Brunel University of London

Pocket Guide

HiLight 2026 FLIM & Photonics

Innovating Multiphoton FLIM for Biomedical Research and Healthcare

4th-5th March 2026, London

Across Europe, one FLIM community

Social media: #HILIGHT2026

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Welcome

Dear delegates,

It is a pleasure to bring together colleagues and friends from across the UK and Europe for two days focused on advancing fluorescence lifetime imaging microscopy (FLIM) and the wider photonics that underpin next-generation biomedical imaging and diagnostics.

This meeting forms part of the outreach activity of the **EU Horizon HILIGHT project,** which aims to address key bottlenecks that limit the speed, accessibility, and translational reach of time-resolved optical methods.

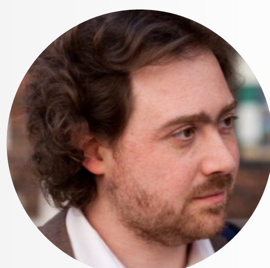
The programme reflects this scope, covering enabling technologies, from laser and detector development to analysis approaches, alongside biomedical applications. We have intentionally brought together engineers, physicists, and life scientists to encourage practical exchange between technology developers and end-users.

Posters will be presented during the breaks, and I encourage you to use these sessions to engage with presenters and fellow delegates. The meeting will close with a panel discussion in which we will debate the future of the field and how we can shape developments to address unmet challenges, particularly for biomedical research.
Ask the questions you rarely have the opportunity to explore about FLIM and two-photon microscopy.

Thank you to all speakers and delegates for contributing, the organising team for their work, the HILIGHT consortium, the EU/Innovate UK funding and Brunel University of London for hosting. We also thank our sponsors for their support, which enabled this extended two-day format.

I hope you find the discussions useful and leave with new ideas, contacts, and a clearer view of where our field is heading.

Welcome to HILIGHT 2026 and enjoy the event.



Dr Alessandro Esposito
Lecturer in Biosciences & Director CenGEM
Brunel University of London, UK

Chairman HILIGHT 2026

HiLight

Event **Statement**

HILIGHT 2026: FLIM & Photonics is a two-day, free in-person event focused on advancing Fluorescence Lifetime Imaging Microscopy (FLIM) and photonics for biological and medical applications. It forms part of our **EU Horizon HILIGHT project outreach.**

The meeting brings together leading researchers, engineers, and industry professionals from the UK and EU to share innovations, tackle technical challenges, and foster collaborations across imaging, diagnostics, and optical technologies.

The programme features **Laser Developments**, **Optical Development, Detector Innovations, FLIM Applications**, and a **Cross-topic Panel Discussion** addressing trends, challenges, and future directions.

We aim to provide a platform for anyone passionate about light-based imaging and photonics for healthcare, offering opportunities to network, gain insight, and help shape the future of FLIM and biomedical imaging.

Organisers:

Dr Alessandro Esposito
Dr Conor Treacy
Dr Mauro Chiacchia

College of Health,
Medicine and Life Sciences
Dept. of Biosciences

Social media: #HILIGHT2026

Grant Agreement
GAP-101135034

EU Project HILIGHT

In the realm of biomedical research, the limitations of current light sources hinder progress in crucial areas. Conventional instruments confine two-photon microscopy and time-resolved detection to specialised labs, impeding breakthroughs in cancer research and diagnostics. With this in mind, the **EU Horizon HILIGHT project** aims to overcome existing limitations and pave the way for transformative advancements in biomedical and healthcare applications. Specifically, it will **introduce a miniaturised optical pulse burst source and cutting-edge sensing technology.** Two key applications underscore its **potential impact: instantaneous digital histopathology and cancer research.**

With a **consortium** comprising leading partners **from five European countries,** the collaborative effort is expected to facilitate faster diagnoses and more effective treatments.

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Event Programme

4 March, 2026

Day 1 Morning

09:30 **Registration / Coffee**

10:00 **Chair introduction**
Dr Alessandro Esposito
Brunel Universty of London, UK
Welcome + Framing the Event

FLIM Applications I

10:20 Dr Thomas Blacker
University College London, UK
Biochemical mechanisms controlling metabolic sensitivity in autofluorescence.

10:45 Prof Banafshe Larijani
University of Bath, UK
A step towards precision in cancer early diagnostics and treatment.

11:10 **Coffee Break**

11:30 **Flash Talks I (4 x 3 min)**

Laser Development

11:55 Dr Gunnar Blume
Ferdinand-Braun-Institute, Germany
Short pulse semiconductor laser development at FBH for fluorescence lifetime imaging and two-photon applications.

12:20 Dr Sylvain Boust
III-V Lab, France
Versatile laser diode for 2ph FLIM at 8 Mpx/s in HILIGHT project.

12:45 **Flash Talks II (4 x 3 min)**

13:00 **Lunch / Poster Session / Sponsor tables**

HLight

Event Programme

4 March, 2026

Day 1 Afternoon

Detector Development

Time	Speaker	Talk
14:05	Dr Sara Pellegrini STMicroelectronics, UK	Pixels for FLIM: SPADs in monolithic and 3D stacked technology.
14:30	Dr Leonardo Gasparini Fondazione Bruno Kessler (FBK), Italy	Advancing FLIM Capabilities: Twenty Years of SPAD Technology Development at FBK.
14:55	Dr Claudio Bruschini École Polytechnique Fédérale de Lausanne (EPFL), Switzerland	One photon at a time - CMOS SPAD imagers for FLIM and beyond.

15:20 Coffee Break

Sponsor Tech Bite Talk

Time	Speaker	Talk
15:40	Dr Isabel Groß, PicoQuant GmbH	New Analysis Options Push the Limits of FLIM Imaging Modalities.

Optical Development

Time	Speaker	Talk
16:05	Prof Gail McConnell University of Strathclyde, UK	Multiphoton mesoscale imaging with the Mesolens.
16:30	Dr Simon Poland King's College London, UK	Novel imaging strategies for high-speed fluorescence lifetime imaging.
16:55	Prof Simon Ameer-Beg King's College London, UK	It's About Time: Using FLIM-FRET to See Molecular Interactions.

17:20 Poster Session / Sponsor tables

18:10 Networking Dinner

HLight

Event Programme

5 March, 2026

Day 2 Morning

09:30 Registration / Coffee

10:00 Morning Remarks
Dr Alessandro Esposito
Brunel Universty of London, UK

FLIM Applications II

Time	Speaker	Title
10:20	Prof Ruslan Dmitriev Ghent University, Belgium	Visualising internalisation and the biological impact of nanoplastics in live organoid models using fluorescence lifetime imaging microscopy (FLIM).
10:45	Dr Conor Treacy Brunel University of London, UK	Development of fast single-shot FLIM for digital histopathology.
11:10	Prof Marina Kuimova Imperial College London, UK	Measuring cellular viscosity with molecular rotors and FLIM.

11:35 Coffee Break

Resolving Biological Complexity

Time	Speaker	Title
11:55	Dr Falk Schneider University of Warwick, UK	Seeing Metabolism Clearly: Navigating Cross-Excitation Challenges in Multiplexed NADH FLIM.
12:20	Dr Joëlle Goulding University of Nottingham, UK	Utilising lifetime to understand membrane protein organisation.
12:45	Prof Amanda Wright University of Nottingham, UK	Optical imaging deep into biological samples.

