour decontamination process. After the cycle, the system returns to ambient temperature. Parameters such as cycle duration and UV activation intervals can be adjusted by Grenova support.

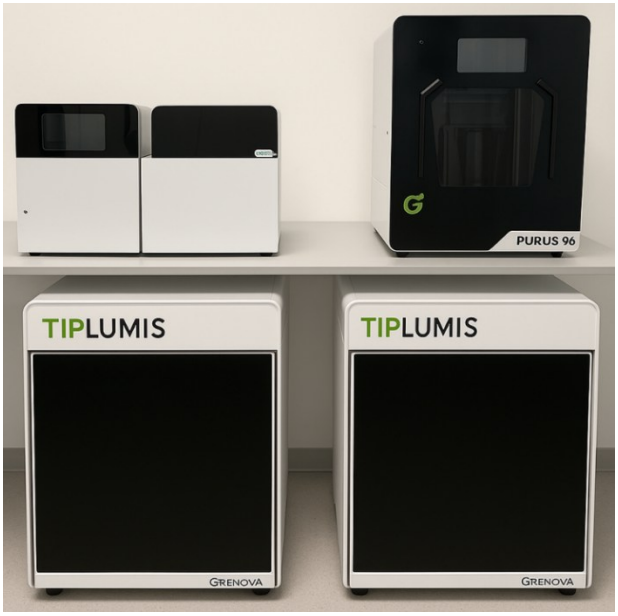

Fig. 3 | Overview of Grenova laboratory devices.

Top left: TipNovus system, consisting of the washing unit (left) and the dryer (right). Top right: PlateNovus unit for automated plate washing. Bottom: Two TipLumis units designed for storage and drying of pipette tips and microwell plates.

1.5 Material integrity and performance of reusable laboratory plastics

Automated washing and controlled storage systems such as TipNovus, PlateNovus and TipLumis address critical aspects of sustainability and sterility in laboratories. While these technologies ensure that consumables can be safely reused, their effectiveness ultimately depends on the physical properties of the items being processed. Pipette tips and microplates are precision-engineered components that directly influence experimental accuracy.

1.5.1 Pipette tips

Pipettes are highly calibrated instruments used to transfer defined volumes of liquid in laboratory settings. Most commonly, air displacement pipettes are used in combination with disposable plastic tips[25]. These tips play a critical role in ensuring accurate and reproducible liquid handling. Their structural integrity and surface quality directly influence the reliability of experimental results.

To evaluate whether repeated washing affects the performance of pipette tips, specific quality parameters need to be defined. Key structural features of a pipette tip include the sealing rim, which ensures a secure connection to the pipette cone, the height, which determines the volume capacity and the orifice, the lowest point through which liquid is aspirated and dispensed. The surface quality of the orifice is particularly critical, as irregularities can lead to droplet deflection, residual liquid, or inaccurate volume transfer [26].

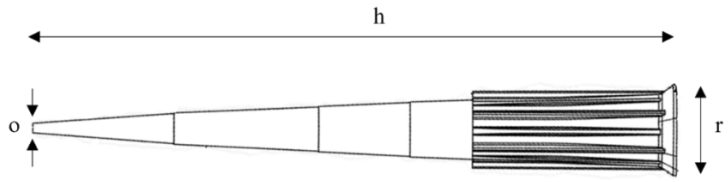

Fig. 4 | Describing parameters of a pipette tip.

h (height), r (sealing rim) and o (orifice)[27].

1.5.2 Microwell plates

In addition to pipette tips, microwell plates constitute another fundamental class of plastic-based laboratory consumables. They are especially vital in cell culture and high-throughput screening workflows. Designed to enable the parallel processing of multiple samples under standardized conditions, microwell plates have become indispensable tools in modern experimental setups. According to ANSI/SLAS standards, which define the dimensions and automation compatibility of microplates, these plates are commonly available in 6-, 24-, 96-, 384-, and 1536-well formats to meet a wide range of experimental requirements.

Microwell plates in cell culture, like Greiner 384-microwell plates, are predominantly manufactured from PS, a material chosen for its optical clarity, rigidity, and cost-effectiveness. For cell culture applications, the surface of the wells is often physical treated, commonly using gas plasma, to enhance cell adhesion and growth.

E-Plates are specialized cell culture microplates equipped with integrated gold microelectrodes at the bottom of each well, enabling label-free, real-time monitoring of cellular events through electrical impedance measurements. When cells adhere and spread across the electrode surface, they partially obstruct the electrical current, causing a measurable increase in impedance. This signal, expressed as the Cell Index, provides insights into cell number, morphology, and adhesion strength. The integrity of the gold electrodes is essential for assay performance, as any physical damage or residual contamination can impair signal transmission and compromise data quality. In pharmacological applications, impedance-based parameters such as the normalized Cell Index (nCI) are tracked over time, allowing calculation of metrics like the area under the curve (AUC) to quantify cumulative responses following agonist stimulation. These assays typically employ cells expressing the relevant receptor, ensuring that ligand binding triggers a detectable and physiologically meaningful response.

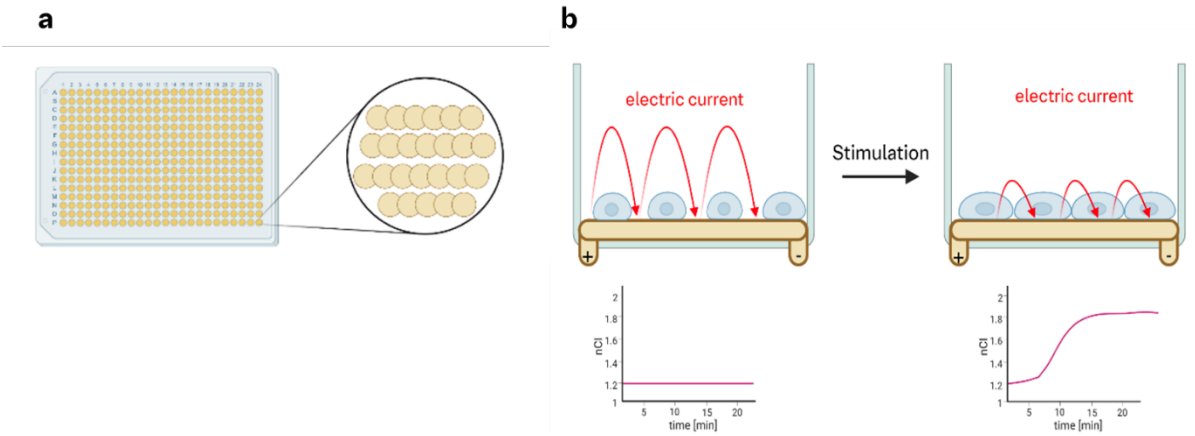

Fig. 5 | Impedance-based cell monitoring assay using a 384-well E-Plate.

a, Top view of the plate with integrated gold electrodes at the bottom of each well. **b,** Side view illustrating electric current flow before and after stimulation. The corresponding impedance signal (Cell Index) remains stable prior to stimulation and increases upon cellular response, reflecting changes in adhesion and morphology. Image provided by Frederik Bangnowski.

1.6 Aim of the study

The extensive use of single-use plastics in laboratory environments represents a major contributor to environmental pollution and resource depletion, thereby intensifying global challenges such as climate change and plastic waste accumulation. Implementing sustainable practices in research workflows is therefore of increasing importance. This thesis aims to identify strategies for reducing single-use plastic in laboratory settings by evaluating the feasibility and performance of tip- and plate washing systems as a potential solution. Grenova's devices are employed as a case study to examine their effectiveness and environmental impact compared to conventional single-use approaches.

The evaluation focuses on (I) the preservation of critical quality attributes such as sterility, physical integrity, and functional performance after repeated washing cycles, (II) the effectiveness of the washing process in removing biological and chemical contaminants, (III) the potential integration of the washing system into existing workflows at Boehringer Ingelheim, (IV) the estimated reduction in single-use plastic waste and associated GHG emissions compared to conventional single-use practices and (V) the assessment of cost aspects, evaluating economic feasibility of implementing the washing systems.

2 Materials and Methods

2.1 Process efficiency and environmental impact of automated washing devices

Performance assessment encompasses protocol complexity, operational emissions, and resource demand. Cleaning workflows for TipNovus and PlateNovus are characterized by multi-step, sensor-controlled sequences. Acoustic measurements quantify noise exposure during critical phases, while energy profiling determines consumption patterns across washing, drying, and storage.

2.1.1 Cleaning protocols for TipNovus and PlateNovus

The cleaning process of TipNovus follows a custom protocol adapted to the tip size. It starts with an initial rinse with deionized water and UV exposure for approximately two minutes. This is followed by a low-level soaking phase with DI water, sonication, and UV for around nine minutes. A GrenoClean purge step under continued UV exposure is then applied for about two minutes. Subsequently, a high-level soak with sonication and DI water is performed for approximately ten minutes, fully submerging the tips to enhance particle removal. The cycle concludes with a final three-minute wash phase using DI water and UV[22]. The complete washing cycle in TipNovus takes approximately 26 minutes (**Fig. *6***a).

The cleaning procedure applied for washing microwell plates in PlateNovus consists of four integrated subprotocols. The process begins with a two-minute rinse phase, during which each microwell plate receives four GrenoClean purges under UV illumination. In the second subprotocol, the plates are soaked in DI water under continuous sonication and UV exposure while being tilted at a 30-degree angle and gently agitated for 330 seconds per plate. The third subprotocol involves a second soak in DI water with sonication and UV, applied for 100 seconds per plate. The final subprotocol consists of a rinse with DI water and UV exposure. At the end of each subprotocol, the plates are spun at 400 rpm for 30 seconds to remove residual liquid. Following the last rinse, an additional final spin is performed under the same conditions to enhance drying. The total duration of the cleaning cycle of four pates is approximately 50 minutes[28] (**Fig. *6***b).

These processes are executed in a programmable sequence via a touchscreen interface, allowing users to tailor protocols to specific tip/ plate types and contamination profiles. Sensor arrays continuously monitor reagent levels, air pressure, UV lamp status, and waste overflow to ensure consistent and safe operation.

Fig. 6 | Cleaning protocols for TipNovus and PlateNovus systems.

a, The TipNovus protocol includes five steps: DI-Rinse (2 min) flushes loose particles and applies UV light for initial disinfection. Soak Low (9 min) immerses tips in DI water with UV light and sonication to loosen embedded contaminants. PreWash (2 min) uses GrenoClean solution and UV light to remove remaining residues. Soak High (10 min) cleaning with UV exposure, sonication, and DI-water. DI-Wash (3 min) completes the cycle with mechanical agitation and UV light. Total duration: 26 minutes for four tip racks. **b,** The protocol consists of four steps: Rinse (2 min) flushes surface contaminants using GrenoClean and UV light. Soak (30 min) loosens embedded residues with deionized water, UV light, and sonication. A second DI-Soak (15 min) combines UV light and sonication to dislodge remaining particles. DI-Wash (3 min) completes the process with mechanical agitation and UV light to ensure thorough removal of water and reagents. Total duration: 50 minutes for four plates. Icons were created using Copilot.

2.1.2 Noise level measurements during operational cycles

To assess the acoustic footprint of Grenova devices during operation, noise level measurements were performed across four key process steps: tip washing, tip drying, and self-cleaning using TipNovus, as well as plate washing using PlateNovus. According to the user manual, TipNovus and PlateNovus are expected to emit sound levels of approximately 75 dBA, respectively. To verify these specifications under laboratory conditions, sound profiles were recorded using a sound level meter, capturing A-weighted decibel values (dBA) at 0.5-second intervals. Measurements were taken at three locations: directly adjacent to the device (approx. 0.5 m), at the opposite end of the laboratory (approx. 5 m) and in an adjacent room separated by a standard laboratory door.

2.1.3 Energy consumption of washing and drying devices

Following the acoustic evaluation, the energy efficiency of the washing and storage processes was examined to further characterize the operational footprint of the Grenova system. According to the manufacturer, TipNovus operates at up to 1100 W, while PlateNovus reaches a maximum of 750 W[22,28]. Energy profiles were recorded at one-minute intervals using Voltcraft energy logger monitoring systems during the following operational steps: tip washing and drying in TipNovus, plate washing in PlateNovus, and storage of tips and plates in TipLumis. Due to the limited sampling frequency, multiple runs of each process were performed to obtain representative datasets. The average energy consumption

for each step was calculated by determining the area under the curve (AUC) using GraphPad Prism software.

2.2 Quality assessment of laboratory articles

To assess whether washing and reusing single-use laboratory plastics compromises their integrity and continued suitability for use, specific quality parameters were defined. In this study, both new and previously washed pipette tips and microwell plates were systematically compared.

2.2.1 Consumables for comparative analysis

The tested consumables included:

- Eppendorf epT.I.P.S. tips in 0.1–10 µL (dark grey), 2–200 µL (yellow), and 50–1000 µL (blue)
- Sartorius Optifit tips in 0.1–10 µL (grey), 0.5–200 µL (yellow), 5–350 µL (orange), and 50–1200 µL (purple)
- Greiner-Cell-Culture-384 microwell plates
- Agilent Technologies 384-E-Plates

Eppendorf tips were washed 3, 6, 9, and 12 times, Sartorius tips 4, 8, and 12 times, while Greiner microwell plates underwent up to 5 washing cycles and E-Plates were washed 3 times.

