

UCLA

UCLA Electronic Theses and Dissertations

Title

Rapid Quantification of Microplastics using Smartphone

Permalink

<https://escholarship.org/uc/item/213891c7>

Author

Leonard, Dona Jamie L

Publication Date

2021

Peer reviewed|Thesis/dissertation

UNIVERSITY OF CALIFORNIA

LOS ANGELES

Rapid Quantification of Microplastics using Smartphone

A Thesis Submitted In Partial Satisfaction of

The Requirements For the Degree

Masters of Science in Civil Engineering

by

Dona Jamie Leigh Leonard

2021

© Copyright by
Dona Jamie Leonard
2021

ABSTRACT OF THE THESIS

Rapid Quantification of Microplastics Using Smartphone

by

Dona Jamie Leigh Leonard

Master of Science in Civil Engineering

University of California, Los Angeles 2021

Professor Sanjay K. Mohanty, Chair

Rapid detection of microplastics is critical to understand the extent of microplastic contamination in the environment and identify appropriate mitigation strategies. However, current methods for isolating and counting microplastics from environmental samples can take several hours to days and their efficiencies may vary widely. Herein a rapid method to detect microplastics is described, which involves staining them with Nile Red before taking picture using a mobile phone fitted with add-on camera. The method is capable of detecting and quantifying microplastics as small as 10 μm and identifying microplastics among other debris, thereby eliminating the need for time-consuming digestion steps using hazardous chemicals. The method is effective over a wide range of light-colored plastic polymers, including polystyrene, polypropylene, and polyethylene terephthalate. This field deployable method can provide accessible and user-friendly tool to even citizen scientists and non-specialized labs to detect microplastics in environmental samples.

The thesis of Dona Jamie Leigh Leonard is approved.

Shaily Mahendra

David Jassby

Sanjay K. Mohanty, Committee Chair

University of California, Los Angeles

2021

Table of Contents

1	INTRODUCTION	1
2	METHODOLOGY	4
2.1	CREATION OF MOBILE PHONE ATTACHMENT AND ASSOCIATED ALGORITHM.....	4
2.2	CREATION OF MICROPLASTICS AND MICROPLASTIC STOCK SOLUTIONS	5
2.3	SAMPLE FILTER PREPARATION	6
2.4	PROTOCOL VALIDATION: DETECTION LIMIT AND RANGE	6
2.5	PROTOCOL VALIDATION: DETECTION IN ENVIRONMENTAL MEDIA	6
2.6	IMAGE ACQUISITION AND ANALYSIS.....	7
3	RESULTS AND DISCUSSION	8
3.1	RAPID DETECTION AND QUANTIFICATION OF MICROPLASTICS.....	8
3.2	DETECTION LIMIT ESTABLISHED AT 10 MICRONS	8
3.3	RELIABLE QUANTIFICATION RANGE SET BELOW 0.5 MG/L.....	9
3.4	THE METHOD WORKS FOR A RANGE OF PLASTIC POLYMERS	10
3.5	THE METHOD CAN QUANTIFY MICROPLASTICS SPIKED IN ENVIRONMENTAL MATRICES	10
3.6	FUTURE WORK	11
4	CONCLUSION	13
5	REFERENCES	14
6	APPENDIX	19

List of Figures

Figure	Page
1: (A) 3D rendering of the mobile attachment to capture images of filter papers with labeled components. (B) graphical summary of associated filter preparation methodology	5
2: (A) Smartphone prototype images of filters after serial dilution of sanded polystyrene microplastics in distilled water (B) Bar Graph depicting the detected number of particles on each diluted filter using smartphone algorithm.....	9
3: Filter results from smartphone detection of sanded microplastics: polystyrene, tire wear, PET, PP. Fluorescence means smartphone detection and quantification was successful. ..	10
4: (A) Smartphone image of filter of polystyrene sanded microplastics in spiked soil environment after dilution taken by the prototype. (B) Image showing the count of microplastics found by the algorithm after analysis. (C) Boxplot visualizing a statistical significant difference between microplastic numbers in spiked versus non-spiked soil experiment.	11
5: Depiction of a semi-linear relationship between area in pixel sizes versus known microplastic particle size	12

List of Acronyms

KI	Potassium Iodide
NaI	Sodium Iodide
DNG	Digital Negative Image
MATLAB	Matrix Laboratory Software
PS	polystyrene
PET	polyethylene terephthalate
PP	polypropylene
FTIR	Fourier Transform Infrared (FTIR) microscopy
SEM	Scanning Electron microscopy (SEM)

Acknowledgements

First and foremost, I am extremely grateful to my advisor and committee chair, Dr. Sanjay K. Mohanty for his invaluable advice, unwavering support, and patience during my academic journey. In a single sentence, I cannot fully encapsulate the extent of his positive influence during both my undergraduate and my burgeoning graduate journey. Without his encouragement and mentorship, I would not be here today. I also thank other committee members, Dr. Shaily Mahendra and Dr. David Jassby for time and energy in reviewing my thesis.

A special thanks to Dr. Hatice Ceylan Koydemir with the Ozcan Research Group at UCLA for her intellectual contributions and development of the mobile phone technology and algorithm. This research would not exist without her innovation and tireless hours of lab work and collaboration to finalize the best technology and techniques.

I would also like to recognize the support and mentorship of my peers in the Subsurface Engineering and Analysis Laboratory (SEALab) at UCLA. Working as an undergrad in this lab before embarking on my own graduate education journey has been an inimitable experience, strengthened by the camaraderie of my lab mates. In particular I would like to thank Vera Smirnova Koutnik and Ansh Borthakur - not only for their critical help in the lab and the development of this methodology, but also for their unwavering support and constructive criticism at every stage of my work.