13:10 Lunch / Poster Session / Sponsor tables

Event Programme

5 March, 2026

Day 2 Afternoon

14:10	**Data Analysis Session**	
14:25	**Cross-Topic Panel Discussion**	Dr Joëlle Goulding, University of Nottingham, UK Prof Marina Kuimova, Imperial College London, UK Prof Simon Ameer-Beg, King's College London, UK Dr Isabel Groß, PicoQuant GmbH Dr Sara Pellegrini, STMicroelectronics, UK Dr Alessandro Esposito, Brunel University of London, UK
15:25	**Closing remarks**	
15:45	**Departure**	

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Speaker Directory

Click here for full bio

Dr Thomas Blacker

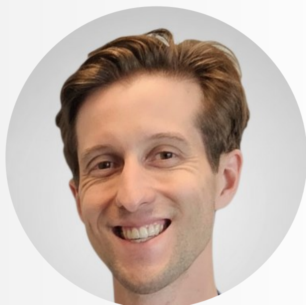
Lecturer in Cellular Biochemistry
UCL, UK

Prof Banafshé Larijani

Director of Consortium for Precision Health (CPH),
University of Bath, UK

Dr Gunnar Blume

Senior researcher
Ferdinand-Braun-Institut (FBH),
Germany



Dr Sylvain Boust

Project coordinator
EU HILIGHT project.
Programme lead,
III–V Lab, France

Dr Sara Pellegrini

Communication & Academic Collaborations Manager
STMicroelectronics, UK

Dr Leonardo Gasparini

Head of the Integrated Readout-ASICs & Image Sensors (IRIS) research unit
Fondazione Bruno Kessler (FBK), Italy

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Speaker Directory

Click here for full bio

Dr Claudio Bruschini

Lecturer and Scientist
AQUA Laboratory,
EPFL, Switzerland

Dr Isabel Groß

Sales and Application
Specialist for Microscopy
PicoQuant

Prof Gail McConnell

Professor of Biophotonics
Strathclyde Institute of Pharmacy
and Biomedical Sciences
University of Strathclyde, UK

Dr Simon Poland

Senior lecturer in Biophysics and
UKRI Future Leaders Fellow
Comprehensive Cancer Centre
King's College London, UK

Prof Simon Ameer-Beg

Professor of Optical Bioimaging
King's College London, UK

Speaker Directory

Click here for full bio

Prof Ruslan Dmitriev

Associate Professor
Ghent University,
Belgium

Dr Conor Treacy

Research Fellow
Brunel University of London, UK

Prof Marina Kuimova

Professor in Chemistry
Imperial College London, UK

Dr Falk Schneider

Assistant Professor
Centre for Mechanochemical Cell Biology
University of Warwick, UK

Dr Joëlle Goulding

Senior Research Fellow,
Centre for Membrane Proteins and Receptors (COMPARE)
University of Nottingham, UK

Prof Amanda Wright

Prof of Optics,
Optics and Photonics Research Group,
Faculty of Engineering,
University of Nottingham, UK

HLight

Flash Talks

4 March, 2026

Session I *(at 11:30)*

Ani Jose
Richard Dimbleby Cancer Research Laboratory
King's College London, UK

Kay Polland
Department of Physics, SUPA
University of Strathclyde, Glasgow, UK

Gunnar Blume
Ferdinand-Braun-Institut (FBH), Germany

Zhenya Zang
School of Engineering
University of Edinburgh, UK

Session II *(at 12:45)*

Liam Collard
Comprehensive Cancer Centre,
King's College London, UK

Narain Karedla
Botnar Institute for Musculoskeletal Sciences
University of Oxford, UK

The Rosalind Franklin Institute
Harwell Campus, Didcot, UK

Tommy Pallett
Comprehensive Cancer Centre,
King's College London, UK

Dinesh Beniwal
Comprehensive Cancer Centre
King's College London, UK

Poster Abstracts

1. Adding the Fluorescence Lifetime Dimension to Single-molecule Localization Microscopy with the Confocal Microscope Luminosa.
2. Fast and Sensitive FRET-FLIM in TIRF Using a 3D-Stacked Backside-Illuminated SPAD Array.
3. A 100x100 digital SiPM for high-speed FLIM.
4. Compact Quantum Light Source for Mid-Infrared Spectroscopy with "undetected" photons.
5. Tomographic, multiphoton, fluorescence lifetime imaging.
6. Development of fast single-shot FLIM for digital histopathology.
7. Versatile laser diode for 2ph FLIM at 8 Mpx/s in HILIGHT project.
8. Parallelised Approaches for High-Speed and High-Throughput FLIM.
9. Structure and dynamics of molecules and membranes using Metal-Induced Energy Transfer (MIET).
10. Using FLIM to probe membrane tension and Rho GTPase dynamics during cell migration and division.
11. Advanced FLIM Techniques Reveal Metabolic Insights in HER2+ Breast Cancer Cell Models.
12. Classifying the metabolic profiles of cancer cell populations using autofluorescence lifetime imaging and computer vision.
13. Second harmonic generation imaging for the assessment of collagen as a marker for tonsillar diseases.
14. Histogramless, Bandwidth-Efficient SPAD Array–Based FLIM Imaging.
15. Development of a fast two-photon FLIM microscope for practical intraoperative use.
16. FRET-Cascade FLIM for multicomponent interaction and force sensing in focal adhesions.

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P1

Adding the Fluorescence Lifetime Dimension to Single-molecule Localization Microscopy with the Confocal Microscope Luminosa

Isabel Groß[1], *Evangelos Sisamakis*[1], *Maria Loidolt-Krüger*[1], *Samrat Basak*[2], *Fabio Barachati*[1], *Roman Tsukanov*[2], *Oleksii Nevskyi*[2], *Cecilia Zaza*[3], *Germán Chiarelli*[3], *Guillermo Acuna*[3], *Jörg Enderlein*[2], *Rainer Erdmann*[1]

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[2] *University of Göttingen, Third Institute of Physics, Friedrich-Hund-Platz 1, 37077 Göttingen*
[3] *University of Fribourg, Department of Physics, Ch. Du Musée 3, 1700 Fribourg*

Confocal fluorescence microscopy is an essential tool in many research disciplines, particularly in the life sciences. Its axial sectioning capability is particularly useful for imaging thick samples. Additionally, it can be upgraded to detect fluorescence lifetimes, which can facilitate e.g. multiplexing, environmental sensing, or FLIM-FRET imaging. However, all images have a limited spatial resolution due to diffraction.

Single-molecule localization microscopy (SMLM) approaches such as PAINT or STORM enable fluorescence imaging with spatial super-resolution. But they are usually implemented on camera-based widefield microscopes, which do not support photon counting-based lifetime imaging.

The aim of this research was to add fluorescence lifetime contrast to super-resolution imaging and provide confocal sectioning ability. The fluorescence lifetime information should be exploited either for multiplexing, complementary to spectral approaches, or for providing enhanced axial resolution via MIET-SMLM, or for FRET imaging. To this end, DNA-PAINT and dSTORM image acquisition were implemented on our time-resolved confocal microscope Luminosa.