In addition to standard cleaning using the TipNovus and PlateNovus systems, accelerated aging was simulated to evaluate the combined long-term effects of repeated washing with GrenoClean detergent and subsequent drying on the material integrity of pipette tips and microwell plates. Accelerated stability testing is commonly used in food, chemical, and pharmaceutical research to simulate long-term exposure within a shorter timeframe. It relies on the principle that chemical and physical degradation processes occur more rapidly at elevated temperatures[29]. This principle was applied to simulate 100 washing and drying cycles of pipette tips and microwell plates.

For the washing simulation, pipette tips were placed in a glass container filled with preheated GrenoClean at 40 °C and incubated in a water bath at the same temperature for 57 minutes and 36 seconds. This duration was calculated based on a reference temperature of 25 °C, a target temperature of 40 °C, a total exposure time of 10,000 seconds (equivalent to 100 washing cycles), and an estimated Q_{10} value of 2. Microwell plates were incubated in GrenoClean for 72 minutes, reflecting the cumulative rinse time in PlateNovus (140 seconds per cycle). After incubation, the GrenoClean solution was discarded, and the items were rinsed with deionized water. Drying simulation followed immediately. Pipette tips were placed in a glass bowl and transferred to a heating chamber at 80 °C for 4 hours, 4 minutes, and 48 seconds, simulating 100 drying cycles of 10 minutes each at 60 °C. Microwell plates

were dried at 60 °C for 3 days, 3 hours, and 12 minutes, corresponding to 100 drying cycles in TipLumis, each lasting 3 hours at 40 °C.

2.2.2 Structural and functional integrity of pipette tips

To assess quality parameters and evaluate consumables quality after washing, several analytical methods were applied. Gravimetric analysis was used to measure the accuracy of liquid dispensing across different volumes. Light microscopy enabled the examination of the tip orifice, to detect any deformation or surface irregularities. Additionally, the sealing rim was evaluated regarding sealing quality and mechanical stability, ensuring that no leakage or detachment occurred during simulated pipetting routines.

Gravimetric accuracy test

Pipetting accuracy was evaluated using gravimetric calibration, which assumes that 1 mg of water equals 1 µL of volume. This relationship is valid under controlled conditions: 20 °C room temperature, 50% relative humidity, 101.3 kPa barometric pressure, and grade 3 DI water. According to ISO 8655-2:2002, air-displacement pipettes with variable volumes must meet defined limits for systematic error[30]. These limits depend on the nominal volume and increase with larger capacities: 20 µL pipette: max systematic error = 0.2 µL, 200 µL pipette: max systematic error = 1.6 µL, 350 µL pipette: max systematic error ≈ 2.8 µL and 1000 µL pipette: max systematic error = 8.0 µL.

To investigate whether repeated washing affects the performance of pipette tips, washed Eppendorf epT.I.P.S. and Sartorius Optifit tips were compared with new tips of the same type. Gravimetric measurements were performed using the corresponding pipette for each tip type (**Table *1***). Each tip was pre-wetted by aspirating and dispensing water once before the actual measurement. The target volume was then dispensed into a small tube placed on a fine analytical balance. For Eppendorf epT.I.P.S., two volumes were tested per tip size: 5 µL and 10 µL for dark grey tips, 20 µL and 200 µL for yellow tips, and 100 µL and 1000 µL for blue epT.I.P.S. For Sartorius Optifit tips, only the upper nominal volumes were tested: 10 µL for grey tips, 200 µL for yellow tips, 300 µL for orange tips, and 1200 µL for purple tips. For each volume, two to four individual tips were tested in five replicates. The resulting data were analyzed using GraphPad Prism. Random error was expressed as the repeatability standard deviation across the measurements.

Microscopic surface analysis

To further assess the quality of pipette tips, the tip orifice, the narrowest part at the distal end of the tip through which liquid is aspirated and dispensed, was examined using light microscopy. An intact surface and stable geometry of the orifice are essential for ensuring high precision pipetting[26]. Microscopic images were taken of all pipette tips used in the study, including new and washed tips. For each washing

condition, one tip was randomly selected from the respective box and imaged using a Leica light microscope equipped with a 4× objective. In addition, one tip was intentionally scraped across a laboratory bench to serve as a negative example, demonstrating that even minor physical alterations can be detected under microscopic inspection.

Sealing rim fit evaluation

The sealing rim of a pipette tip is designed to establish a tight and secure connection with the pipette cone, which is essential for uninterrupted and reliable pipetting performance. Since the tightness of the seal is influenced by the force applied during tip attachment, the first step is to determine the average force typically exerted by laboratory personnel when picking up tips. For this purpose, seven randomly selected scientists were asked to pick up tips of different sizes from a tip rack placed on an analytical balance. The change in weight during tip pickup was used to estimate the applied force. Using the calculated average pickup force, a pipetting simulation routine was performed. In this routine, a tip was attached to the pipette using the calculated average force, a small volume of water was aspirated and dispensed and the tip was then returned to the rack. This cycle was repeated 25 times for five tips of each size and each condition (new and washed). Any instances of tip detachment (fall-offs) during the simulation were recorded. The results are expressed as an adhesion rate, defined as the percentage of cases in which the tip remained securely attached throughout all 25 repetitions. These values were analyzed using GraphPad Prism to assess whether repeated washing had a measurable impact on the mechanical fit and sealing performance of the tips.

2.2.3 Structural and functional integrity of microwell plates

To evaluate whether repeated washing affects the usability of microwell plates in cell-based assays, both cell adhesion and surface integrity were systematically assessed using Greiner 384-well plates and E-Plates under experimental conditions.

Cell adhesion and surface integrity of Greiner-384-microwell plates

To assess whether washing affected cell adhesion in normal cell culture plates, 500 HEK-293-WT and Colo-320-DM cells per well were seeded in 60 µL of medium into freshly unpacked, five times washed and 100x washed plates. The plates were incubated at 37 °C with 5 % CO_2 for five days. Daily brightfield images were acquired using the Automated Cell Culture (ACC) microscope at 4× magnification. Confluency was automatically calculated by the system and visualized using GraphPad Prism.

Surface and assay integrity of E-Plates

To determine whether previously used E-Plates retain functional integrity after cleaning, plates underwent three consecutive washing cycles prior to performance testing in an xCELLigence impedance-based assay. The pharmacological experiment was conducted by Frederik Bangnowski on the xCELLigence high-throughput platform. Initially, 20 µL of cell culture medium was dispensed into each well, followed by centrifugation and acquisition of background impedance values. Subsequently, cells were seeded at a density of 5,000 cells per well in 30 µL of medium. Plates were maintained at room temperature for one hour to promote cell attachment and then incubated for 48 hours at 37 °C in a humidified atmosphere with 5 % CO_2. After incubation, the medium was replaced with 40 µL of assay buffer (H_2O, 1× HBSS, 20 mM HEPES, pH 7.4), and plates were equilibrated for 40 minutes under identical conditions. Normalization measurements were recorded prior to compound addition. Test compounds, designed to interact with the expressed receptor, were then applied to each well, and impedance changes were monitored at 15-second intervals immediately following stimulation. Cell growth was tracked over 47 hours via the Cell Index and assay performance was quantified using the baseline-normalized Cell Index (bnCI) AUC. Data were analyzed using GraphPad Prism and compared to results obtained from new, unused E-Plates.

2.3 Cleanliness and washing effectiveness of TipNovus and Plate Novus

Ensuring the cleanliness of reusable laboratory consumables is a critical parameter for maintaining experimental integrity, particularly in sensitive analytical and cell-based assays. To systematically evaluate residual contamination following automated washing, three targeted assays were employed: a Bradford assay to detect protein residues, a Fluorescein assay for fluorescent contaminants, and a Neutral Red assay to assess dye retention and measured in a plate reader. Bradford, Fluorescein and Neutral Red tests were performed both on Greiner 384-multiwell plates and on Eppendorf epT.I.P.S. in three sizes (dark grey, yellow, blue).

Additionally, cell-based handling experiments were conducted to determine whether washed plates support sterile conditions and whether the PlateNovus system, in combination with standardized washing protocols, can reliably restore consumables for use in cellular assays.

2.3.1 Bradford protein assay

The Bradford assay quantifies protein concentration based on the color shift of Coomassie Brilliant Blue G-250 upon protein binding, measured at 595 nm[31]. To assess residual contamination on washed pipette tips and plates, bovine serum albumin (BSA) was used as a model protein and detected with Bradford reagent.

To assess residual protein contamination in reused pipette tips, epT.I.P.S. were subjected to defined treatments and subsequently tested using a Bradford assay. Tips were either new (blank control), BSA-contaminated without cleaning (assay), BSA-contaminated and washed using the TipNovus system (immediately or after 24 h desiccation), or BSA-contaminated and manually rinsed with deionized water. Contamination was introduced by repeatedly aspirating and dispensing a 40 mg/mL BSA solution, ensuring that the inner surfaces of the tips were uniformly coated with the protein. All tips were then used to dispense 200 µL of Bradford reagent into 96-well plates. Absorbance values were recorded, combined and analyzed in GraphPad Prism to quantify residual protein levels across conditions.

While the previous experiment focused on potential protein residues inside pipette tips after washing, the following approach addressed residual contamination in plates themselves. Initially, each well of 384-microwell plate was filled with 50 µL Bradford reagent and measured in a microwell plate reader to establish the blank signal. To account for potential background noise, measurements were performed twice in succession. Subsequently, the same wells were supplemented with 10 µL BSA (40 mg/mL), and absorbance was recorded again to determine the assay signal. After this step, plates were washed in the PlateNovus system according to the custom protocol, refilled with 50 µL Bradford reagent, and re-measured in the plate reader to evaluate cleanliness after washing. Four 384-multiwell plates were tested, and data was analyzed and visualized using GraphPad Prism.

2.3.2 Fluorescein residue test

Fluorescein sodium ($C_{20}H_{10}O_5Na_2$) is a water-soluble xanthene dye with strong fluorescence, absorbing at ~485 nm and emitting at ~520 nm[32]. Its intensity is pH-dependent, increasing under alkaline conditions. For this study, a 10 mM stock was prepared in 0.1 M NaOH, and a 100 µM working solution was buffered in 50 mM Tris (pH 7.5). Due to its high quantum yield and low toxicity, fluorescein sodium serves as an ideal marker for detecting residual contamination and evaluating cleaning efficacy[32].

Eppendorf epT.I.P.S. were used to investigate fluorescein residue after manual handling and subsequent cleaning. Initially, 200 µL of 50 mM Tris buffer (pH 7.5) were pipetted into wells of a 96 well plate using fresh tips of each type (blanks). To simulate contamination, the tips were repeatedly aspirated and dispensed with a 10 mM fluorescein sodium stock solution, ensuring the inner surfaces were coated with the compound. A subset of these contaminated tips was set aside for 24 hours to allow the dye to dry onto the tip surfaces. The remaining tips were immediately used to pipette 200 µL of fresh Tris buffer into new wells, representing the Assay condition. After initial measurements, both the freshly contaminated and the dried tips were cleaned using the TipNovus system. The washed tips were then reused to pipette 200 µL of Tris buffer and fluorescence was measured again to assess residual contamination. Data from all tip sizes were combined, analyzed, and visualized using GraphPad Prism.

To evaluate the washing efficiency of fluorescein sodium salt using the PlateNovus system, an initial blank measurement was performed. Each well of the plates was first filled with 50 µL of 50 mM Tris buffer (pH 7.5). The blank was measured twice to confirm consistency. After the blank readings, the buffer was removed, and 50 µL of a 100 µM fluorescein sodium working solution was added to the same plates (Assay). Subsequently, the plates were washed using the PlateNovus protocol and refilled with 50 µL of 50 mM Tris buffer (pH 7.5). A final measurement was performed in the plate reader to assess residual fluorescence after washing. Data was analyzed and visualized using GraphPad Prism.

2.3.3 Neutral Red residue test

Neutral Red (Toluylene Red) is a cationic dye used as a lysosomal viability marker and pH indicator, with absorbance at ~540 nm and fluorescence at ~640 nm[33]. For this study, a 20 mg/mL stock was prepared in DMSO and diluted to 50 µg/mL in DI water. Blanks contained diluted DMSO, to match the solvent background.

To assess Neutral Red residues persisted inside pipette tips after cleaning, Eppendorf epT.I.P.S. of different sizes were tested. As an initial control, 200 µL of diluted DMSO were dispensed into wells of a 96-well plate using unused tips (blank). For contamination, tips were exposed to a 20 mg/mL Neutral Red stock solution by repeated aspiration and dispensing, ensuring thorough contact with the inner surfaces. Some of these tips were stored under sterile conditions for 24 hours to allow the dye to dry, while the remaining contaminated tips were immediately used to transfer 200 µL of diluted DMSO into fresh wells, forming the assay condition. After the initial readings, both freshly contaminated and dried tips were subjected to cleaning in the TipNovus system. Post-wash, the same tips were reused to pipette the diluted DMSO and absorbance was measured again to determine any residual dye. All measurements were pooled across tip sizes and analyzed using GraphPad Prism.