The support of my family and friends have set me on course for success at every stage of my academic career, for which I am grateful. Thank you.

My gratitude as well to the Department of Civil and Environmental Engineering at UCLA for awarding me the 2020-2021 Graduate Opportunity Fellowship which provided the financial support for me to stay on for my Masters of Science.

Gratefully,

Jamie Leonard

1 Introduction

Plastic is one of the most abundant man-made materials found in the environment (Geyer et al., 2017) and their concentration there is expected to double by 2030 (Hale et al., 2020). They are especially insidious due to both their sheer volume and predicted toxicity, which stems from their resistance to biodegradation, leaching of harmful additives, and pollutants adsorbed on their surface (Koyuncuoğlu & Erden, 2021; Mohajerani & Karabatak, 2020).

While the adverse effects of microplastic contamination are being further explored, the true extent of contamination has still not yet been defined and can vary by orders of magnitude based on isolation methodologies (Koutnik, Leonard, et al., 2021). These microplastic isolation and quantification methods are currently unstandardized. In particular, reported microplastic concentrations have been shown to vary based on methodology variability such as: sample collection method, sample volume, filter cutoff size used to isolate plastic particles from water, organic digestion method, density stratification solution, counting methodology, and detection limit (Koutnik, Alkidim, et al., 2021). Traditional methodologies to isolate microplastics include: density stratification (Fries et al., 2013; Nuelle et al., 2014), screening and sieving (Zobkov & Esiukova, 2017), and organic digestion (Zobkov & Esiukova, 2017). Then microplastics are quantified with either a normal microscope, Raman spectroscopy (Primpke et al., 2017; Renner et al., 2017), Fourier Transform Infrared (FTIR) microscopy (Tagg et al., 2015), Scanning Electron microscopy (SEM) (Fries et al., 2013), or a combination of pyrolysis with gas chromatography and mass (Beaurepaire et al., 2021). Typically, these detection techniques are performed in conjunction with image acquisition software that automatically count microplastics from an image. While many of these methodologies are highly sensitive and accurate, they often require complex sample preparation procedures, expensive and bulky microscopy instruments,

and can take hours to days by professionally trained personnel (Lusher et al., 2020). Thus, there is a clear need to rapidly and more easily detect microplastics from environmental samples to better understand the scope of environmental contamination and thus the extent of predicted toxicity.

Recently technological advancements have enabled the use of smartphone to rapidly quantify many pollutants: mercury (Wang et al., 2016; Xiao et al., 2016), food allergens (Coskun et al., 2013), 2,4,6-trinitrotoluene (TNT) (Zhang et al., 2015), *E. coli* (Zhu et al., 2012) and other pathogenic bacteria (Gopinath et al., 2014), and other biological components such as prostate-specific antigen (PSA) (Barbosa et al., 2015). A combination of smartphones and various connected devices (i.e., high-performance cameras and light sensors) are used to differentiate the contaminant of interest from other debris or interferences. However, a smartphone has never been used to quantify microplastics before. The challenge of detecting microplastics from a picture taken by a smartphone is that it alone does not have ability to distinguish plastics from any other particles. However, Nile Red dye has been demonstrated to be capable of detecting microplastics among other debris (Maes et al., 2017). This method takes advantage of the fact that plastics coated with Nile Red are fluorescent under UV light whereas organic debris or other natural materials are not (Erni-Cassola et al., 2017; Kang et al., 2020; Prata et al., 2019). Therefore, the dye can be used to distinguish microplastics from other debris in a picture taken by smartphone. The thesis uses this underlying hypothesis to develop a smartphone-based method.

The objective of this thesis is to develop a smartphone-based methodology to replace traditional labor-intensive microplastic detection methods, and thus significantly improve the ease of detection of microplastics in the environment. To provide an accurate and portable

platform to sensitively quantify microplastic in water and soil samples, a system that consists of a Lumia 1020 smartphone and a lightweight 3D printed attachment was created to image and count microplastics as small as 10 μm with increased efficiency and decreased processing time.

2 Methodology

2.1 Creation of mobile phone attachment

The mobile phone sensor system was assembled manually in the lab; Figure 1 displays a labeled 3D model of the current prototype. It consists of a smartphone and mobile phone attachment whose components include: 3D printed housing, 4 light emitting diodes (LEDs) (Digikay, C503B-BAN-CY0C0461), an emission filter (Semrock, BLP01-532R-25), an external lens (Edmund Optics, 49-662,), and a rechargeable battery (Kastar, BOOUPVK2B6) (Figure1A). This prototype was built for an inexpensive Windows Nokia Lumia 1020 smartphone with a centered back camera of 48 megapixels. Blue LEDs were then equidistantly positioned to uniformly light up sample filters and excite the fluorophores on the stained microplastics. A 25 mm (diameter) by 30 mm aspheric achromatic external glass lens provided magnification once the sample holder was adjusted to the lens's focal point to improve image clarity. A 25 mm long pass edge emission filter was used to filter out wavelengths under 532 nanometers. This configuration provides a large field of view (FOV) (i.e., ~490 mm²) that could capture the image of the entire 25 mm membrane in a single image.