For PAINT, the acquisition time per image frame needed to match the binding kinetics of the DNA-PAINT imager strands, such that it was significantly smaller than the average binding time. For dSTORM, the acquisition time should be tuned according to the blinking kinetics of the fluorophore. Furthermore, an autofocus proved necessary for the lengthy acquisition of a sufficient number of frames, and stable temperature to minimize lateral drift. The resulting confocal FLIM movies were analyzed similarly as for SMLM measurements to obtain a final super-resolved image. Lastly, the photon arrival times of each single-molecule event were retrieved and analyzed to get the lifetime contrast.

For 3D super-resolution imaging with MIET-PAINT, the lifetime values were converted to axial positions using the MIET lifetime-distance dependency calculated based on the particular fluorophore properties. For FRET-PAINT, the lifetime was used to detect FRET events.

In conclusion, we demonstrated super-resolved FLIM with PAINT and dSTORM, MIET-PAINT to enable 3D super-resolution as well as super-resolved FRET-PAINT. Importantly, these cutting-edge imaging approaches were realized on a commercially available microscope. Luminosa's combination of stability, imaging speed, sensitivity and precision in lifetime determination facilitates the easy adaptation of state-of-the-art super-resolution imaging methodologies by other research groups.

P2

Fast and Sensitive FRET-FLIM in TIRF Using a 3D-Stacked Backside-Illuminated SPAD Array

D. Beniwal[1], T. Pallett[1], A. Erdogan[2], R. Henderson[2], S. Ameer-Beg[1]

[1]Comprehensive Cancer Centre, King's College London, UK; [2]Scottish Microelectronics Centre, The University of Edinburgh

High-resolution fluorescence imaging in biological systems is often hindered by variability in fluorophore concentration, which can arise from differences in expression levels or transfection efficiency. Fluorescence lifetime imaging microscopy (FLIM) overcomes this limitation because lifetime measurements are inherently independent of fluorescence intensity, making FLIM a powerful approach for quantifying molecular interactions and investigating cellular function. When paired with Förster resonance energy transfer (FRET), FLIM becomes especially valuable, as it reports on interactions at nanometre length scales and enables the study of dynamic molecular processes. However, conventional time-correlated single-photon counting (TCSPC) approaches used in FLIM require large photon numbers to accurately reconstruct fluorescence decay curves, leading to compromises between imaging speed and temporal precision.

In this work, we address these limitations by integrating a newly developed time-gated single-photon avalanche diode (SPAD) array, the ATLAS system, into a custom total internal reflection fluorescence lifetime imaging microscopy (TIR-FLIM) platform. This combination enables fast, sensitive, and high-resolution lifetime imaging. The ATLAS SPAD sensor consists of a 512 × 512 pixel array with a 100% fill factor, delivering a photon detection efficiency of 55% and supporting photon count rates up to 32 Mcps. Its time-gating capability provides gate widths with 11–33 ps resolution, while a rolling-shutter acquisition mode operating at 1.2 kfps with 22-bit photon counting allows rapid data collection. Together, these characteristics enable high-speed FLIM acquisition without sacrificing spatial or temporal detail, which is particularly advantageous for live-cell studies.

Using this TIR-FLIM system, we investigated FRET-based biosensors to examine the organization of the actin cytoskeleton near focal adhesions under different mechanical conditions. The intrinsic advantages of TIRF illumination, such as reduced background fluorescence and improved axial confinement, enhanced our ability to detect subtle lifetime variations. These measurements revealed clear differences in molecular organization at the cell membrane, highlighting the capability of the system to probe nanoscale dynamics in living cells.

This development opens new opportunities for applications including super-resolution FRET-FLIM, single-particle tracking FLIM, and high-speed live-cell imaging. By combining structural imaging with quantitative lifetime information, this approach offers a powerful means to explore how molecular organization relates to cellular function.

P3

A 100x100 digital SiPM for high-speed FLIM

A. Tontini[1], G. Maddinelli[1], and L. Gasparini[1]

[1]Center for Sensors and Devices, Fondazione Bruno Kessler, Trento, Italy

Fluorescence Lifetime Imaging Microscopy (FLIM) is a powerful technique for biological imaging, providing functional information by analysing fluorescence decay. However, conventional lifetime measurement approaches often face limitations in scalability, acquisition speed, and data handling.

In this work, we present a CMOS sensor based on Single-Photon Avalanche Diodes (SPADs) for high-speed FLIM applications. The proposed solution exploits single-photon detection and on-chip processing to enable efficient fluorescence lifetime measurements with reduced acquisition complexity. The architecture is designed to support fast measurements while maintaining high temporal resolution and low photobleaching.

By integrating local memory and parallel signal processing, the sensor allows effective photon accumulation and real-time data management. This approach enables flexible imaging modalities, including both lifetime and intensity measurements, while improving robustness against noise.

The results demonstrate the potential of SPAD-based imaging systems for fast, scalable, and low-impact FLIM applications. These technologies open new opportunities for real-time biological imaging and advanced biomedical diagnostics.

P4

Compact Quantum Light Source for Mid-Infrared Spectroscopy with "undetected" photons

G. Blume[1], P. Hildenstein[1], N. Werner[1], K. Bodenhagen[1], F. Mauerhoff[1], A. Sherwani[2], S. Ramelow[1,2,3] and K. Paschke[1]

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Sensing applications in the mid infrared (MIR) spectral range are challenging due to the high cost and low performance of the available light sources and detectors. This issue can be bypassed by utilizing the process of spontaneous parametric down-conversion (SPDC) to generate tailored MIR-NIR photon-pairs. The nature of SPDC enforces the generation of frequency-entanglement between these paired photons, which can be used for spectroscopy with "undetected photons". Using a nonlinear interferometric configuration, the MIR radiation is employed for the illumination and sensing of a specific material, whereas the NIR radiation is analysed using a conventional spectrometer [1]. This technique, which is referred to as "sensing with undetected photons", relies on table-top optical setups that are fragile, spacious and immobile, hence miniaturization is highly desirable [2].

Here we present the design, manufacturing and application of a miniaturized quantum light module, with a footprint of less than 80 x 60 mm^2, which is powered by a purpose-built diode laser at 720 nm [3]. The module contains a nonlinear crystal as the quantum light source, the interferometric reference arm for the NIR beam and mechanical connectors for the external MIR sensing arm. It also provides an interconnection to a spectrometer for the information-carrying NIR photons. This study presents insights into the optical design of the module, its manufacturing and the nonlinear optics quantum light source. The module emits a MIR-NIR photon-pair rate of about 109 s-1 W-1, which allows for the implementation of efficient real-time sensing applications. A test with a mirror in the MIR path showed a visibility of up to 30%. The MIR wavelengths near 3.4 µm are designed to match the absorption of polymers in aqueous solutions and aims at the detection of micro-plastics in wastewater in the field using the module in a mobile device. Potentially, the miniaturization of the sensing with undetected photons will enable a variety of other MIR-sensing applications.

References

[1] A. Vanselow et al. "Ultra-broadband SPDC for spectrally far separated photon pairs," Opt. Lett. 44, 4638-4641 (2019).
[2] E. Pearce et al. "Practical quantum imaging with undetected photons," Optics Continuum 2 (11), 2386-2397 (2023).
[3] F. Mauerhoff et al. „GaAs based edge emitters at 626 nm, 725 nm and 1180 nm" IEEE JSTQE 31, 1500710 (2025).