The performance of the PlateNovus system in removing Neutral Red residues was assessed using 384-microwell plates. Initially, each well was filled with 50 µL of diluted DMSO and absorbance was recorded twice with a microplate reader to establish baseline values. Following baseline acquisition, the solvent was aspirated, and 50 µL of a 50 µg/mL Neutral Red working solution, prepared in DI water, was dispensed into the same wells. Subsequently, the plates underwent a cleaning cycle in the PlateNovus system and refilled with 50 µL of diluted DMSO. A final reading was taken to quantify any residual dye. All data were analyzed and graphically represented using GraphPad Prism.

2.3.4 Cell-based experiments

Cell-based assays were performed to assess cleanliness, as residual contamination could compromise experimental reliability. The evaluation included standard cell culture plates and E-Plates under different conditions.

GrenoClean efficiency

GrenoClean was tested for its ability to remove biological residues while preserving material integrity. Its efficacy was compared to commonly used laboratory disinfectant Incidin Active (1%).

HEK-293-WT cells were seeded at 3,000 cells per well in 40 µL of medium and incubated overnight at 37 °C and 5 % CO_2. Three conditions were tested: Medium (blank control), Incidin Active (1%), and GrenoClean. In each condition, 40 µL of reagent was added at 15-minute intervals, resulting in cumulative exposure times of 15, 30, 45, and 60 minutes. After the final exposure, 10 µL of CellTiter-Glo (CTG) reagent was added and incubated for 15 minutes. Luminescence was then measured using a plate reader. Higher ATP levels indicated greater cell viability.

Effectiveness of cell and compound removal in standard cell culture plates

Greiner 384-microwell plates were seeded with 500 HEK-293-WT cells per well in 60 µL of medium and incubated for five days at 37 °C in 5 % CO_2. After incubation, plates were washed using the TipNovus system, and fresh medium was added to each well. The plates were then incubated for additional five days under identical conditions. Confluency was assessed for all wells using automated imaging with the automated cell culture (ACC) system. Washed plates with medium were compared to new plates with medium.

To evaluate whether PlateNovus effectively removes cytotoxic compounds, a Plate Uniformity (PU) test was adapted. HEK-293-WT and Colo-320-DM cells were seeded at 500 cells per well in 60 µL of medium. HEK-293-WT cultures contained 1 % Penicillin–Streptomycin, while Colo-320-DM cultures were antibiotic-free. After 24 hours, three plates were prepared using an Echo 650 acoustic dispenser with defined layouts (**Fig. 7**): Low concentration (green): DMSO, indicating high viability, intermediate concentration (yellow): 12 nM for HEK-293-WT, 40 nM for Colo-320-DM and high concentration (red): 1 µM, indicating strong cytotoxicity. Plates were incubated and assessed using 10 µL CTG: Colo-320-DM after 3–4 days, HEK-293-WT after 5 days. Following initial measurement, plates were washed and reseeded at the same density, then reassessed after identical incubation periods. Reduced viability in previously high-concentration wells indicated residual compound contamination. Data were analyzed in GraphPad Prism, normalizing DMSO wells to 100 % viability.

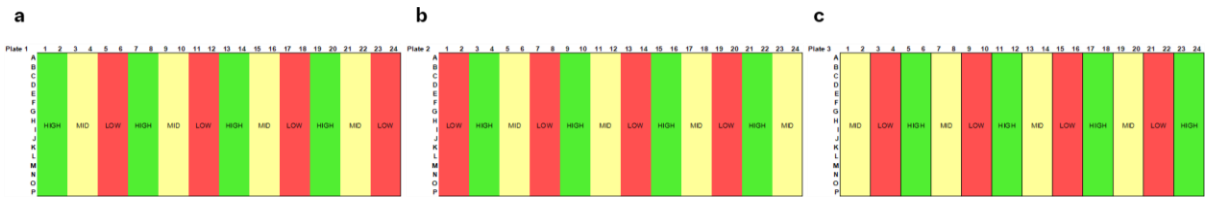

Fig. 7 | Plate Uniformity test layout.

a, Plate 1, **b,** Plate 2, **c,** Plate 3. Green wells indicate high cell viability (DMSO control), yellow wells represent mid toxic compound concentration (12 nM for HEK-293-WT, 40 nM for Colo-320-DM) and red wells correspond to the highest concentration (1 µM), resulting in strong cytotoxic effects.

Effectiveness of cell removal in E-Plates

To assess PlateNovus cleaning performance in E-Plates, two conditions were tested. In the first condition, 500 HEK-293-WT cells were seeded in 60 µL per well and cultured for five days and washed directly. In the second condition, an identical plate was cultured for five days, after which the medium was removed, and the plate was allowed to dry completely before washing. Following the respective washing procedures, both plates were stained with Alexa Fluor WGA-555, which binds N-acetylglucosamine residues on cell membranes to visualize potential cellular remnants. Imaging and quantitative analysis were performed by Frederik Bangnowski using an Opera Phenix high-content system, and results were compared to new plates and plates containing cells.

2.4 Impact of repeated washing on cell-based assay performance

To determine whether repeated washing of pipette tips and microplates affects cell-based assay outcomes, a proliferation experiment was performed under two conditions: new consumables and washed consumables. Washed items included Sartorius Biohit pipette tips (orange and purple) washed 12 times and Greiner 384-microwell plates washed three times.

Two cell lines, Colo-320-DM and LS 513, were used, and two cytotoxic compounds were applied to evaluate potential washing effects across a broad experimental setup. In both conditions, 500 cells per well were seeded in 60 µL of medium containing 1 % Penicillin–Streptomycin. Each plate was divided so that top rows were seeded using orange tips and bottom rows using purple tips. Plates were incubated overnight at 37 °C and 5 % CO_2 for cell attachment.

On the following day, defined concentrations of the two test compounds were dispensed using an Echo 620 acoustic liquid handler. Starting from the highest concentration (10 µM, in duplicates), serial 1:3 dilutions were applied across the plate, ending with DMSO-only wells as blanks. Each compound was applied to both tip conditions on the same plate. Plates were incubated for five days under standard conditions.

On day 5, 10 µL of CTG reagent was added to each well and incubated for 15 minutes before luminescence measurement. Dose–response curves for new versus washed consumables were generated and analyzed using GraphPad Prism.

3 Results

3.1 Acoustic and energy profiles of Grenova systems

Noise exposure and energy demand of TipNovus, PlateNovus, and TipLumis were measured during wash, dry, and self-clean cycles.

3.1.1 Noise levels

Noise exposure was recorded at three positions: near the device, at the far end of the room, and in an adjacent room. During the wash cycle, TipNovus exhibited pronounced fluctuations near the device, with peaks approaching 90 dBA during the Soak Low and Soak High phases (**Fig. *8***a). At the far end of the room, levels remained below 80 dBA, while in the adjacent room they were consistently below 50 dBA. PlateNovus displayed a different acoustic profile, with sustained levels between 70–80 dBA throughout the 50-minute cycle (**Fig. *8***b). Additional measurements during the dry and self-clean cycles of TipNovus revealed distinct patterns: the dry cycle produced periodic increases with peak values around 70–80 dBA (**Fig. *8***c), whereas the self-clean cycle generated a sharp rise reaching approximately 95 dBA (**Fig. *8***d). At distant positions, sound levels remained significantly lower in all cases.

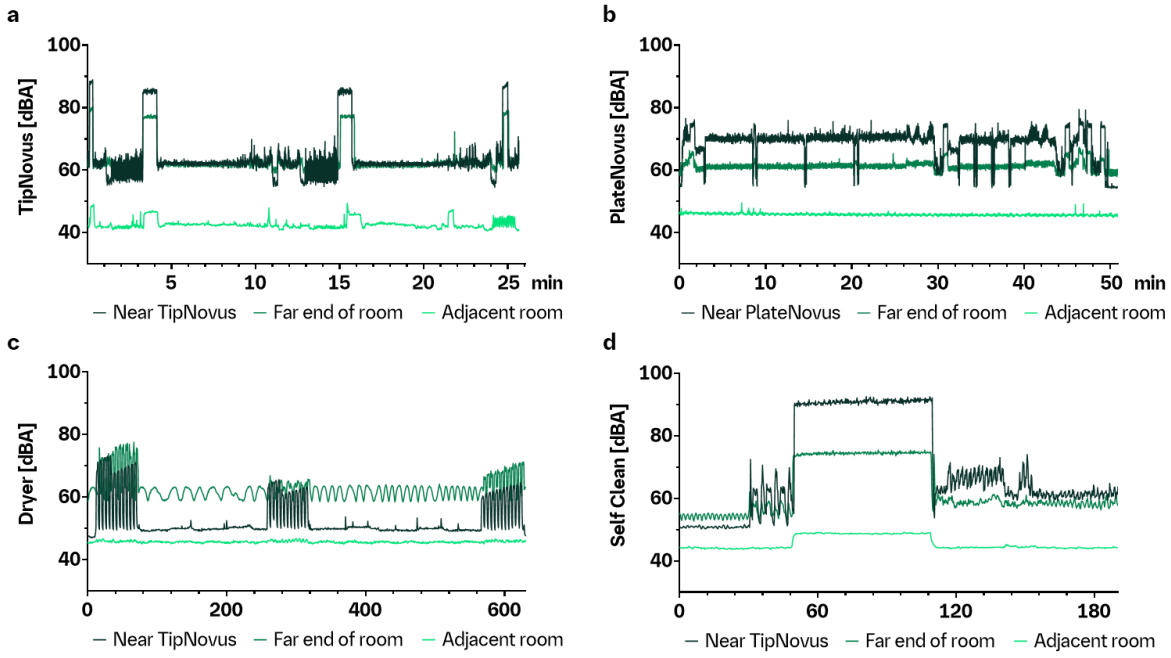

Fig. 8 | Noise level measurements during operational cycles of Grenova devices.

a, TipNovus wash cycle (≈25 min) with multiple steps shows pronounced peaks near the device, approaching 90 dBA during soaking phases, while distant positions remain below 80 dBA and 50 dBA. **b,** PlateNovus wash cycle (≈50 min) maintains 70–80 dBA near the device with minimal variation at other positions. **c,** TipNovus drying cycle (≈10 min at 60 °C) exhibits periodic increases up to 80 dBA near the device; other positions remain stable**. d,** TipNovus self-clean cycle (≈180 min) produces sustained high levels near the device, peaking around 95 dBA, while far-end and adjacent-room measurements stay below 60 dBA. All measurements represent single runs (n = 1).

3.1.2 Energy consumption

Energy demand was quantified for washing and drying processes. A single TipNovus wash cycle (25 minutes) required approximately 3,897 W (**Fig. *9***a), corresponding to 0.065 kWh, while PlateNovus consumed about 8,996 W during a 50-minute cycle (**Fig. *9***b), equating to 0.15 kWh. Drying processes showed higher cumulative energy use: TipNovus drying (10 minutes at 60 °C) resulted in an AUC of 5,237 W (**Fig. *9***c), with an initial peak near 800 W followed by a gradual decline, totaling 0.085 kWh. In contrast, TipLumis drying (3 hours at 40 °C) required approximately 105,776 W (**Fig. *9***d), characterized by an initial constant high-power phase (~800 W) and intermittent peaks during the remaining period, amounting to 1.76 kWh.

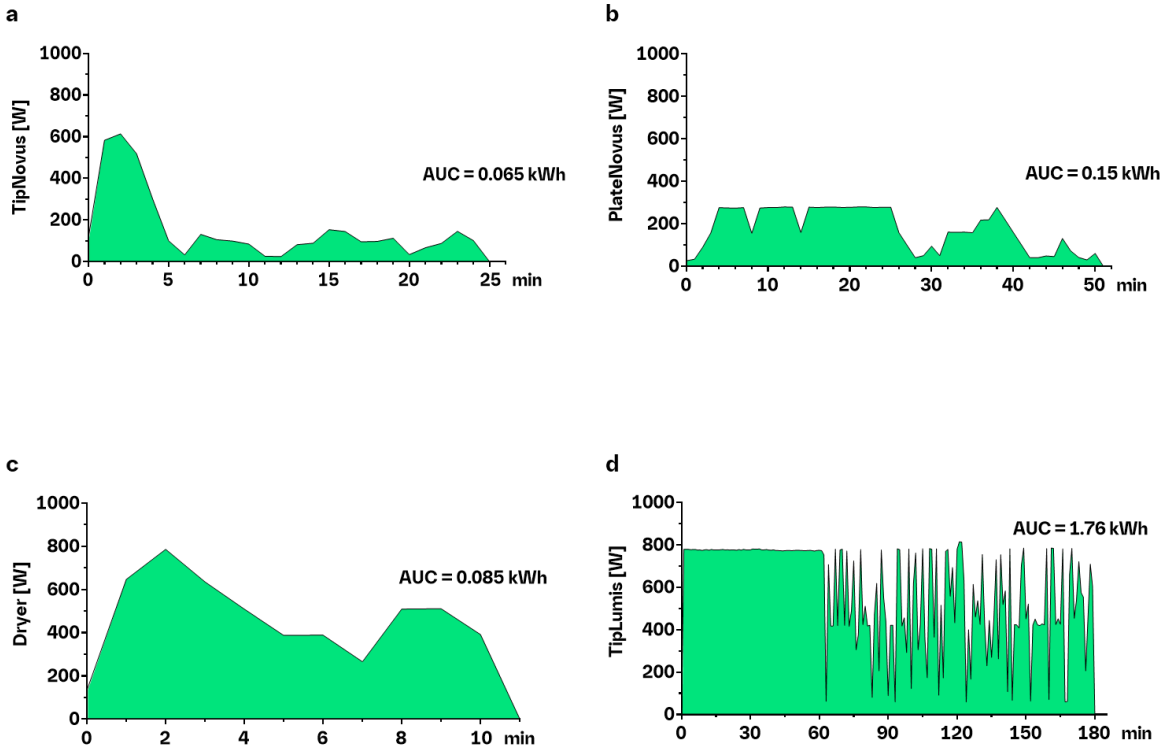

Fig. 9 | Energy consumption during washing and drying processes in of Grenova devices.