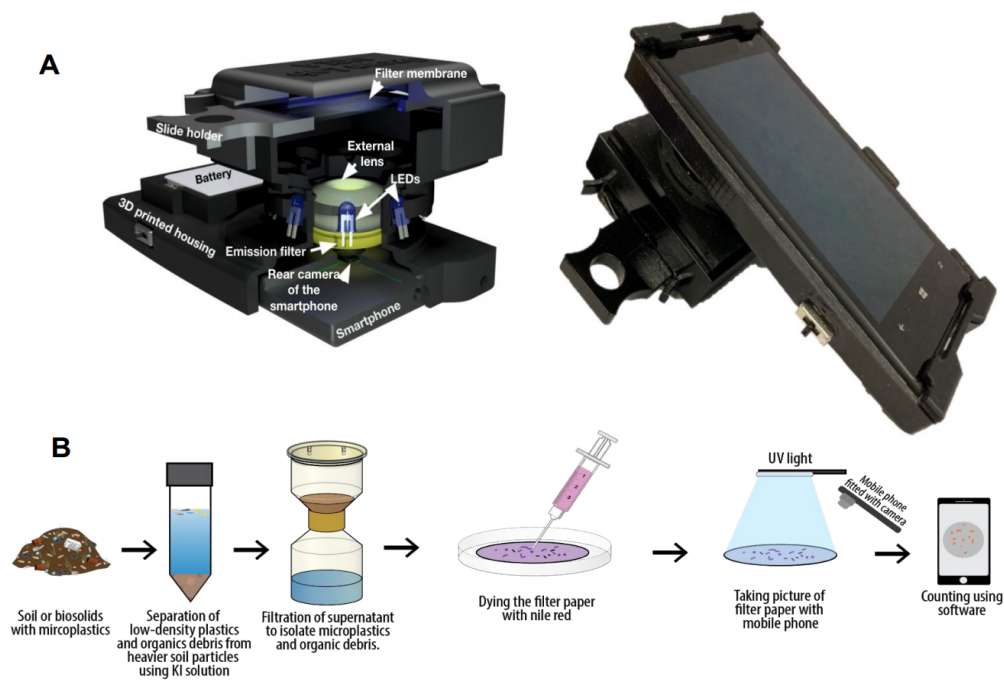


Figure 1: (A) 3D rendering of the mobile attachment to capture images of filter papers with labeled components. (B) graphical summary of associated filter preparation methodology.

2.2 Creation of microplastics and microplastic stock solutions

To validate the imaging, multiple types of plastic polymer and shape were used: standard polystyrene microsphere solution of various sizes (ThermoFisher Scientific, F13838), and microplastics created in the lab by sanding plastic objects both manually and with a commercial sanding gun. To determine the detection limit, initial non-fluorescent microsphere stocks from a flow cytometry calibration kit (product no. F13838, ThermoFisher Scientific) were diluted by adding 10 μL of the stock solution to 10 mL of DI water and then serially diluting them to achieve lower concentrations. In order to test a range of plastic polymers: polystyrene, polyethylene terephthalate, polypropylene, and tire (Nylon) particles, stock suspension were

made by suspending 0.5 g of abraded microplastics in 1 L deionized water and serially diluting the stock up to five orders of magnitude.

2.3 Sample filter preparation

Microplastic suspensions were vacuum filtrated using 25 mm G4 glass fiber filters with 1.2 μm pore size (Thermofisher Scientific, 09-804-24C). Filters containing microplastics were stained with 0.17 ml of 0.5 $\mu\text{g mL}^{-1}$ Nile Red in chloroform solution (Maes et al., 2017) in a glass petri dish and dried for around 30 min to an hour in the fume hood. Filters were then transferred to glass slides and imaged using the mobile phone and an Olympus microscope with Cy-3 attachment and 10x magnification for comparison.

2.4 Protocol validation: detection limit and range

To quantify the size detection limit of the method, polystyrene microsphere suspensions of size 2, 4, 6, 10, 15, 20, 42, 98, and 222 μm were used. As fluorescence light emitted from Nile Red stained microplastics was proportional with size, the detection limit was based on the ability to discern the fluorescence conferred on the particles by Nile Red from the background fluorescence of the filters. To determine the effectiveness of this method to detect a wider range of plastic polymers, abraded PET, PP, and tire nylon particulates were also tested as is without separating them into different size classes.

2.5 Protocol validation: detection in environmental media

To quantify microplastics from environmental samples, contaminated samples were created by spiking 1 gram of soil with 10 mL of the 1000x serially diluted polystyrene stock solution. Denser sediments were settled in 40 mL of 1.6 g cm^{-3} KI solution (Roch & Brinker, 2017) using a centrifuge (5000 rpm for 30 minutes). The supernatant was vacuum filtered and

discarded. The membrane with filtered debris containing microplastics was stained with 0.17 mL of $0.5 \mu\text{g mL}^{-1}$ Nile Red in chloroform solution (Maes et al., 2017) in a glass petri dish and dried for around 30 min to an hour. Filters were then transferred to glass slides and imaged using the prototype smartphone. Triplicate spiked soil samples were compared to triplicate uncontaminated control samples (no added microplastics).

2.6 Image acquisition and analysis

The smartphone attachment was powered up which illuminated the LEDs. The glass slides were then inserted into the attachment, where the LED light further excited the Nile Red microplastic fluorophores. This fluorescence emitted red light, which passed through the external lens to the smartphone camera where the image was recorded in DNG format (Digital Negative Image). These images were analyzed in MATLAB, where the red channel, corresponding to the fluorescence wavelength of Nile Red, was isolated and the number of particles and the pixel area, eccentricity, orientation, equivalent diameter, major axis length, minor axis length and perimeter of each particle was recorded. The algorithm used for analysis was successful at differentiating the background and fluorescent particles and estimating the number of particles on each filter.

To compare the microplastic counts from the smartphone with a traditional microscopy technique, an Olympus IX83 microscope equipped with a Cy-3 attachment was used. The microscope was covered with a black cloak to eliminate light leakage from other sources. Glass slides containing the microplastic samples were mounted onto an automated stage and imaged under 4x and 10x objective lenses. The images were analyzed using a MetaMorph software to count the number of microplastics in the samples. These resulting images were analyzed in MATLAB using similar algorithm.