HLight

P5

Tomographic, multiphoton, fluorescence lifetime imaging

Liam Collard[1], Fahmida Rahman, Ani Augustine Jose[1], Tommy Pallet[1], Conor Treacy[1], Simon Ameer-Beg[1], and Simon Poland[1]

[1]Comprehensive Cancer Centre, King's College London, UK

Time-correlated single-photon counting (TCSPC) is an exceptionally precise technique to determine the fluorescence lifetime. By exciting a sample with pulsed laser light and measuring the arrival time of the fluorescent photons the lifetime may be determined. Whilst TCSPC is extremely accurate method for determining lifetime, it requires long exposure times often on the order of several minutes for a single image acquisition. To mitigate this, researchers have developed two main strategies aimed at accelerating lifetime imaging. i) Parallelization of the imaging readout. Whilst this has achieved improvement in imaging frame rates, SPAD arrays are themselves inefficient [1,2]. ii) Alternatively, compressive imaging techniques [3] can be used to accelerate lifetime imaging frame rates whilst using conventional "bucket" detection systems. Here, spatial patterns are encoded into the excitation light. The resultant image can then be recovered by applying an optimization algorithm.
In this work, we show how a compressive tomographic approach known as SLAP[4] may be used to accelerate FLIM imaging acquisition speeds. The sample is excited with a line beam projected at different angles and a Lucy-Richardson algorithm is then used to recover the image at each time gate recorded by the card. The system was used to topographically image beads with multiple fluorescent lifetimes, the tomographic projections are shown in 1b. The resulting Lucy Richardson reconstruction and a reference raster image are shown in panel c. The two images exhibited strong visual similarity both in intensity and lifetime mapping indicating the accuracy of tomographic FLIM in imaging sparse samples. Ultimately, the tomographic approach is shown to yield accurate lifetime and intensity reconstruction at less than 5% of the equivalent raster scanning acquisition time.

P5 *(continued)*

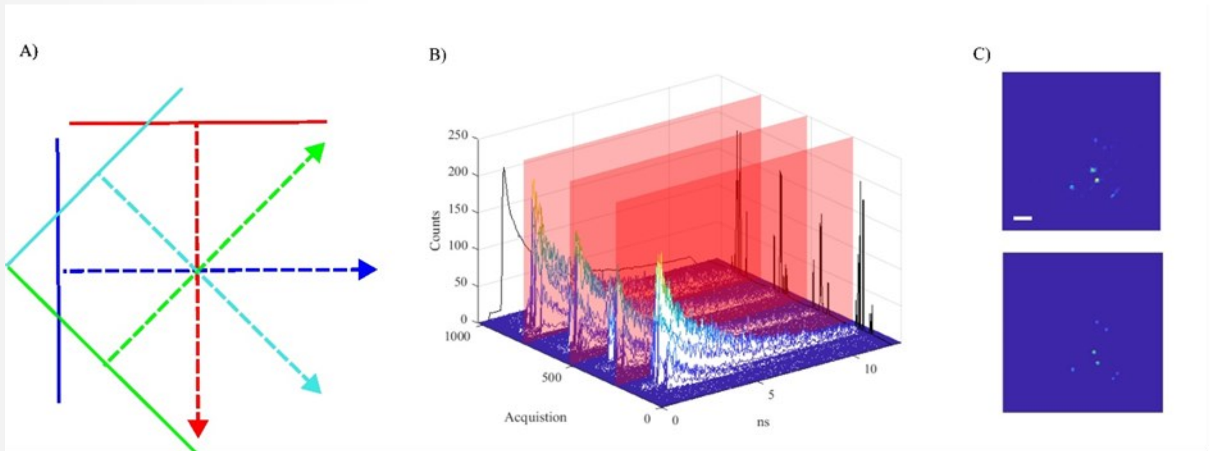

Figure 1 A) The principle of tomographic projection for a four angle projection. B) Example tomographic/lifetime datasets, the average lifetime trace and intensity projection are shown in black. Red planes indicate a change in angle. C)Preliminary intensity tomographic (top) and raster images (bottom) of beads.

References:

1 Poland SP, Krstajić N, Monypenny J, Coelho S, Tyndall D, Walker RJ *et al.* A high speed multifocal multiphoton fluorescence lifetime imaging microscope for live-cell FRET imaging. *Biomed Opt Express* 2015; **6**: 277–296.

2 Mai H, Jarman A, Erdogan AT, Treacy C, Finlayson N, Henderson RK *et al.* Development of a high-speed line-scanning fluorescence lifetime imaging microscope for biological imaging. *Opt Lett* 2023; **48**: 2042–2045.

3 Ghezzi A, Lenz AJM, Soldevila F, Tajahuerce E, Vurro V, Bassi A *et al.* Computational based time-resolved multispectral fluorescence microscopy. *APL Photonics* 2023; **8**: 46110.

4 Kazemipour A, Novak O, Flickinger D, Marvin JS, Abdelfattah AS, King J

P6

Development of fast single-shot FLIM for digital histopathology

Conor A. Treacy[1], Alessandro Tontini[2], Leonardo Gasparini[2], and Alessandro Esposito[1].

[1]Centre for Genome Engineering and Maintenance, Brunel University of London, Uxbridge, UK.
[2]Fondazione Bruno Kessler (FBK), Trento, Italy

Two-photon excitation and fluorescence lifetime imaging microscopy (FLIM) have a significant untapped potential for clinical applications[1-4]. Despite significant progress in FLIM acquisition speed and the availability of suitable two-photon excitation sources, important constraints remain that limit the usefulness of two-photon FLIM for enhancing contrast in digital histopathology. In this work, we aim to achieve sub-micron lifetime imaging over ~1 cm^2 of tissue with effective acquisition rates of **~8 × 10^6 pixels/s** (≈1 minute per cm^2), with minimal loss of resolution.

To overcome existing limitations, we are developing new sensors, laser technologies, and scanning architectures as part of the EU HORIZON HiLIGHT consortium (https://hilighthorizon.eu), aiming to achieve target forward-scan or peak pixel rates of **25 × 10^6 pixels/s**, with fluorescence decay detected using single-pulse or burst-mode excitation schemes.

Here, we show preliminary results on the generation of digital H&E-like staining[5-7] using the clinically approved Acridine Orange probe. Acridine Orange is a photophysically complex probe that binds nucleic acids. It is already used in digital histopathology[6,8] to stain cell nuclei. Using three-dimensional cultures of pancreatic cells, we will show that two-photon FLIM can provide information on chromatin compaction in complex 3D environments that recapitulate tissue sections.

We also show the very first images acquired with a point detector formed by a large time-resolved digital silicon photomultiplier (dSiPM) array, designed to support aggregate photon count rates exceeding **2.5 × 10^{11} photons/s**. Together with the development of new acquisition schemes and compact solid-state lasers, we aim to overcome remaining engineering obstacles to fast FLIM, approach the physical acquisition speed limits, and enable its applications in digital histopathology.