Power profiles were recorded at one-minute intervals using an energy logger. **a,** TipNovus wash cycle (≈25 min, n = 14) peaked near 800 W before declining (AUC = 3897 W = 0.065 kWh). **b,** PlateNovus wash cycle (≈50 min, n = 4) showed intermittent peaks with sustained mid-range power (AUC = 8996 W = 0.15 kWh). **c,** TipNovus drying cycle (≈10 min at 60 °C, n = 6) exhibited an initial peak (~800 W) followed by gradual decrease (AUC = 5237 W = 0.085 kWh). **d,** TipLumis drying cycle (≈180 min at 40 °C, n = 2) required the highest energy input, with constant high power and repeated peaks (AUC = 105,776 W = 1.76 kWh).

3.2 Quality assessment of laboratory articles

To ensure the reliability of reusable consumables, structural and functional properties of pipette tips and microwell plates were examined after multiple washing cycles. Analyses included gravimetric performance, tip orifice morphology, and plate-based cell culture and pharmacological testing.

3.2.1 Pipette tip performance and surface integrity

Gravimetric analysis of Eppendorf epT.I.P.S. (yellow, 2-200 µL, 20 µL nominal volume pipette) showed that all tested tips dispensed volumes within the ISO 8655-2:2002 tolerance limits for systematic error. Measurements at 20 µL and 200 µL were consistent across new and washed tips (n = 4 per group, five replicates each, **Fig. *10***a,b). Comparable results for other tip sizes and brands can be found in the Appendix. Microscopic examination of the tip orifice at 4× magnification revealed no visible structural changes or surface irregularities associated with repeated washing. The geometry of the orifice remained stable across all conditions. A negative example shows an interrupted surface resulting from inadequate handling of a pipette tip (**Fig. *10***e). The average force applied during tip pickup was approximately 3000 g (**Fig. *10***c). When tips were attached using this force, all tested tips maintained a high adhesion rate throughout 25 simulated pipetting cycles. No correlation between the number of washing cycles and tip detachment was observed (**Fig. *10***d).

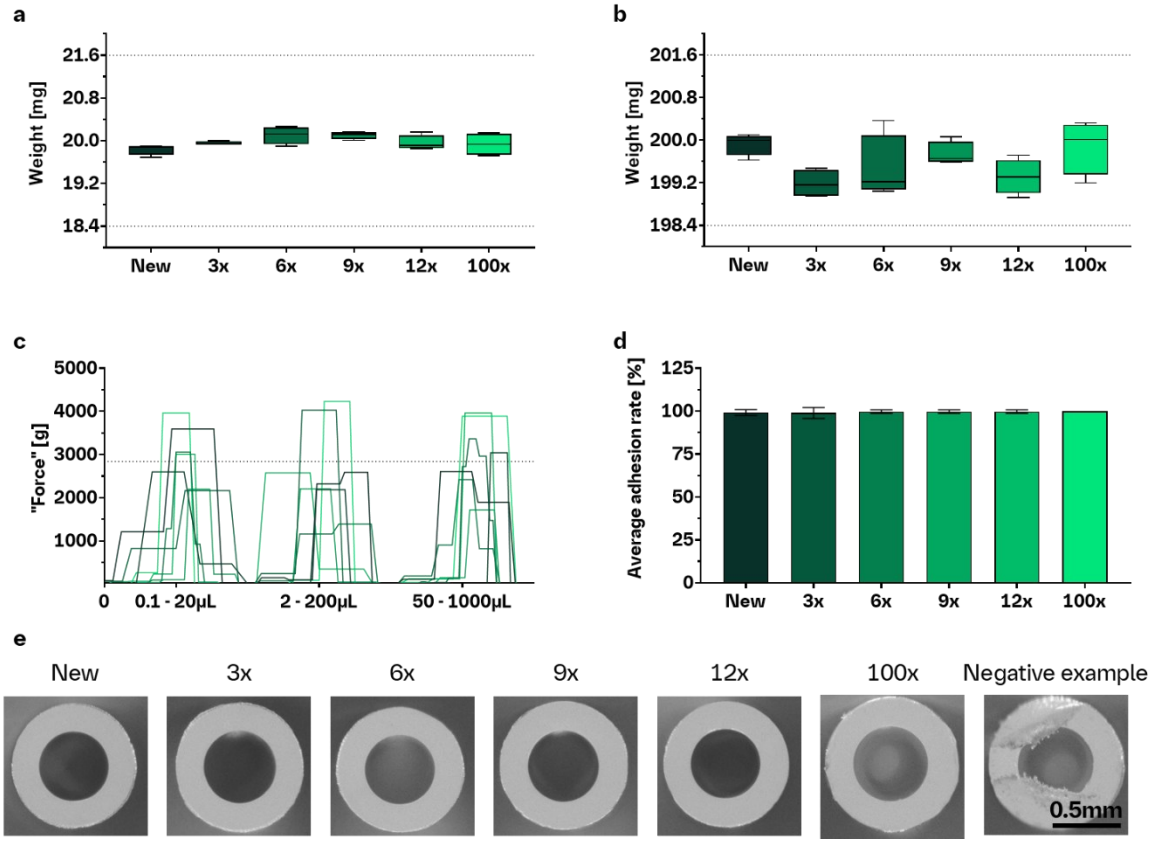

Fig. 10 | Structural and functional evaluation of epT.I.P.S. after repeated washing cycles.

a, b, Gravimetric measurements at target volumes of 20 µL (a) and 200 µL (b); n = 4 tips per group, five replicates each. Dotted lines indicate ISO 8655-2:2002 limits for systematic error. All values remained within permissible ranges across washing conditions. **c,** Force profiles for tip pickup at three volume ranges (0.1–20 µL, 2–200 µL, 50–1000 µL) show consistent performance, with an average force of ~3000 g; n = 7. **d,** Average adhesion rates during simulated pipetting (25 cycles) remained stable, with no trend toward increased detachment after repeated washing; n = 5 tips per group, 25 repetitions each **e,** Light microscopy images (4×) of tip orifices reveal intact geometry and smooth surfaces for all conditions; n = 1. A negative example illustrates damage from improper handling.

3.2.2 Microwell plate integrity and cell-based assays

To determine whether repeated washing and drying cycles compromise the usability of Greiner 384-well plates, cell growth and pharmacological responses were examined under three conditions: a new plate, a plate washed and dried five times, and a plate subjected to 100 simulated washing and drying cycles (**Fig. *11***). HEK-293-WT and Colo-320-DM cells were cultured for five days, and confluency was monitored daily. Confluency increased progressively from day one to day five across all conditions. Initial confluency was minimal, while day five displayed the highest values with overlapping error bars among groups. Microscopic images of the same wells confirm visible cell presence from day one and a pronounced increase in coverage between day three and day four, particularly for HEK-293-WT cells. The image rows correspond to the new plate (top), the five-times washed plate (middle), and the plate washed 100 times (bottom). Colo-320-DM cells exhibited lower overall confluency, reaching approximately 60% by day five, whereas HEK-293-WT cells achieved about 70% confluency despite appearing visually dense in the images.

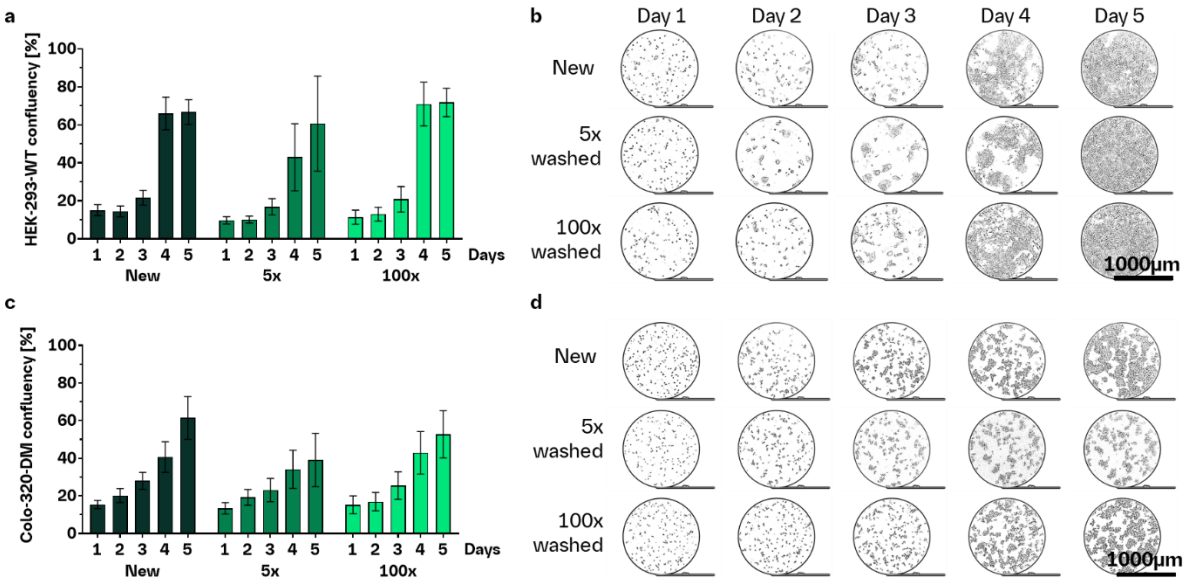

Fig. 11 | Growth performance of cells after repeated washing cycles.

a, Confluency of HEK-293-WT cells over five days in three plate conditions: new, washed five times, and subjected to 100 simulated washing/drying cycles. Cells were seeded at 500 cells per well in 60 µL medium and incubated at 37 °C with 5 % CO_2. Confluency was quantified using the ACC microscope (4× magnification). **b,** Representative microscopic images (4×) of HEK-293-WT cells from the same well across five days under the three conditions. **c**, Confluency of Colo-320-DM cells

under identical conditions, showing comparable growth trends. **d,** Microscopic images (4×) of Colo-320-DM cells across five days for the same plate conditions.

Pharmacological assays were conducted to evaluate E-Plates after repeated washing in the PlateNovus system. Cells expressing a ligand-responsive receptor were seeded, and impedance-based Cell Index values were monitored over 47 h (**Fig. *12***a). Growth curves were generated for washed plates, starting near zero and increasing to >10 over the observation period. For functional assessment, the AUC was calculated across a range of compound concentrations and compared between new and washed plates (**Fig. *12***b). Dose-response curves were fitted to determine EC_{50} values: 6.59 nM for new plates and 10.21 nM for washed plates (**Fig. *12***c).

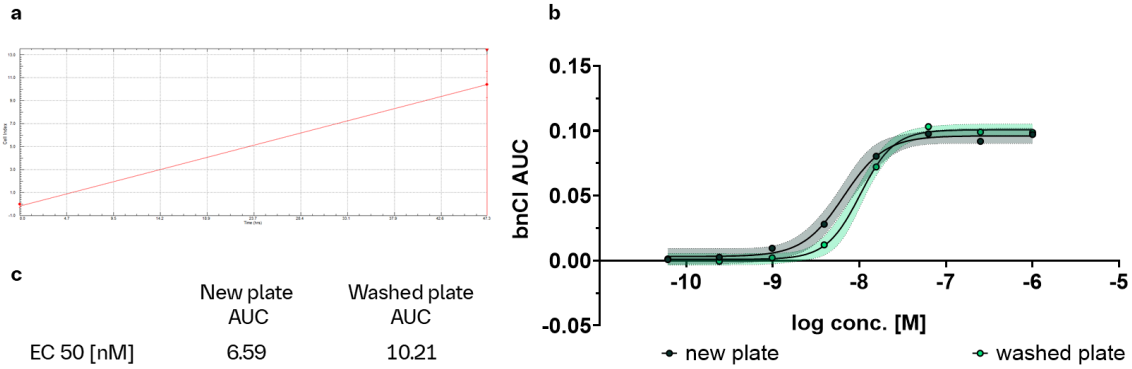

Fig. 12 | Pharmacological assay performance on E-Plates after repeated washing.

a, Cell Index trajectory over 47 h for cells seeded on washed plates. **b,** Dose-response curves based on AUC values across compound concentrations for new (dark green) and washed plates (light green). **c,** Calculated EC_{50} values for AUC: new plate = 6.59 nM; washed plate = 10.21 nM.