3 Results and Discussion

3.1 Rapid detection and quantification of microplastics

The overall filter preparation methodology was a highly streamlined process. Compared to other time-intensive methodologies which take around 2-3 hours to isolate the microplastics, Nile Red techniques significantly reduced the time required to around 30 minutes to an hour with drying time (Al-Azzawi et al., 2020; Tagg et al., 2017). In addition, the time required to quantify the microplastics using the smartphone was significantly less than conventional methods. For instance, the conventional microscopy would require stitching of 254 images from zoomed areas (due to smaller field of view) to capture the entire image of the filter membrane. In comparison, smart-phone method required a single snapshot to detect and quantify the microplastics. Thus, smart phone provides equivalent data with much fewer processing steps. The innovative method will help streamline field quantification studies, making microplastic contamination reporting more accessible and feasible.

3.2 Detection limit established at 10 microns

Detection limit experiments revealed that 10 μm microplastics were the minimum size at which the mobile phone attachment could reliably separate microplastic fluorescence signatures from the background filter fluorescence. While plastics below 10 μm could still be visibly detected, they cannot be accurately quantified by the current algorithm (see Figure S1). Nevertheless, even in its initial form, this matches the majority of existing methods which could not detect microplastics below 20 μm reliably (Koutnik, Alkidim, et al., 2021).

3.3 Reliable quantification range set below 0.5 mg L⁻¹

Dilution of polystyrene stock solution successfully reduced microplastic counts below 0.5 mg L⁻¹ of microplastics, at which reliable quantification was possible. Visual surveyance of filters during the calibration phase of this methodology revealed issues in quantification accuracy when dealing with extremely high contamination levels. High numbers of particles formed clusters (Figure 2A), and fluorescence from larger conglomerates obscured that of smaller particles, creating significant error. Polystyrene stock solutions were diluted prior to spiking soils used in experiments (Figure 2A). As expected, microplastic counts decreased with magnitude of dilution (Figure 2B). Reliable quantification was set at the 1000x dilution mark, or approximately 0.5 mg L⁻¹.

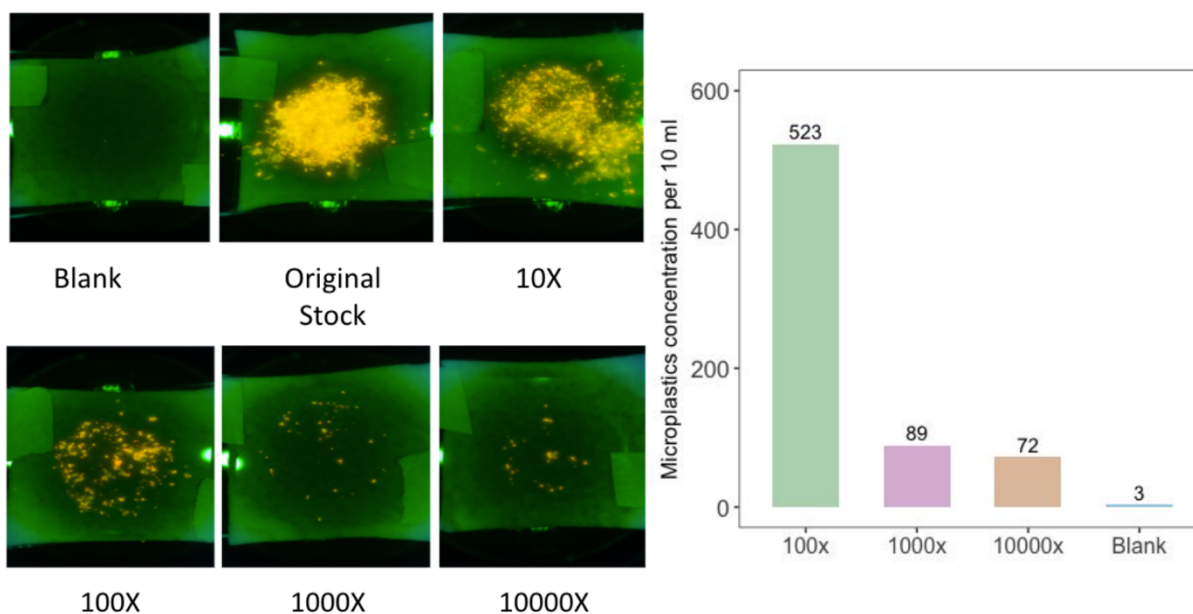


Figure 2: (A) Smartphone prototype images of filters after serial dilution of sanded polystyrene microplastics in distilled water (B) Bar Graph depicting the detected number of particles on each diluted filter using smartphone algorithm.

3.4 The method works for a range of plastic polymers

Polystyrene (PS), polypropylene (PP), and polypropylene terephthalate (PET) were all successfully detected and quantified using this methodology (Figure 3). On the contrary, tire microplastics, which were black in color, were not fluorescent on the filters and therefore were not detected. This result suggests that the method is limited to only light colored plastic polymers which has already been shown as a limitation of using Nile Red dye for identification of microplastics (Stanton et al., 2019).