References:

1 David, W. P., David, R. S. & Watt, W. W. in *Proc.SPIE.* 379-389.
2 So, P. T. C. *et al.* Time-resolved fluorescence microscopy using two-photon excitation. *Bioimaging* **3**, 49-63 (1995).
3 Peter, M. & Ameer-Beg, S. M. Imaging molecular interactions by multiphoton FLIM. *Biol Cell* **96**, 231-236 (2004).
4 König, K., Uchugonova, A. & Gorjup, E. Multiphoton fluorescence lifetime imaging of 3D-stem cell spheroids during differentiation. *Microscopy Research and Technique* **74**, 9-17 (2011).
5 Esposito, A. & Venkitaraman, A. R. Enhancing Biochemical Resolution by Hyperdimensional Imaging Microscopy. *Biophys J* **116**, 1815-1822 (2019).

P7

Versatile laser diode for 2ph FLIM at 8 Mpx/s in HILIGHT project

S. Boust[1], D. Boiko[2].

[1]III-V Lab, Palaiseau, France.
[2]CSEM SA, Neuchâtel, Switzerland.

Progress in time-critical applications for biomedical research and intraoperative diagnostics is often constrained by the performance of available light sources. Conventional histopathology may require several days, whereas surgeons rely on rapid feedback to assess tumor resection margins. Fast two-photon fluorescence lifetime imaging microscopy (2ph-FLIM) has the potential to deliver real-time digital histopathology at depth, but current implementations remain too slow. Most systems employ low-repetition-rate mode-locked lasers and detect, on average, only a single fluorescent photon per excitation pulse, which fundamentally limits acquisition speed.

Within the HILIGHT project, we are developing a novel semiconductor-based laser source capable of generating picosecond pulse bursts at gigahertz repetition rates. This architecture provides a compact footprint suitable for microscope integration and enables the high photon throughput required for rapid FLIM.

In this work, we report our progress toward realizing this high-speed excitation approach and present the laser design and its optimization for achieving 2ph-FLIM imaging at 8 Mpx/s. We show the first experimental characterization results, including measurements of average and peak optical power, optical spectra, and RF spectral properties.

P8

Parallelised Approaches for High-Speed and High-Throughput FLIM

Ani Jose[1], Conor A. Treacy[1], Anneliese Jarman[1], Hanning Mai[1], Ahmet Erdogan[2], Robert Henderson[2], Sofia Fredin[3], Thomas Kavanagh[1], Andrea Serio[3], Simon Poland[1], and Simon Ameer-Beg[1]

[1] Richard Dimbleby Cancer Research Laboratory, King's College London, UK

[2] Institute for Integrated Micro and Nano Systems, University of Edinburgh, UK

[3] The Francis Crick Institute, London, UK

Fluorescence lifetime imaging (FLIM) provides concentration-independent contrast suitable for probing molecular environment and biochemical state. However, conventional raster-scanned implementations rely on sequential acquisition, limiting imaging speed and restricting throughput in dynamic and population-scale studies. Here, we present two FLIM architectures that remove this sequential bottleneck in both microscopy and cytometry.

The first platform, SWARM (SWept ARray Microscope), employs a 32 × 32 SPAD detector array combined with a diffractive optical element to generate 1024 simultaneous excitation foci [1]. This multibeam design parallelises acquisition, enabling Nyquist-sampled FLIM over large fields of view while reducing excitation dose per pixel. We demonstrate high-speed lifetime imaging in live cells across different biological systems, including monitoring ATP:ADP dynamics using the genetically encoded biosensor PercevalHR. The system resolves fast metabolic responses and longer-term cellular state transitions, enabling time-resolved imaging of highly dynamic biological processes in live cells.

The second platform extends FLIM into the cytometry domain using a CMOS SPAD line sensor with integrated time-to-digital conversion and on-chip histogramming [2]. A diffraction-limited excitation line is aligned across a hydrodynamically focused microfluidic channel, with cell motion providing the scan dimension for image reconstruction. Functional lifetime imaging was demonstrated in live cells under continuous flow, including imaging of cells expressing different fluorescent proteins and lifetime multiplexing of spectrally overlapping dyes.

Together, these approaches overcome speed and throughput constraints inherent to raster scanned implementations of FLIM, enabling rapid dynamic imaging and quantitative lifetime analysis across large cell populations.

References:

[1] Ameer-Beg, S. Poland, S., Levitt, J. & Nedbal, J. (2017) UK Patent Application No. 1710743.4
[2] T. Ahmet, R. Walker, N. Finlayson, N. Krstajic, G. Williams, J. Girkin, and R. Henderson, IEEE J. Solid-State Circuits **54**, 1705 (2019)

HLight

P9

Structure and dynamics of molecules and membranes using Metal-Induced Energy Transfer (MIET)

Narain Karedla[1,4], Tao Chen[2], Arindam Ghosh[5], Joerg Enderlein[2,3]

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[3] Cluster of Excellence "Multiscale Bioimaging: from Molecular Machines to Networks of Excitable Cells" (MBExC), Universitätsmedizin Göttingen, Robert-Koch-Str. 40, Göttingen, 37075, Germany
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Metal / Graphene-Induced Energy Transfer (MIET/GIET) leverages the near-field coupling of an excited fluorophore to the electromagnetic modes of a thin metal film to convert fluorescence lifetime into an absolute, nanometre-scale readout of axial distance from the surface. This enables localizing standard single fluorescent molecules and even fluorescent proteins with a range of up to 200 nm from the surface using a standard fluorescence-lifetime imaging microscope [1, 2]. We combined MIET with Fluorescence Correlation Spectroscopy (FCS) to simultaneously determine bilayer structure, such as bilayer and hydration layer thickness, and leaflet-specific lipid diffusion in solid- and polymer-supported lipid bilayers [3]. We further quantify rapid, nanometric out-of-plane membrane fluctuations, providing a direct readout of membrane tension and surface interaction strength [4]. Extending the same axial ruler to membrane–membrane contacts provides a direct readout of intermembrane spacing during adhesion, resolving distinct adhesion states and short-lived intermediates in model E-cadherin junctions and thereby connecting molecular binding pathways to emergent membrane mechanics [5]. Finally, we introduce dynamic MIET spectroscopy (dynaMIET), which uses MIET-FCS to resolve three-dimensional membrane dynamics with nanometer axial sensitivity and microsecond temporal resolution within a single acquisition, enabling concurrent quantification of lateral diffusion and vertical undulations of molecules in living cells across the plasma membrane and internal organelles [6].

References:

[1] Karedla *et al.*, ChemPhysChem 2014. [2] Hauke *et al.*, ACS Nano 2023.
[3] Karedla *et al.*, Angewandte Chemie 2025.
[4] Chen *et al.*, Nat. Commun. 2024
[5] Chen *et al.*, Nat. Commun. Bio. 2024.
[6] Karedla *et al.* (under revision).