3.3 Evaluation of cleaning performance

To assess washing efficiency, residual contamination was quantified in pipette tips and 384-well plates using Bradford, Fluorescein, and Neutral Red assays under defined experimental conditions (**Fig. *13***).

3.3.1 Validation of TipNovus cleaning through biochemical assays

For pipette tips (**Fig. *13***), Bradford assay absorbance at OD_{595} nm was similar for cleaned tips (n = 38), blank tips (n = 30) and dried tips (n = 18). Rinsed tips (n = 23) exhibited significantly higher absorbance than blank, cleaned and dried tips. Assay controls (n = 22) displayed the highest OD_{595} values (**Fig. *13***a).

Fluorescein fluorescence measurements showed similar values for cleaned tips (n = 19), blanks (n = 22) and dried tips (n = 21). Assay controls (n = 21) showed the highest fluorescence (**Fig. *13***b).

Neutral Red absorbance at OD_{540} nm was comparable for blank tips (n = 22), cleaned tips (n = 20) and dried tips (n = 21). Assay controls (n = 19) exhibited the highest absorbance (**Fig. *13***c).

Pipette tips in three volume ranges (0.1–10 µL, 2–200 µL, and 50–1000 µL) were used. All datasets were tested for normality using the Shapiro-Wilk test and analyzed for statistical significance with one-way ANOVA followed by Tukey's multiple comparisons test. Significance levels are indicated by asterisks starting at $p < 0.05$.

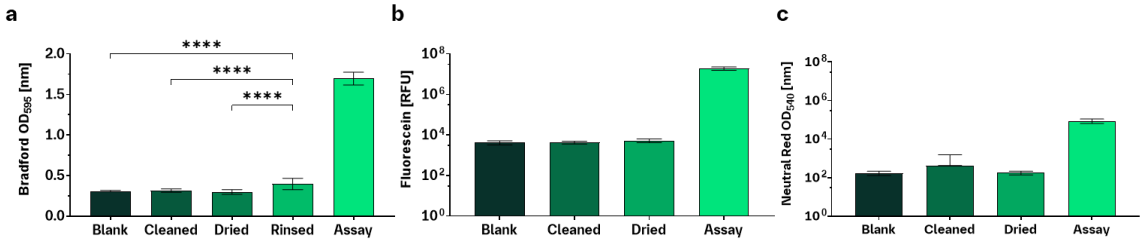

Fig. 13 | Residual contamination analysis of pipette tips.

a, Bradford assay absorbance at OD_{595} nm for blank (n = 30), cleaned (n = 38), dried (n = 18), rinsed (n = 23), and assay controls (n = 22). **b,** Fluorescein fluorescence (RFU) for blank (n = 22), cleaned (n = 19), dried (n = 21), and assay controls (n = 21). **c,** Neutral Red absorbance at OD_{540} nm for blank (n = 22), cleaned (n = 20), dried (n = 21), and assay controls (n = 19). Bars represent mean values with standard deviation. Pipette tips were tested in three volume ranges (0.1–10 µL, 2–200 µL, and 50–1000 µL). Statistical significance was assessed by one-way ANOVA followed by Tukey's multiple comparisons test. Significance levels are indicated by asterisks starting at $p < 0.05$.

3.3.2 Validation of PlateNovus cleaning through biochemical assays

Bradford, Fluorescein, and Neutral Red measurements were also performed on 384-well plates (**Fig. *14***) under blank, cleaned, and assay conditions (n=4). For all three assays, absorbance or fluorescence values for cleaned plates were lower than assay values. Significant differences were observed between the two consecutively measured blanks as well as between blank and cleaned plates.

All datasets were tested for normality using the Shapiro–Wilk test and did not meet normality assumptions. Consequently, statistical analysis was conducted using the Kruskal-Wallis test followed by Dunn's multiple comparisons test. Significance levels are indicated by asterisks starting at $p < 0.05$.

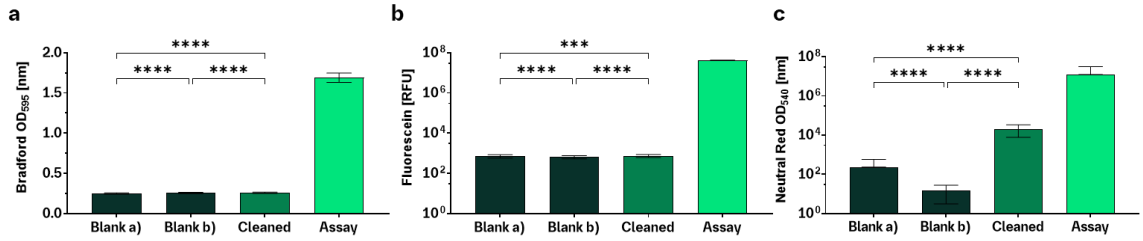

Fig. 14 | Residual contamination analysis of 384-well plates.

a, Bradford assay absorbance at OD_{595} nm, **b,** Fluorescein fluorescence (RFU) and **c,** Neutral Red absorbance at OD_{540} nm for blank a, blank b, cleaned plates, and assay controls (n = 4 per condition). Bars represent mean values with standard deviation. Significant differences were observed between the two consecutively measured blanks and between blanks and cleaned plates. All datasets were tested for normality using the Shapiro-Wilk test and did not meet normality assumptions. Statistical analysis was conducted using the Kruskal–Wallis test followed by Dunn's multiple comparisons test. Significance levels are indicated by asterisks starting at $p < 0.05$.

3.3.3 Validation of PlateNovus cleaning through cell based experiments

Antimicrobial efficacy of GrenoClean

The antimicrobial efficacy of GrenoClean and Incidin was assessed using a HEK-293-WT cell viability assay (**Fig. *15***a). Both reagents reduced viability by >90% within 15 min. GrenoClean showed a steeper decline, reaching >99% cell death after 30 min, whereas Incidin remained above this level even after 60 min. Microscopic images illustrate morphological changes over time: GrenoClean-treated cells aggregated with slight color loss, while Incidin-treated cells rapidly lost color and formed dense clusters (**Fig. *15***b).

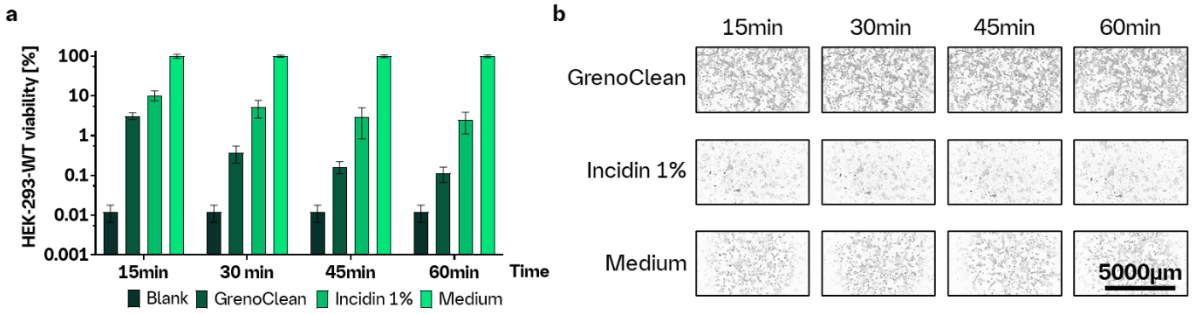

Fig. 15 | Effect of GrenoClean and Incidin on HEK-293-WT cell viability.

a, Viability [%] after exposure to GrenoClean and Incidin (1%) for 15–60 min compared to medium control. GrenoClean reduced viability by >99% within 30 min. Incidin remained above this level after 60 min. **b,** Representative images show morphological changes over time.

Cell removing efficiency in standard and E-Plates

To further assess the washing efficiency in microwell plates, cell-based assays were conducted. Confluency measurements in standard Greiner 384-well plates (**Fig. *16***a) were performed after five days incubation. Previously full and then washed plates show same confluency values as new plates.

Cell counts per image in E-plates (**Fig. *16***b) were quantified for immediately washed plates and dried plates and compared to new plates. New plates with medium showed the lowest cell counts. Immediately washed wells exhibited comparable counts as new plates. Dried plates show high cell

count values and full plates displayed the highest counts. Representative Opera Phenix images are shown alongside the graphs. Stained cells are illustrated by yellow dots.

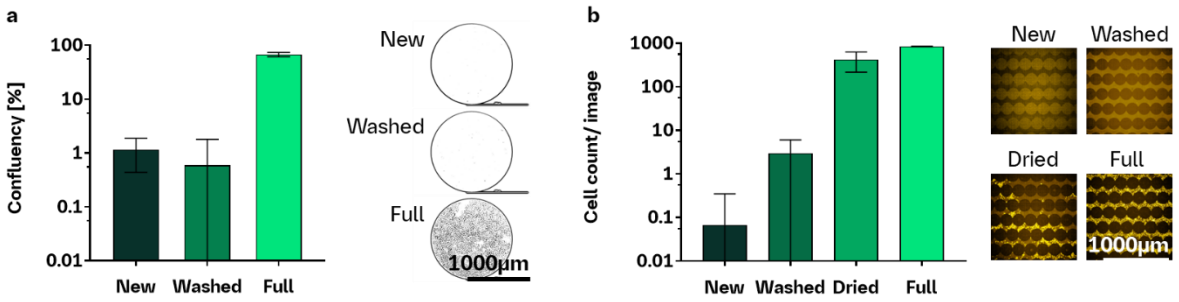

Fig. 16 | Evaluation of washing efficiency in microwell plates.

a, Confluency [%] in Greiner 384-well plates for new wells with medium, washed wells with medium and full plates after five days of incubation. Representative microscopic images are shown on the right. **b,** Cell count per image in E-plates for new plates with medium, washed wells with medium, dried and washed wells with medium and full plates. Representative Opera Phenix images are shown on the right. Bars represent mean values with standard deviation.

Compound removing efficiency

Viability curves for HEK-293-WT and Colo-320-DM cells showed reduced viability in wells exposed to 1 µM and mid-range compound concentrations compared to blanks (**Fig. *17***). After washing, viability patterns across all wells, including those previously exposed to 1 µM, were comparable to blanks, with no trend indicating impaired growth. Similar results were observed for Colo-320-DM, where washed plates showed uniform viability across all wells regardless of prior compound exposure.

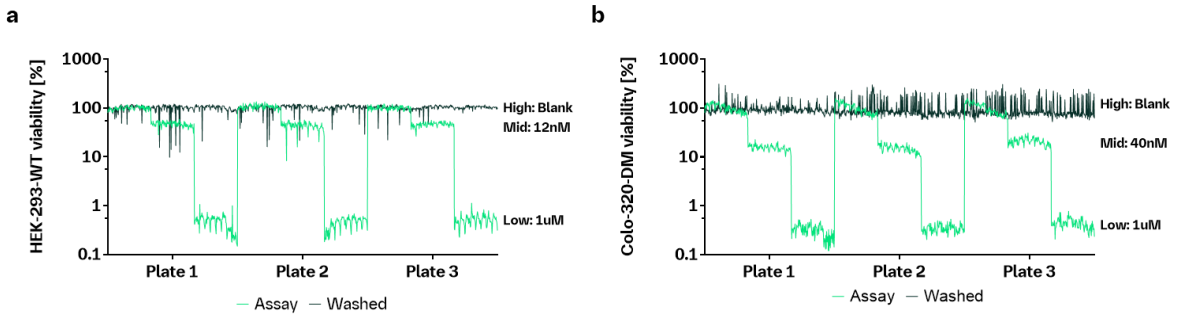

Fig. 17 | Plate Uniformity assay comparing viability in initial and washed plates.

a, Plate uniformity tests with HEK-293-WT and **b,** Colo-320-DM cells, showing cell viability (%) across wells. Initial assay (light green) and washed plates (dark green) are compared. Washed plates show no viability shift, even in wells previously exposed to 1 µM compound.

3.4 Comparison of dose-response-curves for new and washed consumables

To evaluate whether washing consumables affects compound potency or assay performance, dose-response curves were generated for Colo-320-DM and LS513 cells exposed to Compounds A and B using new and washed tips and plates (**Fig. *18***).