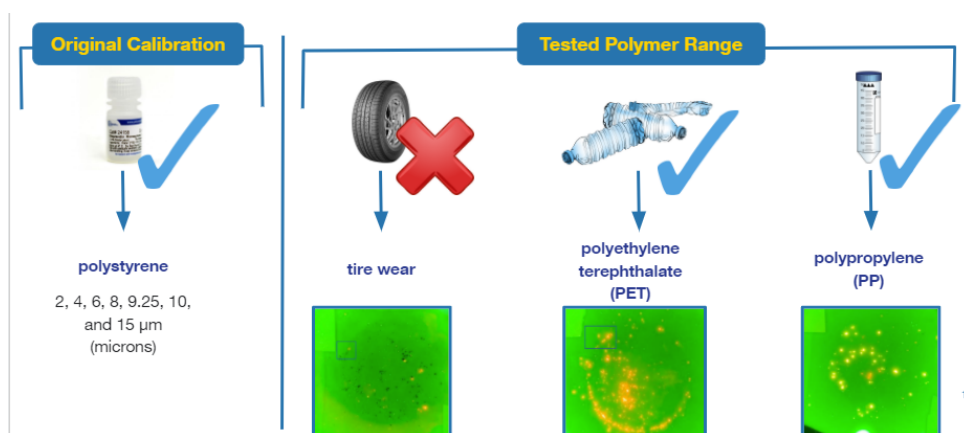


Figure 3: Filter results from smartphone detection of sanded microplastics: polystyrene, tire wear, PET, PP. Fluorescence means smartphone detection and quantification was successful.

3.5 The method can quantify microplastics spiked in environmental matrices

Polystyrene spiked soils were quantified using the smartphone methodology to further validate its utility for environmental samples. To prevent bulky organic material blocking the fluorescent microplastics, these soil samples were diluted once and counts scaled up to estimate true concentrations. A sample filter is displayed below (Figure 4) in order to demonstrate that smartphone technology was able to both visually detect and quantify contaminating microplastic pieces in environmental samples. Figure 4B depicts the statistically significant difference in

microplastic counts between spiked soil and non-spiked soil proving the success of spike and recovery experiments. This confirmed the utility of this methodology to quantify microplastic contamination in environmental matrices.

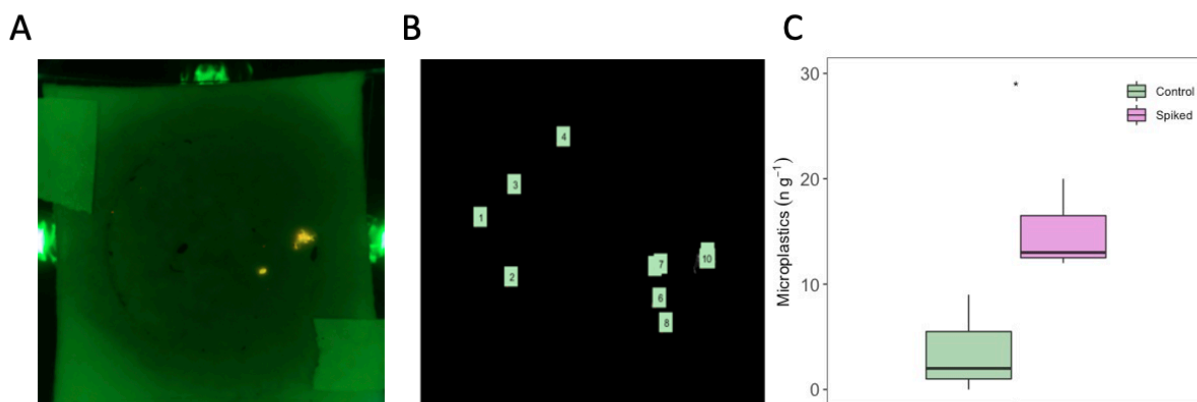


Figure 4: (A) Smartphone image of filter of polystyrene sanded microplastics in spiked soil environment after dilution taken by the prototype. (B) Image showing the count of microplastics found by the algorithm after analysis. (C) Boxplot visualizing a statistical significant difference between microplastic numbers in spiked versus non-spiked soil experiment.

3.6 Future work

This work presents the initial proof-of-concept study to examine whether smartphone can be used to quantify microplastics. These results proved that it can. However, there are many optimization steps needed to make this technology ready for future implementation. Future work should focus on reducing the current limitations of the prototype and improving accuracy.

For example, the algorithm should be improved to better predict the size of microplastics in environmental samples from the pixel size outputted by Matlab. Preliminary data depicted in Figure 5 demonstrates how recorded pixel size from the algorithm increases relatively proportionally with known microplastic size; however, the range also varied since pixel size was

dependent on luminescence radius captured by the smartphone. If pixel size could be definitively correlated to actual size, then microplastic contamination could not only be quantified but size fractions determined as well.

Furthermore, future work should focus on calibrating the prototype to lower the detection limit to the sub-micron level, reformatting the computer algorithm as an open access application, and alternating fluorescent dye beyond Nile Red to try to account for dark colored microparticles such as tire particles, which are one of the most prevalent microplastic types in urban areas. This will allow this technology to supersede all other traditional isolation, detection, and quantification methodologies in both effectiveness and utility.

