



impetux
The Force of Light

SENSOCELL+

Optical tweezers for Cell and Molecular Biophysics

Confocal & Widefield
Microscope add-on
Calibration-free
Measure in cells
Active microrheology
Programmable



Microscopy integration

PhaseStreak Confocal

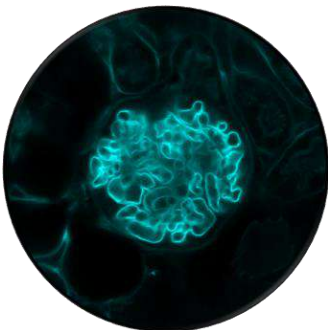
Solid-State Tech: No moving parts. No pinholes



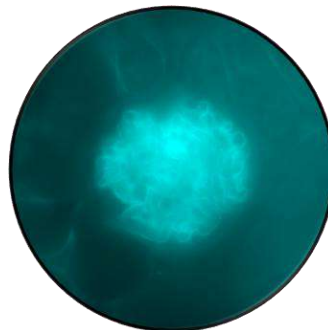
SENSOCELL can be upgraded with our [integrated high-speed PhaseStreak confocal](#) imaging module:

- [Confocal, Widefield and Brightfield](#) modes
- 23.1 mm FOV
- 230 fps acquisition
- [Deep imaging](#): >300 μm penetration capacity
- [Solid-state](#): AOD-based line scanner synchronized with sCMOS rolling shutter for out-of-focus filtering
- [Adjustable sectioning](#). Optimize it for each sample
- [Cell friendly](#). Distributed line excitation. Tunable StB
- 405, 473, 561, 639 nm excitation lines
- ROI illumination selection
- 1-click [instant switch](#) between modes

CONFOCAL



WIDEFIELD



550 μm cleared mouse kidney section. Alexa Fluor 488-WGA/blood vessels. Objective: Plan Apo 60x 1.2 NA WI

Turn-key system or add-on

Adapted to your lab needs



Fig. 1 A [SENSOCELL+](#) platform integrated with [PhaseStreak Confocal](#) installed on a Ti2-E motorized microscope with incubation chamber at the Technical University of Dresden, Germany (Jan Brugués Lab)

SENSOCELL is designed for manual or motorized [inverted microscopes](#) from the Nikon Ti and Ti2 family. It can be delivered as a turn-key system or add-on for [preexisting microscopes](#).

Microscopes can be [manual or fully motorized](#), including Autofocus Function (PFS). With a double-deck configuration, up to [4 additional ports are free](#) (1 fluorescence port, 2 lateral ports and 1 eyepiece port) are free for integrating [different camera and fluorescence imaging options](#).

Several camera models are available (Hamamatsu, Photometrics, Tucsen...) for SENSOCCELL allowing for [synchronized imaging and data acquisition](#).

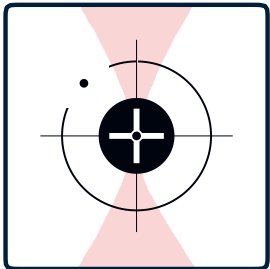
The system can be upgraded with [incubation chambers](#) for live cell experiments designed by Okolab for SENSOCCELL.

Direct force detection

Calibration-free force measurements on multiple traps



Force resolution	< 50 fN
Accuracy (typ. in cells)	< 10%
Precision (typ.)	< 2 %
Position resolution	< 1 nm



No calibrations needed



In cellulo and in vivo



Force clamp mode



Prevent cell damage

Our unique force sensor module operates by calibration-free detection of light momentum changes. This method is independent of the sample and medium conditions and **does not require any previous calibration** by the user prior to start measuring. The detector allows **simultaneous force measurements over multiple traps**.

The direct force detector allows **Intracellular measurements**, even inside living embryos and organisms using internalized microbeads as handles or directly trapping endogenous subcellular structures such as lipid droplets, vesicles, nuclei or protein condensates but also cell contacts or cells in suspension.

It also enables measuring forces on **arbitrarily-shaped objects** including active systems out of the equilibrium like **cells or rod-shaped bacteria**.

The sensor can work in particle tracking mode and continuously **monitors laser power** at the sample plane giving maximum control over the irradiation levels imposed on your samples.

A force feedback loop enables the **Force Clamp mode** to have absolute control of pulling and pushing force rates in creep-compliance tests.