For Colo-320-DM, mean IC_{50} values for Compound A were 25.14 nmol/L (new) and 25.93 nmol/L (washed), while Compound B showed 207.87 nmol/L (new) and 263.00 nmol/L (washed). For LS513, mean IC_{50} values for Compound A were 36.92 nmol/L (new) and 31.64 nmol/L (washed), and for Compound B 214.57 nmol/L (new) and 296.39 nmol/L (washed). Curve fitting was generally robust, with most R^2 values ≥0.94. Exceptions were observed for LS513 Compound B, where R^2 was 0.8846 (new) and 0.9132 (washed). Top and bottom response values remained comparable between new and washed conditions (Appendix, **Table *2***). Curves for new and washed consumables show similar profiles across all compounds and cell lines. Shaded areas represent 95% confidence intervals

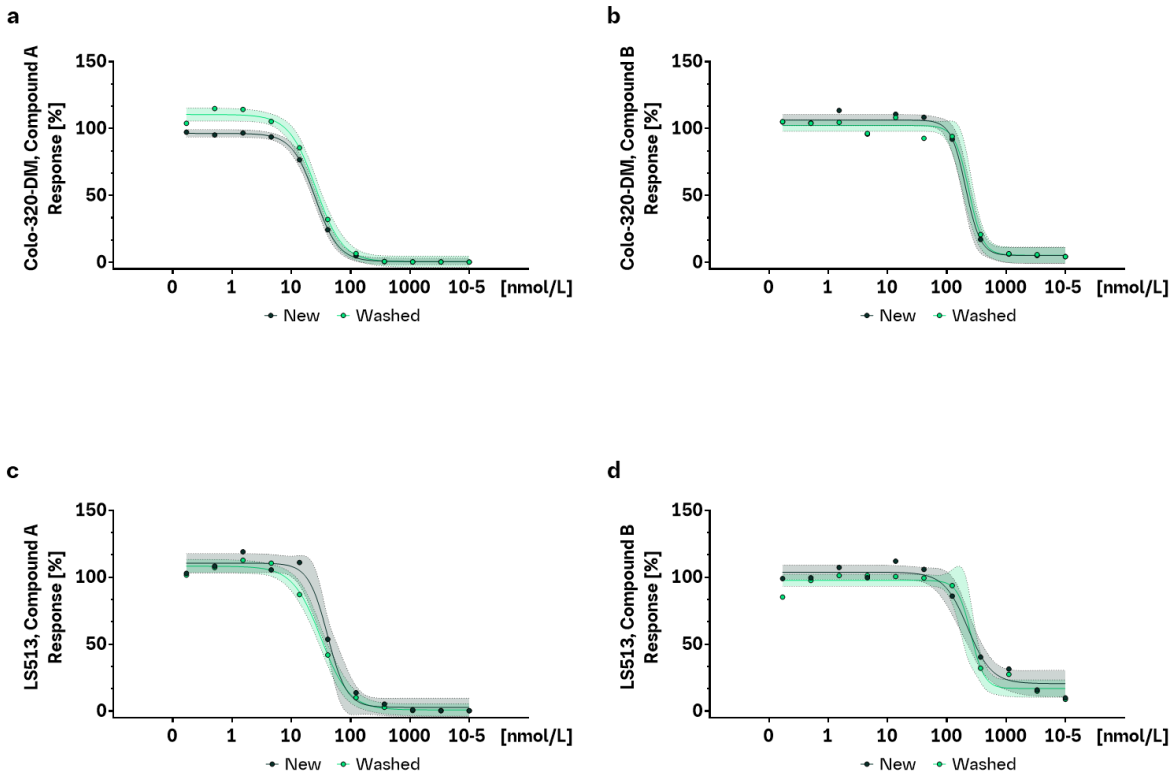

Fig. 18 | Comparison of dose-response curves for new and washed consumables.

a–b, Colo-320-DM response to Compound A and B using new (dark green) and washed (light green) tips and plates. **c–d,** LS513 response to Compound A and B under the same conditions. Curves for new and washed consumables show similar profiles across all compounds and cell lines. Shaded areas represent 95% confidence intervals.

4. Discussion

This study aimed to determine whether automated washing systems (TipNovus and PlateNovus) can enable the reuse of polypropylene pipette tips and polystyrene microplates, including impedance-based E-Plates, without compromising sterility, cleanliness, or data integrity. The evaluation included gravimetric accuracy tests, microscopic inspection, sealing-rim adhesion simulations, and multiple cell-based assays. Washed tips dispensed volumes within ISO 8655 limits and retained intact orifice morphology and stable attachment. Washed Greiner 384-well plates supported cell growth comparable to new plates, and E-Plates delivered pharmacological responses in the same potency range as unused controls. Cleanliness assays (Bradford, Fluorescein, Neutral Red) showed that residual contamination after washing approached blank levels, and plate-uniformity tests confirmed the absence of cytotoxic carry-over. Operational analysis revealed distinct noise profiles and notable energy demand. Microbiological monitoring identified bioburden risks inside washers, which were mitigated by scheduled decontamination. Overall, these findings demonstrate the feasibility of controlled reuse under defined quality standards and hygiene protocols.

4.1 Data integrity of reusable pipette tips

Three independent analyses support the same conclusion. Gravimetric measurements across all tip sizes remained within ISO 8655 limits after up to 12 washing cycles, confirming accuracy. Light microscopy showed intact orifice geometry in washed tips, while a deliberately damaged control demonstrated the sensitivity of this inspection to surface defects. After 100 simulated washing cycles, slight deformations at the tip orifice were observed, these changes are attributable to mechanical stress during washing. Adhesion tests using empirically measured pickup forces indicated stable retention without increased detachment after repeated cycles. These findings suggest that the washing and drying protocol in the TipNovus did not cause deformation of the sealing rim or orifice relevant to pipetting performance. Minor structural changes below the resolution of light microscopy cannot be excluded, but the consistency of accuracy, geometry, and mechanical fit indicates that such changes did not affect liquid handling (**Fig. *10***).

4.2 Reuse of microplates, including E-Plates

Plasma-treated Greiner 384-well plates supported consistent 5-day proliferation of HEK-293-WT and Colo-320-DM cells, even after an accelerated “100-cycle” washing simulation. This indicates that the tissue culture treatment, achieved through plasma modification, tolerated the washing and drying sequence applied in this study. Plasma treatment introduces charged functional groups and increases surface hydrophilicity, which promotes cell adhesion. While repeated washing and drying could theoretically reduce these effects, by altering surface energy or through adsorption of residues, the observed stability suggests that any changes were minimal under the tested conditions. Nevertheless,

more aggressive or prolonged cleaning cycles beyond those simulated here might pose a risk of modifying surface properties over time (**Fig. *11***).

For E-Plates, impedance-based growth curves showed a continuous increase in Cell Index over 47 hours, indicating that cells adhered and proliferated on the gold electrodes even after three washing cycles. Dose-response analysis confirmed functional assay performance: normalized Cell Index AUC values and fitted curves for washed plates were below two-fold derivation of those obtained with new plates. EC_{50} values shifted moderately (6.6 nM for new plates vs. 10.2 nM for washed plates), a difference that remains operationally acceptable. This variation may reflect minor changes in electrode surface conditioning, fluidic history, or biological variability rather than structural damage. Importantly, no qualitative evidence of electrode impairment or signal instability was observed (**Fig. *12***).

4.3 Cleanliness, sterility, and chemical carry-over

Washed pipette tips and plates showed contamination levels close to blank controls in all three assays: Bradford (protein), fluorescein (fluorescent dye), and Neutral Red (cationic dye). Deliberately contaminated samples produced the highest signals, confirming assay sensitivity. Immediately washed and dried tips were effectively cleaned by the TipNovus cycle, reducing values to near-blank levels. In contrast, tips rinsed only with water exhibited significantly higher signals, indicating that manual rinsing is insufficient compared to automated washing (**Fig. *13***). Similar trends were observed for plates, where cleaned samples consistently showed lower absorbance or fluorescence than assay controls. The statistical difference between blank and cleaned conditions can be explained by the very high number of wells tested (four 384-well plates = 1536 wells) and is supported by the significant difference between two consecutive blank measurements. These findings suggest that background variability, rather than residual contamination, accounts for the observed differences (**Fig. *14***).

GrenoClean demonstrated strong cytotoxic activity, outperforming Incidin in the conducted assay. The highest cleaning efficiency was achieved after a 30-minute soak, eliminating more than 99 % of viable cells (**Fig. *15***). This property was confirmed when assessing whether PlateNovus effectively removes cells from wells. After five days of incubation in previously seeded and washed wells, no cell growth was detected, indicating that plates were as clean as new after washing. Both standard cell culture plates and E-Plates showed complete removal of cells, confirming high cleaning efficiency (**Fig. *16***). However, cells that had dried inside wells could not be removed sufficiently, highlighting the need to keep medium in the wells until washing. To improve cleaning in 384-well plates, adjustments such as using a dedicated 384-well manifold (instead of the repurposed 96-well manifold) or reducing plate height to increase washing pressure were considered. These modifications require further evaluation, as higher pressure may deform wells. Extended sonication was also tested but did not improve cleaning results (**Fig. *16***).

In addition to cell removal, compound carry-over was assessed using plate-uniformity tests. Wells previously exposed to high cytotoxic compound concentrations did not impair cell growth after washing, indicating that chemical residues were effectively eliminated (**Fig. *17***).

4.4 Impact of washing on assay accuracy performance

Dose–response analyses using Colo-320-DM and LS513 cells demonstrated that washing consumables (pipette tips washed 12×, plates washed 3×) did not substantially alter assay outcomes. IC_{50} values for both compounds remained within the same order of magnitude when comparing new and washed conditions. For Colo-320-DM, Compound A showed 25.14 nmol/L (new) versus 25.93 nmol/L (washed), and Compound B 207.87 nmol/L versus 263.00 nmol/L. For LS513, Compound A shifted from 36.92 nmol/L to 31.64 nmol/L, and Compound B from 214.57 nmol/L to 296.39 nmol/L. These differences represent less than a two-fold deviation, which is generally considered acceptable in high-throughput screening contexts. Curve fitting was robust, with R^2 values mostly ≥0.94, indicating consistent data quality across conditions. Top and bottom response plateaus were comparable, suggesting that washing did not affect maximal or minimal signal levels. The slight variability observed, particularly for LS513 with Compound B, likely reflects biological variation rather than systematic bias introduced by washing. Overall, these findings support the conclusion that repeated washing of tips and plates does not compromise pharmacological assay integrity

4.5 Operational footprint: noise and energy as adoption constraints

Noise measurements revealed short peaks of approximately 90–95 dBA during specific TipNovus phases and sustained levels of 70–80 dBA for PlateNovus wash cycles. Sound levels decreased rapidly with distance from the devices. From an ergonomic perspective, this supports locating washers in a separate room or applying time-window policies and acoustic shielding.

Energy analysis showed that extended drying in TipLumis (≈3 h at 40 °C) accounted for the largest share of total energy consumption compared to individual wash cycles. Potential optimization strategies include reducing or batching drying cycles, lowering temperature or duration. These operational considerations must be weighed against the sustainability benefits. Even with current energy demand, washing significantly reduces cradle-to-grave CO_2 emissions compared to single-use consumables for both tips and plates.

4.6 Sustainability and cost savings through consumable reuse

Washing and reusing pipette tips results in a measurable reduction in CO_2 emissions compared to ordering and incinerating new consumables. For example, a single tip box (96 tips) generates approximately 281 g CO_2e[34] when produced and disposed of via incineration, whereas washing including drying requires only 6.7 g CO_2e, representing a reduction of about 98%. For 384-well plates,

the difference is smaller but still significant: washing accounts for 83 g CO_2e, compared to 239 g CO_2e for production and incineration which represents a reduction of nearly 65%. At the assumed washing frequency (TipNovus: 1,040 cycles/year; PlateNovus: 624 cycles/year) the annual CO_2 reduction represents around 1.53 metric tons (1.14 t from tips and 0.39 t from plates), reinforcing the benefit of sustainability.

On a global scale, the potential impact of reuse strategies is substantial. The global carbon footprint associated with the production and disposal of single-use pipette tips is estimated at approximately 7.7 million metric tons of CO_2e annually. If only 1% of these tips were washed and reused one time instead of being replaced, the resulting save would amount to roughly 75000 metric tons of CO_2e. This represents more than 68000 flights from Vienna to New York. This underscores the significant mitigation potential of implementing reuse strategies in laboratory workflows. Furthermore, combining washing with subsequent recycling could amplify these benefits, extending the lifecycle of consumables while minimizing environmental impact.

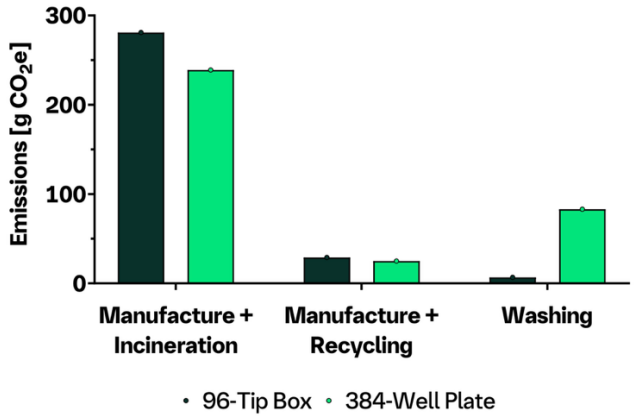

Fig. 19 | Carbon footprint of consumables under different end-of-life strategies.