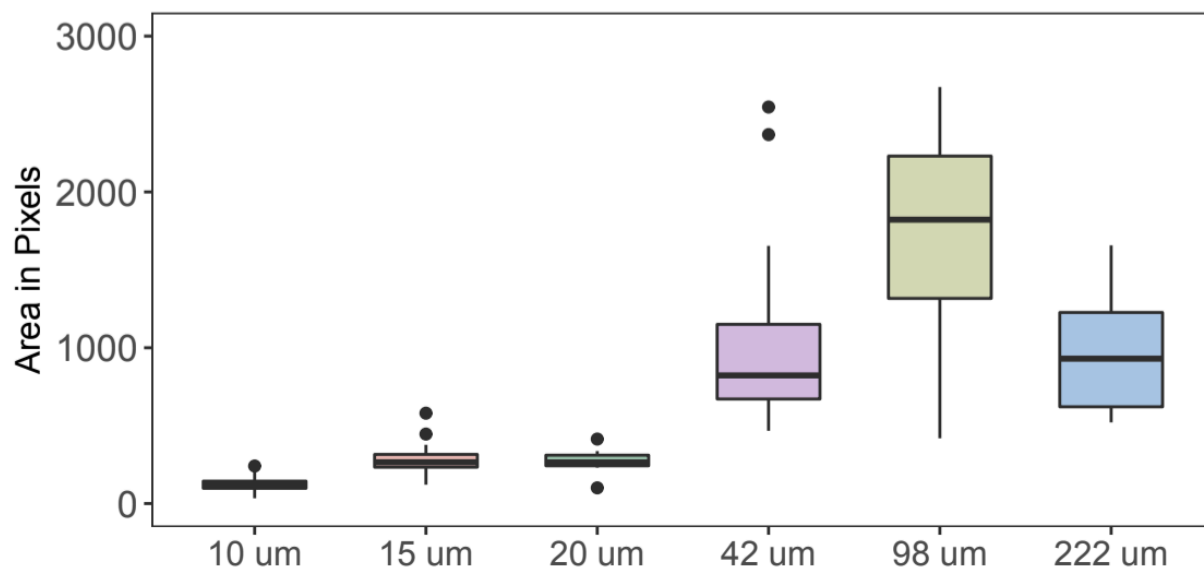


Figure 5: Depiction of a semi-linear relationship between area in pixel sizes versus known microplastic particle size

4 Conclusion

The work describes a novel portable technology to rapidly quantify microplastics in environmental samples, which could replace manual microplastic fluorescence microscopy and thus significantly improve the ease of detection of microplastic contamination. The main findings are:

- An inexpensive smartphone can be retrofitted with an external lens attachment and used to capture images of and quantify fluorescent microplastics from contaminated environmental matrices.
- This technology has a detection limit of 10 μm , which is on par with the lowest detection levels in existing methods from environmental matrices such as soil, sludges, and wastewater influent/effluents.
- This technology can successfully be used to detect and quantify common plastic polymer types: PS, PP, PET.

Further work is needed to calibrate, calculate, and improve the accuracy and precision of this methodology; nevertheless, this work provides the first description of a completely novel technology with widespread applications for understanding microplastic contamination in all environmental matrices.

5 References

- Al-Azzawi, M. S. M., Kefer, S., Weißer, J., Reichel, J., Schwaller, C., Glas, K., Knoop, O., & Drewes, J. E. (2020). Validation of Sample Preparation Methods for Microplastic Analysis in Wastewater Matrices—Reproducibility and Standardization. *Water*, *12*(9), 2445. <https://doi.org/10.3390/w12092445>
- Barbosa, A. I., Gehlot, P., Sidapra, K., Edwards, A. D., & Reis, N. M. (2015). Portable smartphone quantitation of prostate specific antigen (PSA) in a fluoropolymer microfluidic device. *Biosensors & Bioelectronics*, *70*, 5–14. <https://doi.org/10.1016/j.bios.2015.03.006>
- Beaurepaire, M., Dris, R., Gasperi, J., & Tassin, B. (2021). Microplastics in the atmospheric compartment: A comprehensive review on methods, results on their occurrence and determining factors. *Current Opinion in Food Science*, *41*, 159–168. <https://doi.org/10.1016/j.cofs.2021.04.010>
- Coskun, A. F., Wong, J., Khodadadi, D., Nagi, R., Tey, A., & Ozcan, A. (2013). A personalized food allergen testing platform on a cellphone. *Lab on a Chip*, *13*(4), 636–640. <https://doi.org/10.1039/c2lc41152k>
- Erni-Cassola, G., Gibson, M. I., Thompson, R. C., & Christie-Oleza, J. A. (2017). Lost, but Found with Nile Red: A Novel Method for Detecting and Quantifying Small Microplastics (1 mm to 20 μ m) in Environmental Samples. *Environmental Science & Technology*, *51*(23), 13641–13648. <https://doi.org/10.1021/acs.est.7b04512>
- Fries, E., Dekiff, J. H., Willmeyer, J., Nuelle, M.-T., Ebert, M., & Remy, D. (2013). Identification of polymer types and additives in marine microplastic particles using

- pyrolysis-GC/MS and scanning electron microscopy. *Environmental Sciences: Processes and Impacts*, 15(10), 1949–1956. Scopus. <https://doi.org/10.1039/c3em00214d>
- Geyer, R., Jambeck, J. R., & Law, K. L. (2017). Production, use, and fate of all plastics ever made. *Science Advances*, 3(7). <https://doi.org/10.1126/sciadv.1700782>
- Gopinath, S. C. B., Tang, T.-H., Chen, Y., Citartan, M., & Lakshmipriya, T. (2014). Bacterial detection: From microscope to smartphone. *Biosensors & Bioelectronics*, 60, 332–342. <https://doi.org/10.1016/j.bios.2014.04.014>
- Hale, R. C., Seeley, M. E., Guardia, M. J. L., Mai, L., & Zeng, E. Y. (2020). A Global Perspective on Microplastics. *Journal of Geophysical Research: Oceans*, 125(1), e2018JC014719. <https://doi.org/10.1029/2018JC014719>
- Kang, H., Park, S., Lee, B., Ahn, J., & Kim, S. (2020). Modification of a Nile Red Staining Method for Microplastics Analysis: A Nile Red Plate Method. *Water*, 12(11), 3251. <https://doi.org/10.3390/w12113251>
- Kappler, A., Fischer, D., Oberbeckmann, S., Schernewski, G., Labrenz, M., Eichhorn, K. J., & Voit, B. (2016). Analysis of environmental microplastics by vibrational microspectroscopy: FTIR, Raman or both? *Analytical and Bioanalytical Chemistry*, 408(29), 8377–8391. <https://doi.org/10.1007/s00216-016-9956-3>
- Koutnik, V. S., Alkidim, S., Leonard, J., DePrima, F., Cao, S., Hoek, E. M. V., & Mohanty, S. K. (2021). Unaccounted Microplastics in Wastewater Sludge: Where Do They Go? *ACS ES&T Water*, 1(5), 1086–1097. <https://doi.org/10.1021/acsestwater.0c00267>
- Koutnik, V. S., Leonard, J., Alkidim, S., DePrima, F. J., Ravi, S., Hoek, E. M. V., & Mohanty, S. K. (2021). Distribution of microplastics in soil and freshwater environments: Global