P10

Using FLIM to probe membrane tension and Rho GTPase dynamics during cell migration and division

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Abstract: Both cell migration and division are key physiological processes which underpin development, wound healing, tissue regeneration and drive metastasis and proliferation of cancer. Cell migration requires polarisation and local protrusions via actin polymerisation at the leading edge and contractility at the rear. Cell division meanwhile requires more global contractility as cells round up to enter mitosis and remain round while chromosomes separate. I have found that these cell remodelling events through division and migration are orchestrated by both membrane biophysical properties and Rho GTPase signalling. Low membrane tension provides spatial signals in fast moving cells, through the formation of caveolae, to control the assembly of F-actin and actomyosin contractility at the rear via RhoA activation (Hetmanski et al., 2019), while similar 'hot spots' of low tension exist along the membrane as cells round up and maintain roundness prior to furrow formation, and I hypothesise that these instigate signals via RhoA which enhance the fidelity of successful division. While membrane tension has been traditionally hard to measure, especially in 3D matrix which is inaccessible by 'gold standard' optical bead pulling approaches, I have extensively used live FLIM of the membrane tension reporter Flipper TR (Colom et al., 2018) to measure and analyse local fluctuations in membrane properties. Similarly, as Rho GTPases are switch like proteins controlled by the balance of activators and inhibitors and mere localisation imaging does not provide an accurate read out of activity, I have use FRET and FLIM of RhoA biosensors (Yoshizaki et al., 2003) to assess the spatiotemporal dynamics of proteins such as RhoA. Altogether, FLIM of biosensors has been a vital tool to uncover important drivers of both cell migration and cell division.

References:

Hetmanski, Joseph HR, et al. "Membrane tension orchestrates rear retraction in matrix-directed cell migration." Developmental cell 51.4 (2019): 460-475.

Colom, Adai, et al. "A fluorescent membrane tension probe." Nature chemistry 10.11 (2018): 1118-1125.

Yoshizaki, Hisayoshi, et al. "Activity of Rho-family GTPases during cell division as visualized with FRET-based probes." The Journal of cell biology 162.2 (2003): 223-232.

HLight

P11

Advanced FLIM Techniques Reveal Metabolic Insights in HER2+ Breast Cancer Cell Models

Joseph Navaratne, Tommy L. Pallett, Conor A. Treacy, Margot Kapetan, Simon P. Poland, Simon M. Ameer-Beg

Comprehensive Cancer Centre, School of Cancer and Pharmaceutical Imaging, King's College London, Guy's Campus, London SE1 1UL.

Understanding cellular metabolism at the nanoscale is crucial for deciphering cancer progression and therapeutic resistance. My research employs advanced fluorescence lifetime imaging microscopy (FLIM) to non-invasively probe metabolic activity in HER2+ breast cancer cells. By integrating autofluorescence lifetime imaging of endogenous cofactors (NAD(P)H and FAD) with genetically encoded FRET-based biosensors, we establish a spatially resolved, multiplexed framework for quantifying metabolic phenotypes in live cells. This dual approach enables us to correlate intracellular metabolic signatures with microenvironmental changes, offering a powerful platform for studying dynamic processes at subcellular resolution.

Our current focus involves 2D monolayer models, with future work extending to 3D spheroids to better mimic tumour architecture. This methodology reveals heterogeneous metabolic states within cancer populations, highlighting potential vulnerabilities for therapeutic targeting.

I am particularly interested in the potential of nanochemistry to refine biosensor design, improve imaging precision, and enhance drug delivery. Attending the Doctoral Nanochemistry Camp would provide an outstanding opportunity to engage with pioneers in nanoscale science and apply emerging insights to cancer biology. I am excited by the prospect of contributing to interdisciplinary dialogue and advancing the interface between nanochemistry, imaging, and biomedicine.

P12

Classifying the metabolic profiles of cancer cell populations using autofluorescence lifetime imaging and computer vision

Rogelio Garcia-Aguirre[1,†], Tommy Pallett[2,†], Tony Poon[2], James Moneypenny[2], Simon Ameer-Beg[2], Siân Culley[1]

[1] Randall Centre for Cell and Molecular Biophysics, King's College London, UK.
[2] Comprehensive Cancer Centre, King's College London, UK.

Antibody-drug conjugates (ADCs) have improved outcomes for breast cancer, but drug resistance remains a major challenge[1]. Identifying cancer cell subpopulations that respond to drugs or have upfront resistance would facilitate personalised treatment. Using 2-photon autofluorescence lifetime imaging of NAD(P)H, a well-established metabolic sensing method[2], we assessed whether metabolic profiling could classify ADC- responsive or resistant cell populations. Interpretation of autofluorescence data is challenging because concentration and lifetime vary across different cellular compartments, and NAD(P)H lifetime changes both with phosphorylation and upon binding to other molecules. Recent attempts to decipher these shifts[3] require exceptional temporal resolution, high signal-to-noise ratios, or large datasets: limitations in biological and clinical contexts. Conventional metrics such as weighted-average fluorescence lifetime and lifetime-weighted quantum yield, or phasor representations, can indicate altered metabolic state and guide investigation. Yet interpreting lifetime distributions in a biologically meaningful way is challenging, and classifying cell populations to a clinical standard, let alone individual cells, is currently impossible. Deep learning models excel at classification, and tolerate noisy data, but typically require hundreds of thousands of data entries for effective training. Since our application is characterised by limited datasets, we repurposed a pretrained computer vision model[4] using data augmentation and transfer learning. We introduced an encoder prior to the pretrained models which leverages the high-dimensional FLIM data to learn an optimal representation in the three-dimensional colour space of natural images, on which computer vision models are trained. We validated performance and determined the information required to discriminate image patches containing responsive or resistant cells. We discovered that raw fluorescence time-domain data was not only sufficient for classification, but out-performed other parameterised inputs, indicating that conventional data fitting could introduce bias. This new approach could enable identification of phenotypic variation within diverse populations of cancer cells in situ and offer the prospect of efficient treatment monitoring and adaptation.

P12 *(continued)*

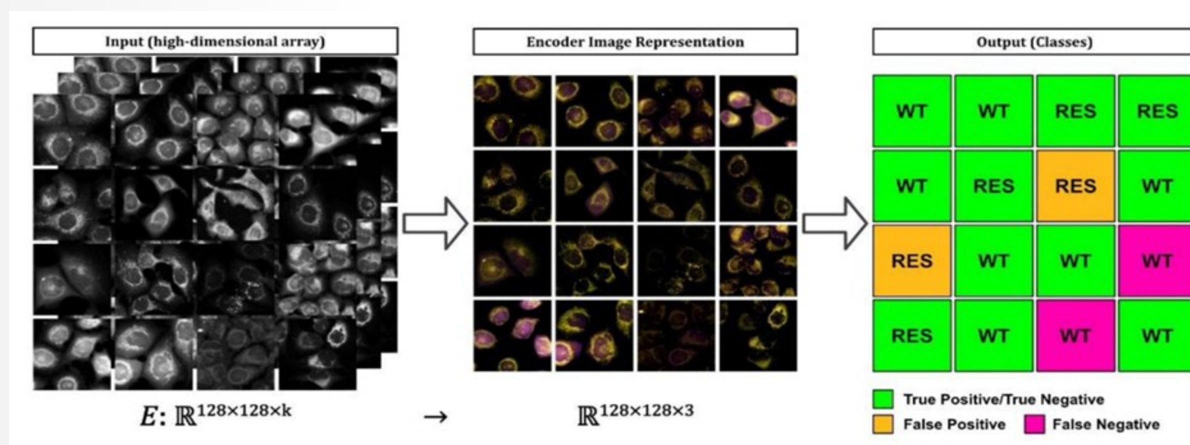

Fig. 1: An example of the classification of randomly selected and unseen patches of wild-type (WT) and ADC resistant (RES) cell populations. The encoder block receives k-dimensional arrays of 128x128 pixel raw image data as an input and maps them to 3-channel (colour) image space before the pretrained computer vision model is applied.