In addition to the substantial reduction in CO_2e emissions, washing and reusing consumables provides significant cost advantages. Based on the operational parameters (Appendix, **Table *3***), the total cost per wash cycle, including water, electricity, reagents, and labor, amounts to €9.93 for TipNovus and €10.38 for PlateNovus. This translates into a wash cost per item of €2.48 for a tip box and €2.60 for a 384-well plate, compared to purchase prices of €15 and €7 respectively. Consequently, each washed tip box saves approximately €12.52, and each washed plate saves €4.40 relative to single-use procurement.

At the assumed washing frequency (TipNovus: 1,040 cycles/year; PlateNovus: 624 cycles/year), the annual financial savings reach €52,070 for tips and €10,993 for plates, totaling €63,063 per year. When compared to the combined capital investment for both washing systems (€200,000), the return on investment (ROI) is highly favorable: the payback period is approximately 3.2 years. This demonstrates

that implementing washing workflows is not only environmentally responsible but also economically viable within a relatively short timeframe.

Overall, these findings highlight that reuse strategies deliver measurable reductions in both carbon footprint and operational costs. Scaling such approaches across laboratory networks could amplify these benefits, reducing global resource consumption while improving cost-effectiveness in routine workflows.

4.7 People, adoption, and risk mitigation

An internal survey identified strong support for re-use after successful validation and salient concerns, notably additional time, reliability and safety, tracking burden, space, and noise. The chosen operating model should directly address these points: centralized or automated models reduce user time and variability, locating devices near autoclaves reduces handling time and quiet hours or shielding mitigate noise. A short, competency-based training and certification for users should precede roll-out. Clear role assignments and escalation paths complete the management system.

4.8 Conclusion

Within the boundaries tested here, automated washing and controlled storage supported the reuse of pipette tips and microplates without detectable degradation of core quality attributes or assay outcomes, while offering meaningful reductions in plastic consumption and associated emissions. Successful adoption, however, depends on coupling validated wash cycles with robust hygiene management, traceability, and pragmatic operational controls around noise and energy. Implemented as a quality-assured workflow with clear release criteria, reuse of laboratory plastics can align scientific integrity with sustainability goals in high-throughput environments.

Implementation concepts for re-use of pipette tips and microwell plates

This implementation concept translates pilot findings on washing and re-use of pipette tips and microplates into an operational model that can be adopted within a regulated research environment. The framework focuses on three core elements:

I. enabling infrastructure,
II. robust workflows (centralized, decentralized, automated), and
III. quality and hygiene controls that collectively safeguard scientific integrity.

1. Infrastructure and prerequisites

Successful implementation requires a dedicated washing area in proximity to autoclaves to streamline decontamination and re-release of consumables. The room must provide DI-water supply, sewer connection, and sufficient electrical capacity, as well as storage for chemical canisters. Device footprints and weights inform space planning (TipNovus ≈ 750 × 775 × 470 mm, 52 kg; PlateNovus ≈ 730 × 540 × 725 mm, 82 kg; TipLumis ≈ 760 × 620 × 810 mm, 50 kg). Noise and vibration should be considered in the HSE assessment and may justify a separated “wash room”.

2. Workflow design options

Three operating models were evaluated.

Option 1: Centralized washing

Consumables are collected daily, scanned, washed centrally, dried and autoclaved (tips) as required, and redistributed to laboratories. This model concentrates expertise and fosters process consistency and traceability. It reduces training needs and minimizes user burden at the bench. However, it introduces internal logistics, creates a lag between use and availability, and depends on operator availability and backup plans.

Option 2: Decentralized washing

Users bring their used racks or plates to the wash room immediately after assays, perform barcode scans and wash cycles, and return material. This shortens turnaround time and avoids transport queues, but requires broad training, behavioral change and carries a higher risk of procedural variability. It also increases user workload.

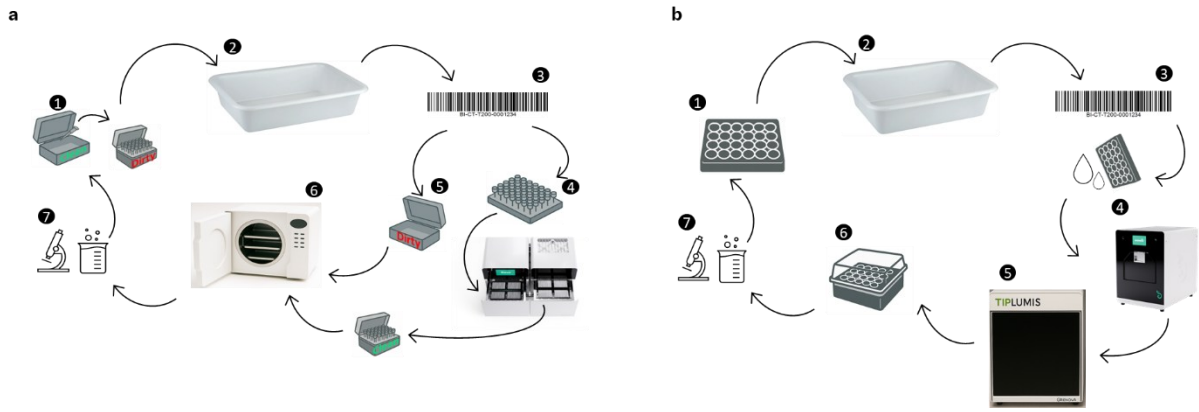

Fig. 20 | Workflow illustration of centralized and decentralized washing processes.

a, (1) tips are placed from clean (green) and to dirty (red) boxes; (2) full boxes are collected in a transport tray; (3) items are scanned for barcode-based tracking; (4) used tips are transferred to the washing device (TipNovus); cleaned tips are returned to the clean box; (5) dirty box is wiped with ethanol; (6) both boxes are autoclaved; (7) tips are redistributed for reuse in experiments. **b,** (1) plates are placed in a transport tray after use; (2) trays are collected; (3) plates are scanned and emptied; (4) plates are washed in PlateNovus; (5) dried overnight in TipLumis; (6) plates are covered with sterile lid; (7) plates are reused in subsequent assays. Icons were created using Copilot.

Option 3: Automation

Full or partial robotic handling standardizes loading, cycle initiation, and documentation, and minimizes hands-on time. It offers highest throughput and the best integration with digital tracking. The trade-offs are high capital cost, increased validation complexity, and format rigidity (compatibility constraints), as well as footprint requirements on the automation deck. A phased approach, starting centrally and progressing toward automation once volumes and SOPs are stabilized, is recommended. Currently, automation is only available for TipNovus.

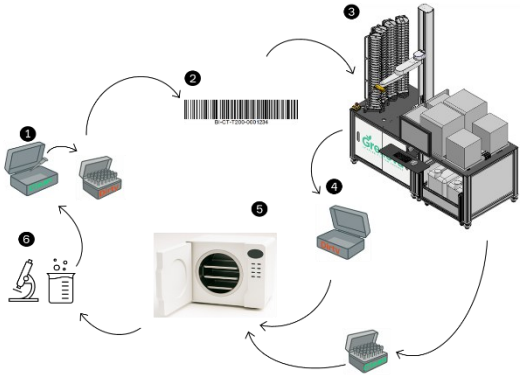

Fig. 21 | Automated washing workflow for pipette tips.

(1) Deposit used tips in “dirty” box; (2) Barcode scan for tracking; (3) Robotic loading and wash cycle; (4) Return to clean box and wipe dirty box with ethanol; (5) Autoclave; (6) Laboratory return. Icons were created using Copilot.

Across all options, tip flows follow the same logic: tips are transferred from Box A (clean) to Box B (dirty). Box A and B are collected, tips from Box B are washed, dried, and stored back in Box A after cleaning. Box B is wiped with ethanol, and both boxes are autoclaved if required before returning to laboratories. Plates are collected filled with liquid; liquid is removed immediately before transferring plates into PlateNovus. After drying in TipLumis, plates are equipped with a new cover and returned to laboratories.

To ensure lot-level and cycle-level traceability, a minimal barcode scheme is proposed:

BI-CT-{TYPE}{SIZE}-{UNIQUEID}

where {TYPE} denotes item class (T = tip, P = plate), {SIZE} defines the format (e.g., 200, 1000, 384), and {UNIQUEID} is a seven-digit counter serving as the primary key in the database. The barcode persists across multiple uses. The cycle counter is maintained digitally and incremented at scan events (collection, wash start, wash end/release, decontamination). Color coding on the physical boxes (e.g., Box A = green, Box B = red) complements electronic tracking and reduces handling errors.

3. Quality and hygiene controls

Implementation is contingent upon evidence that washed items perform equivalently to new items within defined use limits. Validation data demonstrate:

1. Mechanical integrity of tips after repeated washing: pipetted volumes remained within acceptance windows over multiple cycles (≥12 washes), and tips maintained stable connection to pipettes, supporting the absence of deformation relevant to accuracy and precision.
2. Assay performance on plates: cell confluence and pharmacological readouts on washed plates (including E-Plates with sensitive gold electrodes) were indistinguishable from new controls across time courses and dose-response curves, indicating that surface properties and electrode function were preserved even after up to five washes.
3. Cleanliness verification: protein (Bradford), fluorescein, and neutral red removal through TipNovus and PlateNovus and microscopy-based confirmation that cells are reliably cleared by the PlateNovus cycle.

Maintaining sterility and operational reliability of washing systems requires a structured hygiene protocol. For contamination-sensitive workflows such as cell culture, decontamination of TipNovus and PlateNovus using a 2% sodium hypochlorite solution is recommended every three weeks. For less critical applications, intervals of eight to twelve weeks may be sufficient. Preventive maintenance and functional checks should be integrated into the facility's equipment management schedule to ensure consistent performance and minimize downtime.

Sterility monitoring during routine operation revealed microbial growth inside washer chambers when decontamination was delayed, highlighting the importance of timely intervention. Scheduled bleach treatment effectively eliminated this bioburden, restoring sterility. Throughout the observation period, GrenoClean and deionized water reservoirs remained sterile, indicating that contamination risk is primarily associated with internal washer surfaces rather than reagent or water containers. These findings emphasize the need for systematic cleaning and monitoring as part of good laboratory practice, particularly in workflows requiring high sterility standards.

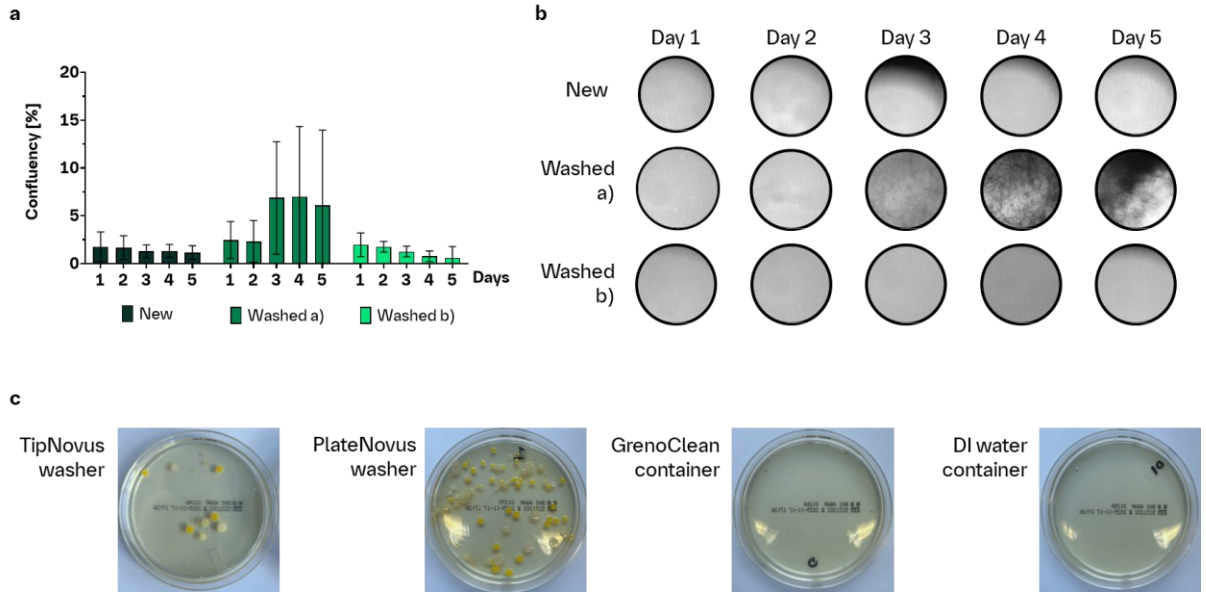

Fig. 22 | Contamination monitoring and decontamination efficiency in washing systems.

a, Confluency measurements (%) over five days in plates incubated at 37 °C, 5% CO_2 with fresh medium only (New), plates washed before decontamination (Washed a)), and plates washed after decontamination (Washed b)) of PlateNovus. Bar graph shows mean ± SD. **b,** Representative images illustrate contamination progression in Washed a and absence of growth in Washed b. **b,** BHI agar plate tests for contamination sources after five days at 37 °C. Microbial colonies were detected in TipNovus and PlateNovus washers, whereas GrenoClean and DI water containers remained sterile.