- analysis and framework for transport modeling. *Environmental Pollution*, 274, 116552.
<https://doi.org/10.1016/j.envpol.2021.116552>
- Koyuncuoğlu, P., & Erden, G. (2021). Sampling, pre-treatment, and identification methods of microplastics in sewage sludge and their effects in agricultural soils: A review. *Environmental Monitoring and Assessment*, 193(4), 175. <https://doi.org/10.1007/s10661-021-08943-0>
- Lusher, A. L., Munno, K., Hermabessiere, L., & Carr, S. (2020). Isolation and Extraction of Microplastics from Environmental Samples: An Evaluation of Practical Approaches and Recommendations for Further Harmonization. *Applied Spectroscopy*, 74(9), 1049–1065.
<https://doi.org/10.1177/0003702820938993>
- Maes, T., Jessop, R., Wellner, N., Haupt, K., & Mayes, A. G. (2017). A rapid-screening approach to detect and quantify microplastics based on fluorescent tagging with Nile Red. *Scientific Reports*, 7. <https://doi.org/10.1038/srep44501>
- Mohajerani, A., & Karabatak, B. (2020). Microplastics and pollutants in biosolids have contaminated agricultural soils: An analytical study and a proposal to cease the use of biosolids in farmlands and utilise them in sustainable bricks. *Waste Management (New York, N.Y.)*, 107, 252–265. <https://doi.org/10.1016/j.wasman.2020.04.021>
- Nuelle, M. T., Dekiff, J. H., Remy, D., & Fries, E. (2014). A new analytical approach for monitoring microplastics in marine sediments. *Environmental Pollution*, 184, 161–169.
<https://doi.org/10.1016/j.envpol.2013.07.027>
- Prata, J. C., da Costa, J. P., Duarte, A. C., & Rocha-Santos, T. (2019). Methods for sampling and detection of microplastics in water and sediment: A critical review. *TrAC Trends in Analytical Chemistry*, 110, 150–159. <https://doi.org/10.1016/j.trac.2018.10.029>

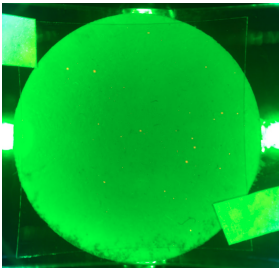
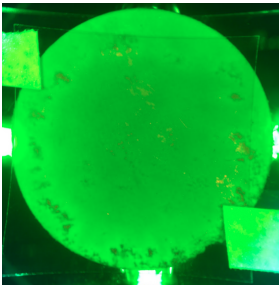

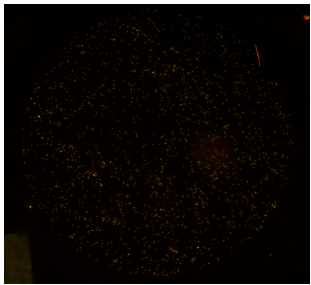
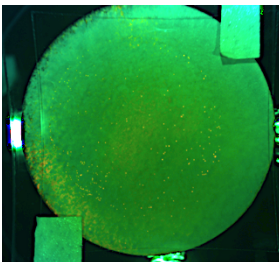
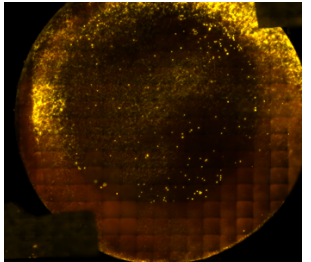
- Primpke, S., Lorenz, C., Rascher-Friesenhausen, R., & Gerdt, G. (2017). An automated approach for microplastics analysis using focal plane array (FPA) FTIR microscopy and image analysis. *Analytical Methods*, 9(9), 1499–1511.
<https://doi.org/10.1039/c6ay02476a>
- Renner, G., Schmidt, T. C., & Schram, J. (2017). A New Chemometric Approach for Automatic Identification of Microplastics from Environmental Compartments Based on FT-IR Spectroscopy. *Analytical Chemistry*, 89(22), 12045–12053.
<https://doi.org/10.1021/acs.analchem.7b02472>
- Roch, S., & Brinker, A. (2017). Rapid and Efficient Method for the Detection of Microplastic in the Gastrointestinal Tract of Fishes. *Environmental Science & Technology*, 51(8), 4522–4530. <https://doi.org/10.1021/acs.est.7b00364>
- Stanton, T., Johnson, M., Nathanail, P., Gomes, R. L., Needham, T., & Burson, A. (2019). Exploring the Efficacy of Nile Red in Microplastic Quantification: A Costaining Approach. *Environmental Science & Technology Letters*, 6(10), 606–611.
<https://doi.org/10.1021/acs.estlett.9b00499>
- Tagg, A. S., Harrison, J. P., Ju-Nam, Y., Sapp, M., Bradley, E. L., Sinclair, C. J., & Ojeda, J. J. (2017). Fenton's reagent for the rapid and efficient isolation of microplastics from wastewater. *Chemical Communications*, 53(2), 372–375.
<https://doi.org/10.1039/c6cc08798a>
- Tagg, A. S., Sapp, M., Harrison, J. P., & Ojeda, J. J. (2015). Identification and Quantification of Microplastics in Wastewater Using Focal Plane Array-Based Reflectance Micro-FT-IR Imaging. *Analytical Chemistry*, 87(12), 6032–6040.
<https://doi.org/10.1021/acs.analchem.5b00495>