References:

[1] Chen, Y. et al., *Cancer Commun.* (2023)
[2] Kolenc, O. & Quinn, K. *Antioxid. Redox Signal.* (2019)
[3] Leben, R. et al., *Int. J. Mol. Sci.* (2019)
[4] Liu, Z. et al., *IEEE/CVF conference on Computer Vision and Pattern Recognition*, New Orleans, USA (2022)

P13

Second harmonic generation imaging for the assessment of collagen as a marker for tonsillar diseases

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[2]Department of Otolaryngology – Head and Neck Surgery, Royal Hospital for Children, Glasgow, UK
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Contact Name: Kay Polland, University of Strathclyde, kay.polland@strath.ac.uk

Acute recurrent tonsillitis (ART) and obstructive sleep apnoea (OSA) are the most common reasons for tonsillectomy worldwide with inflammation being one of the leading symptoms, however there is no standardised diagnostic test to differentiate between disease types. Chronic inflammation can lead to an overgrowth of extracellular matrix proteins, such as collagen, promoting disease development and causing decreased immunological function. We hypothesised that collagen distribution would differ between patients with ART and OSA and have performed label-free second harmonic generation (SHG) imaging of tonsil tissue from patients with ART and OSA to enable measurement of the relative abundance of immature and mature type I collagen within *ex vivo* paediatric tonsils. SHG is a non-linear optical process in which two incident photons of a single frequency are destroyed and a single photon is produced with double the initial frequency. By generating forward and backwards SHG signal the mature and immature type I collagen within the palatine tonsil can be imaged respectively. Representative images for each disease type can be seen below in figure 1.

P13 *(continued)*

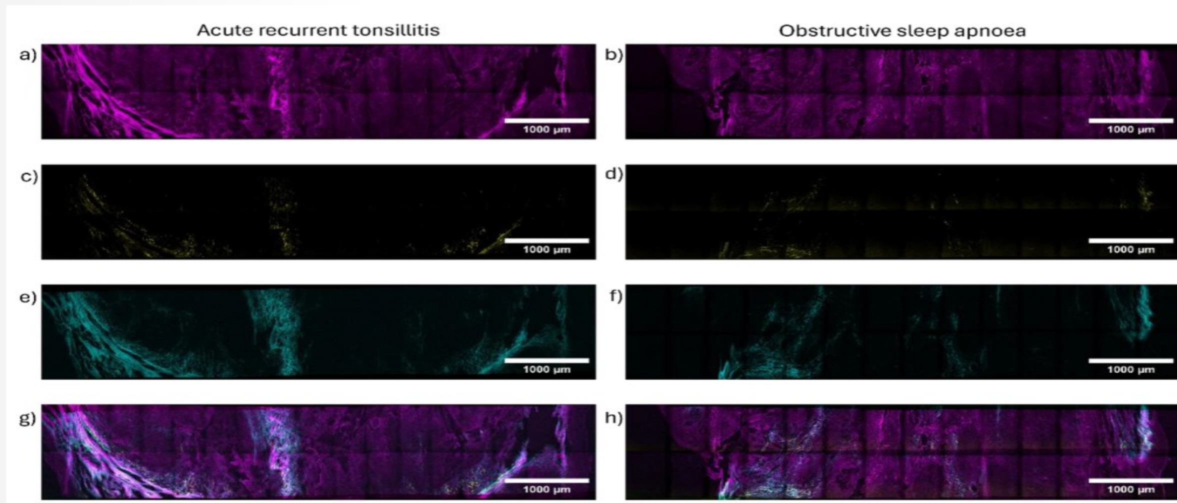

Figure 1: Imaged regions of interest in tonsil tissue from patients with ART and OSA. (a) and (b) show images of tissue autofluorescence (magenta) from OSA and ART respectively. SHG images of type I mature collagen (yellow) distribution in tissue from patients with ART and OSA are shown in (c) and (d), while SHG images of type I immature collagen (cyan) are shown in (e) and (f) for the same diseases. In (g) and (f), false-colour merge images of (a), (c), and (e) and (b), (d) and (f) respectively are presented, showing the location of type I mature and type I immature collagen in the tonsil tissue. These images have been adjusted using CLAHE within Fiji for presentation purposes. Scale bars = 1 mm in all images

We show that while there was no statistically significant difference in the amount of mature Type I collagen in patients with ART compared to those with OSA ($p = 0.61$), a difference in the amount of immature Type I collagen was found to be statistically significantly higher ($p = 0.0059$) in patients with ART than those with OSA. This work implies that the extracellular matrix may be used as a marker for disease type for more accurate diagnosis of tonsillar disease. Furthermore, due to detection of immature type I collagen being in the backwards direction it is possible that preexisting optical technologies, such as SHG endoscopes, could be adapted or repurposed to diagnose disease type *in vivo*.

HLight

P14

Histogramless, Bandwidth-Efficient SPAD Array–Based FLIM Imaging

Zhenya Zang[1], Mike Davies[1], and Istvan Gyongy[1]

[1]School of Engineering, University of Edinburgh

This post presents a compression algorithm for single-photon avalanche diode (SPAD) array–based, time-domain wide-field fluorescence lifetime imaging (FLIM). Emerging high–spatial-resolution SPAD arrays offer outstanding parallel data-acquisition speeds (up to 15 GB/s) across arrayed pixels, compared with scanning-based confocal FLIM systems, enabling fast imaging and high throughput. However, as the spatial resolution of SPAD arrays increases, the resulting data throughput grows exponentially, quickly exceeding the bandwidth limits of available communication protocols. Although existing on-chip histogramming and computational strategies can partially mitigate this issue, they require considerable on-chip hardware overhead. Our statistics-based compression algorithm [1], termed sketched FLIM, uses spline functions to approximate massive time-stamp data in real time, alleviating the bandwidth bottleneck between the sensor and post-processing stages. The proposed method achieves a compression ratio exceeding 99% while maintaining high-fidelity post-processing performance. The algorithm has been validated using both synthetic and real datasets, demonstrating accurate lifetime reconstruction compared with conventional nonlinear least-squares fitting (NLSF) and maximum-likelihood estimation (MLE) methods that rely on full histograms. A firmware implementation of the sketch-based compression has been realized on a SPAD sensor (QuantiCAM [2]) firmware and will be further validated in a FLIM system.

References:

[1]. Sheehan, Michael P., Julián Tachella, and Mike E. Davies. "Spline sketches: an efficient approach for photon counting lidar." *IEEE Transactions on Computational Imaging* 10 (2024): 863-875.