Appendix

Table 1 | List of consumables, materials and devices.

Material	Purpose	Description
Balance	Gravimetric calibration	Mettler Toledo, XPE 205, Max 200 g, min e=1 mg
Bovine Serum Albumin	Bradford assay	Sigma, BSA, heat shock fraction, ≥ 98%
Cell lines	Cell based assays	HEK-293-WT, Colo-329-DM, LS513
DMSO	Assay preparation	Sigma, Dimethyl sulfoxide
Echo	Liquid handling	Beckman Coulter, Echo 650 Acoustic Liquid Handler
Energy logger	Power consumption	Voltcraft, SEM5000 Energy-Logger
GraphPadPrism	Data analysis	GraphPad Prism Version 10.4.1 (620)
GrenoClean	Washing detergent	Oxidizing washing reagent by Grenova
Incidin Active	Disinfection	ECOLAB Incidin® Active Surface disinfection, powder
Microscopy	ACC confluency assessment	BioTek Cytation 1 Cell Imaging Multimode Reader, 4× objective
	Pipette tip integrity	Leica DMIL LED, 4× objective
	Cell count assessment	Revvity, Opera Phenix Plus High-Content Imaging System - Single Camera
Microwell plate	Cell culture assays	Greiner, Cell Culture Microplate, 384 Well, PS, F-bottom, µCLEAR®, Black, Lid, TC-treated, Sterile
		Agilent Technologies, E-Plate 384, xCELLigence assay,
Milli-Q Water	Washing detergent	Biopak Polisher, Q-Pod
Neutral Red	Neutral Red assay	Sigma, Toluidine Red
Pipette	Gravimetric calibration	Eppendorf Research Plus: 2 - 20 µL, 20 - 200 µL, 100 - 1000 µL
PlateNovus	Washing microwell plates	Grenova PR240003, Paurus 96 220V Manual Load
Plate reader	Wavelength detection	Perkin Elmer, EnSight Multimode Plate Reader
TipLumis	Cleanware storage	Grenova TL200009, TipLumis Temperature Control Tip Storage + UV - 220V
TipNovus	Washing pipette tips	Grenova TN210058, TipNovus96Tall 220V Washer Dryer W / Built-in Sonication
Tips	Assays	Eppendorf epT.I.P.S.: 0.1 - 10 µL, 20 - 200 µL, 50 - 1000 µL
		Sartorius Optifit Tips: 0.1 - 10 µL, 20 - 200 µL, 5 - 350 µL, 0 - 1200 µL
Tris-HCL	Fluorescein assay	AccuGENE, 1M Tris-HCL pH 8.0
Volume meter	Volume consumption	PCE-322A sound level meter

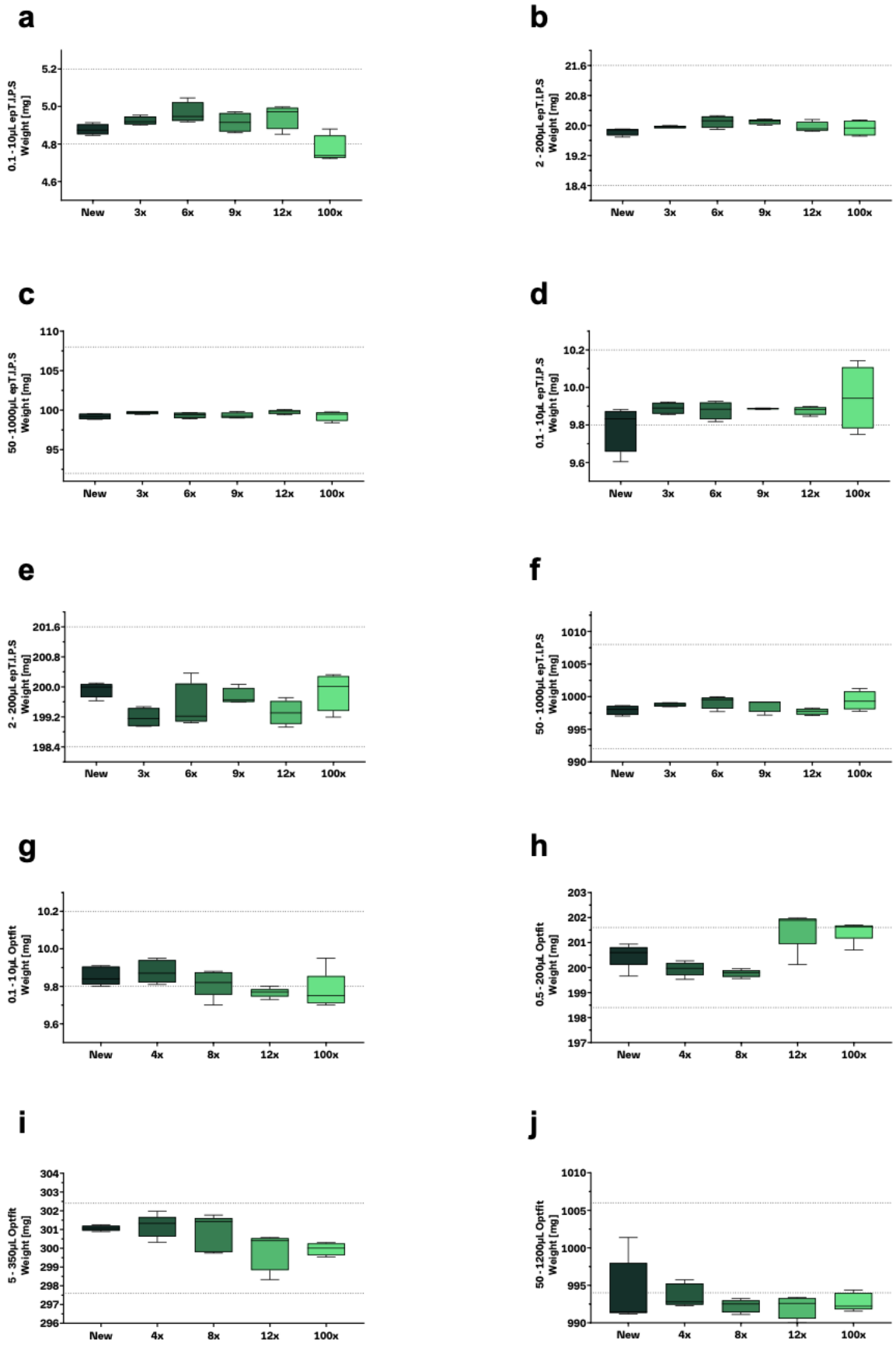

Fig. 23 |Gravimetric calibration results of epT.I.P.S (a-f) and Satorius Optifit tips(g-j)

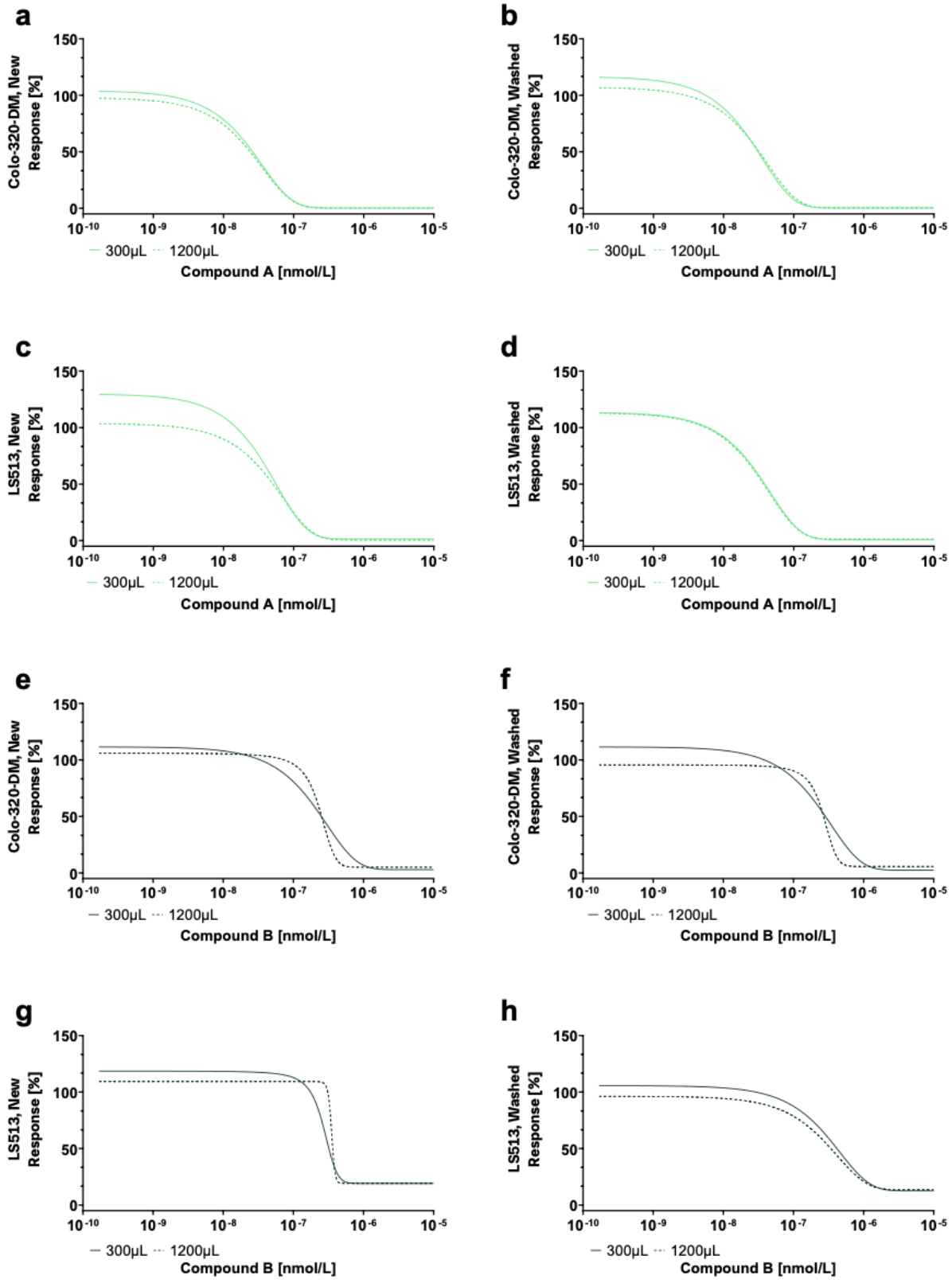

Fig. 24 | Dose-Response curves with new and washed consumables.

Table 2 | Experiment data for Dose-Response analysis.

Cell line			Compound	Top [%]	Bottom [%]	R2	IC50 [nmol/L]
Colo-320-DM	New	300	A	99.55	0.608	0.98851	24.87
		1200		93.27	0.594	0.98794	25.41
	Washed	300		112.69	0.326	0.98077	24.66
		1200		107.81	0.674	0.96855	27.19
	New	300	B	107.13	5.465	0.96648	206.40
		1200		105.58	4.763	0.95197	209.34
	Washed	300		106.38	5.174	0.97112	277.69
		1200		97.77	5.710	0.94331	248.31
LS513	New	300	A	124.39	2.682	0.96304	39.48
		1200		95.438	2.678	0.96967	42.54
	Washed	300		109.79	1.158	0.98501	31.30
		1200		106.52	0.412	0.95451	31.93
	New	300	B	107.97	16.829	0.97221	201.62
		1200		99.90	31.746	0.88459	227.51
	Washed	300		103.09	17.104	0.91316	338.30
		1200		93.83	16.772	0.95058	254.47

Table 3 | Cost and sustainability analysis of TipNovus and PlateNovus washing systems.

System	TipNovus	PlateNovus
Water (L/cycle)	10	15
Electricity (kWh/cycle)	0.15	1.91
Cycle time (min)	35	230
Items/cycle	4	4
Reuse cycles (planned)	12	5
New item cost (€)	15	7
Cycles/year	1040	624
Water cost/cycle (€): €0,002 per L	0.02	0.03
Electricity cost/cycle (€): €0,25 per kWh	0.04	0.48
Reagent cost/cycle (€)	0.5	0.5
Labor cost/cycle (€)	9.38	9.38
Total wash cost/cycle (€)	9.93	10.38
Wash cost per item (€)	2.48	2.6
Avoided purchase per item (€)	15	7
Net savings per item (€)	12.52	4.4
Net savings per cycle (€)	50.07	17.62
Annual savings (€)	52070	10993
CO2e saved per cycle (kg)	1.097	0.624
CO2e saved per year (t)	1.14	0.39

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Additional information:

For this thesis, Microsoft Copilot was utilized to enhance grammar, clarity, and overall readability. The tool provided suggestions for refining complex sections, structuring ideas coherently, and ensuring the thesis adhered to academic standards. A sample prompt used during the process was: "Please review and improve the following section of my thesis for grammar and readability: [Paragraph]”