- Wang, L., Li, B., Xu, F., Shi, X., Feng, D., Wei, D., Li, Y., Feng, Y., Wang, Y., Jia, D., & Zhou, Y. (2016). High-yield synthesis of strong photoluminescent N-doped carbon nanodots derived from hydrosoluble chitosan for mercury ion sensing via smartphone APP. *Biosensors & Bioelectronics*, *79*, 1–8. <https://doi.org/10.1016/j.bios.2015.11.085>
- Xiao, W., Xiao, M., Fu, Q., Yu, S., Shen, H., Bian, H., & Tang, Y. (2016). A Portable Smartphone Readout Device for the Detection of Mercury Contamination Based on an Aptamer-Assay Nanosensor. *Sensors (Basel, Switzerland)*, *16*(11). <https://doi.org/10.3390/s16111871>
- Zhang, D., Jiang, J., Chen, J., Zhang, Q., Lu, Y., Yao, Y., Li, S., Logan Liu, G., & Liu, Q. (2015). Smartphone-based portable biosensing system using impedance measurement with printed electrodes for 2,4,6-trinitrotoluene (TNT) detection. *Biosensors & Bioelectronics*, *70*, 81–88. <https://doi.org/10.1016/j.bios.2015.03.004>
- Zhu, H., Sikora, U., & Ozcan, A. (2012). Quantum dot enabled detection of Escherichia coli using a cell-phone. *The Analyst*, *137*(11), 2541–2544. <https://doi.org/10.1039/c2an35071h>
- Zobkov, M., & Esiukova, E. (2017). Microplastics in Baltic bottom sediments: Quantification procedures and first results. *Marine Pollution Bulletin*, *114*(2), 724–732. <https://doi.org/10.1016/j.marpolbul.2016.10.060>

6 Appendix

Table S 1: Average pixel size of individual particles detected by smartphone prototype as compared to their known microplastic size (μm).

MICROPLASTIC SIZE (μM)	PIXEL SIZE (MEAN \pm STD)
10	123.70 \pm 44.55
15	274.70 \pm 92.96
20	270.01 \pm 74.50
42	1027.00 \pm 583.15
98	2498.59 \pm 1394.36
222	981.63 \pm 382.56

Sizing (microns)	Smartphone Prototype	Olympus Microscope	Average Pixel Size According to Smartphone
2			---
4			---
6			---
10			123.70 ± 44.55

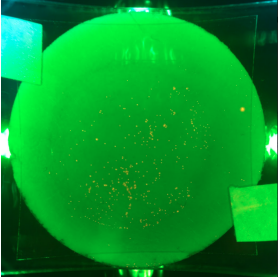
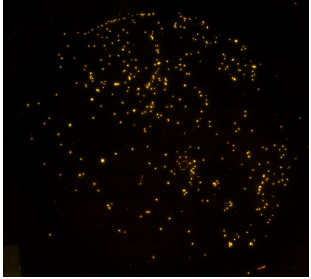
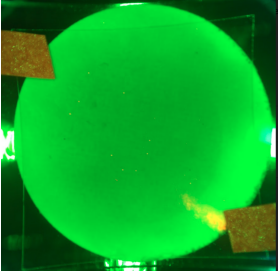
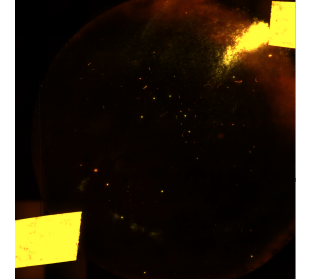
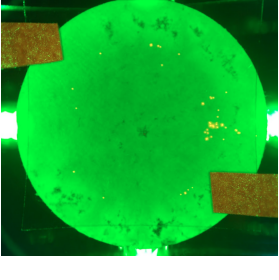
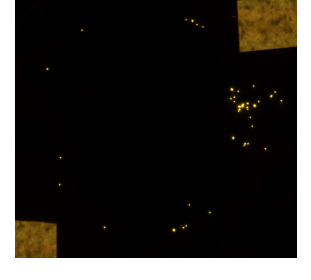
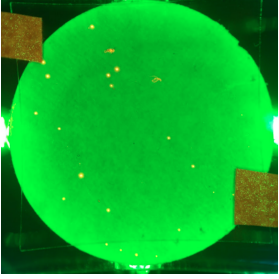

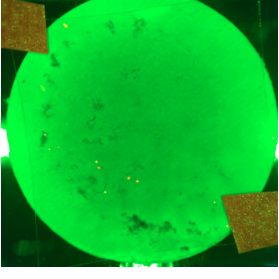
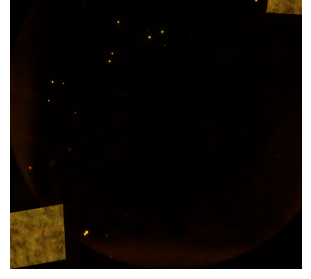
15			274.70 ± 92.96
20			270.91 ± 74.50
42			1027.00 ± 583.15
98			2498.59 ± 1394.36
222			981.63 ± 382.56

Figure S 1: Comparison between filters as imaged by the smartphone prototype and an Olympus microscope with CY3 attachment for full size range with approximately pixel sizes. Note: Olympus microscope comparison was not useful for filters below detection limit.