[2]. Henderson, R. K., Johnston, N., Mattioli Della Rocca, F., Chen, H., Li, D. D.-U., Hungerford, G., Hirsch, R., Mcloskey, D., Yip, P., & Birch, D. J. S. (2019). A 192×128 Time Correlated SPAD Image Sensor in 40-nm CMOS Technology. *IEEE Journal of Solid-State Circuits*, *54*(7), 1907-1916. https://doi.org/10.1109/JSSC.2019.2905163

P15

Development of a fast two-photon FLIM microscope for practical intraoperative use

Philipp Andre[1], Zachary Baltzer[1], Bill Fox[1], Lucas Heinrich[1]

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Standard histopathological analysis requires fixation, embedding, sectioning, and H&E staining — a process that can take up to a full day. This delay limits the use of histopathology where it is needed most: in the operating theatre, for example during Mohs micrographic surgery, where the patient waits under anaesthesia while tissue samples are processed across multiple excision cycles. VivaScope's confocal laser scanning microscopes address this challenge through digital histopathology, generating H&E-like images computationally from fluorescence intensity and reflectance data — providing near-instantaneous optical biopsies at cellular resolution without physical staining. With over 1,300 systems installed worldwide across pathology, dermatology, urology, gynecology, and mammography, VivaScope's technology is established in clinical practice.

However, current systems are limited to single-photon confocal imaging with penetration depths of approximately 100 µm and no molecular specificity beyond fluorescence intensity. The EU Horizon Europe project HILIGHT (Grant Agreement No. 101135034) aims to overcome these limitations by upgrading the VivaScope platform with two-photon excitation and fluorescence lifetime imaging (FLIM) capabilities. Two-photon excitation using near-infrared light (880 nm) promises significantly deeper tissue penetration — up to 300 µm — due to reduced scattering in biological tissue. FLIM adds a new dimension of information by probing the cellular microenvironment: metabolic state, protein interactions, and tissue composition at the single-cell level.

The HILIGHT approach will provide the VivaScope platform with a cost-effective mode-locked semiconductor laser and a SPAD array detector with time-gated photon counting to develop the fastest two-photon FLIM microscope for practical intraoperative use.

P15

FRET-Cascade FLIM for multicomponent interaction and force sensing in focal adhesions

Conor A. Treacy[a,b,c], Tommy L. Pallett [b], Tam Bui[d], Simon P. Poland[b], Mark A. Pfuhl[a], Maddy Parsons[a] and Simon M. Ameer-Beg[b].

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[b]Comprehensive Cancer Centre, School of Cancer and Pharmaceutical Sciences, King's College London, UK.
[c]Centre for Health, Medicine and Life Sciences (CHMLS), Brunel University London, Uxbridge, UK.
[d]Institute of Pharmaceutical Science, School of Cancer and Pharmaceutical Sciences, King's College London, UK.

Förster resonance energy transfer (FRET) enables the measurement of molecular interactions and conformational dynamics in biological systems. FRET-Cascade, a multistep energy transfer system involving three fluorophores, enables spatial and temporal mapping of molecular interactions[1]. Here, we leveraged FRET-Cascade with time-correlated single photon counting multiphoton fluorescence lifetime imaging microscopy[2] (TCSPC-FLIM) to explore the putative interaction between Rap1-interacting Adapter Molecule (RIAM) and vinculin in focal adhesions. We developed a novel three-fluorophore FRET-Cascade system, validated using purified proteins, spectroscopic analysis, structural modelling, and negative-staining transmission electron microscopy (TEM). Putative RIAM-vinculin interactions were explored in vinculin knockout mouse embryonic fibroblasts, revealing RIAM binds to the N-terminus of vinculin in focal adhesions. Vinculin tension-sensing constructs report average forces of 3.0 ± 0.3 pN per focal adhesion, consistent with its role in mechano-transduction. This work establishes cascade FRET as a powerful approach for dissecting multicomponent protein interactions and force-sensing dynamics in live cells.

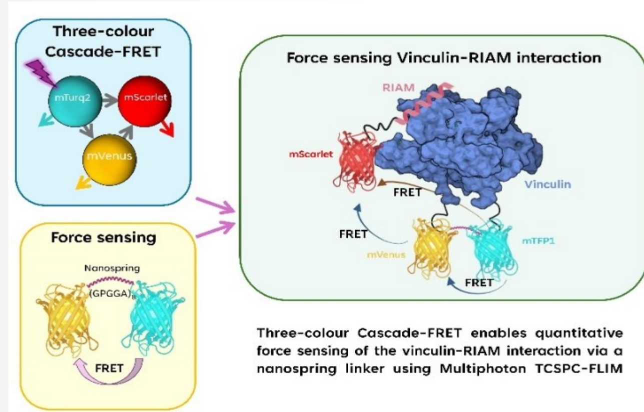

Figure 1 | Schematic of a three-colour FRET-Cascade FLIM approach for quantitative force sensing. A multistep FRET cascade between mTurquoise2, mVenus, and mScarlet is combined with a vinculin tension-sensing construct incorporating an extensible nanospring linker. Multiphoton TCSPC-FLIM enables lifetime-based detection of both molecular interactions and force-dependent conformational states within the vinculin–RIAM complex.

References:

1 Treacy, C. A. *et al.* Development of three-colour FRET cascade for force sensing of the putative RIAM-vinculin interaction in fibroblasts. *Commun Chem* (2025).

2 Peter, M. & Ameer-Beg, S. M. Imaging molecular interactions by multiphoton FLIM. *Biol Cell* 96, 231-236 (2004).

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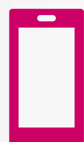

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- Santander Bike Station
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- Pay and Display Machine
- Academic Buildings
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- Recreational Buildings

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F4 Applicant Enquiries
G3 Advanced Metal Casting Centre (AMCC)
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E4 Arts Centre
D4 Bannerman Centre
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F3 Brunel Pathway College
B4 Chadwick
F4 Conference Office
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D4 Design and Print Service
B4 Distribution Centre
F4 Eastern Gateway
F3 Elliott Jaques
E3 Faraday Common Room
F3 Gardiner
B4 Gaskell
D4 Hamilton Centre
D5 Heinz Wolff
D3 Howell
F4 Indoor Athletics Centre
D2 Joseph Lowe
E5 Lancaster Suite
D4 Lecture Centre
D4 Library
C4 Marie Jahoda
F4 Mary Seacole
E3 **Medical Centre**
D4 Medical School
C4 Meeting House
D4 Michael Sterling
D4 Student Hub
D4 Multifaith Prayer Space
D4 Quad North
F4 **Reception**
F3 Russell
C4 **Security**
E4 Sports Centre
H1 Sports Pavilion
F5 St Johns
D3 Student Centre
F4 Student Living Office
D4 Student's Union
B4 Amenity Building and Student Store
D3 Tower A
D3 Tower B
D3 Tower C and D
C4 Wilfred Brown
D5 Wolfson Centre

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16 Galbraith Hall

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18 Meadow Hall
19 Michael Bevis Hall
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30 Shoreditch Hall
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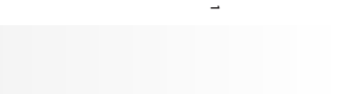

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M25
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HLight

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- Dr Alessandro Esposito
- Email: alessandro.esposito@brunel.ac.uk

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All reports will be handled confidentially.

The organisers may take appropriate action, including removal from the event.

TODAY'S SPECIAL:
FLIM & TONIC
- A BIO-OPTICAL ELIXIR.