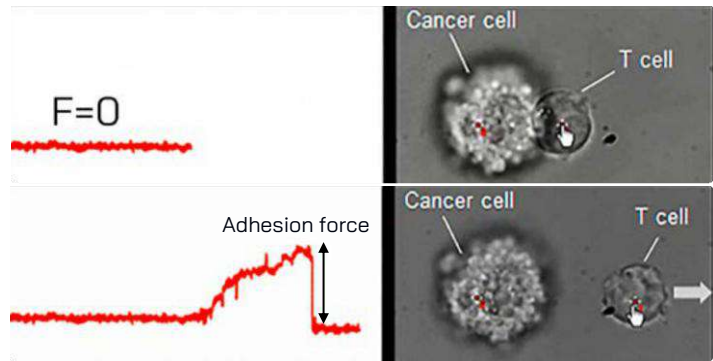


Fig. 1 Example of direct force detection enabling force measurements in complex samples. A T-cell is brought into contact with a cancer cell. Both cells are directly trapped using two optical traps. After a few seconds, the T-cell is pulled away while the adhesion force is recorded.

Optical manipulation

Accurate, extensive and flexible control over multiple traps



SENSOCCELL's optical manipulation module allows generating up to **256 independent simultaneous traps** over a working field of $70 \times 70 \mu\text{m}$ (for a $60\times$ objective). Based on acousto-optic deflection technology, the system allows trap steering at 25 kHz with $< 1 \text{ nm}$ trap position resolution. It includes an **ultra-low noise single frequency laser source** (1064 nm).

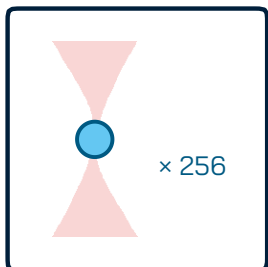
Our control **software suite LightACE** enables precise, extensive and flexible control over multiple traps via **automatized routines**. Apply predefined oscillations and/or trajectories over multiple traps or control them using the click & drag mode. Apply static (256 traps) or dynamic (32 traps) trap patterns. SENSOCCELL can be integrated with our **PhaseSreak Confocal** imaging unit enabling correlated fluorescence position and force data. The setup leaves free microscope lateral and Epi ports for third-party systems.

LightACE software

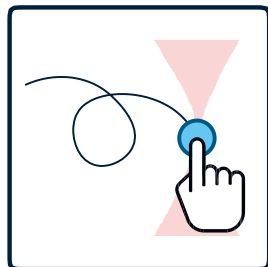


LightACE is our control software suite for SENSOCCELL integrated with μ -Manager. Easy and intuitive to work with, our LightACE software will allow you to **take control over multiple traps**.

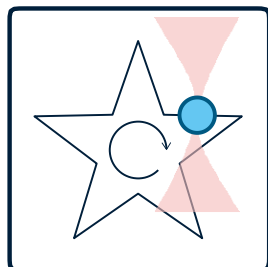
LightACE allows to visualize and store **real time force & position data** for each trapped target, apply force clamp or custom built-in routines with 1-click. By selecting the different options on the interface menu, LightACE offers a varied and flexible set of trapping, manipulation & measurement capabilities. Use our predefined automated routines or **create your own routines** with our Software Development Kit featuring a wide variety of examples.



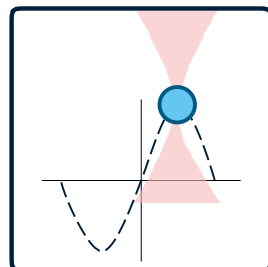
Up to 256 traps



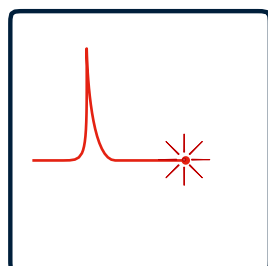
Click & drag mode



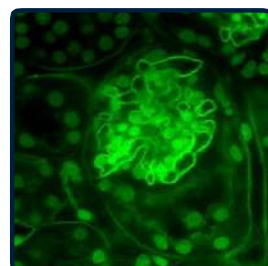
Trap trajectories



Trap oscillations

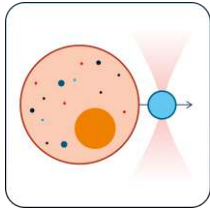


1064nm laser



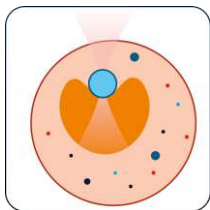
Confocal & Epi-FL

Applications



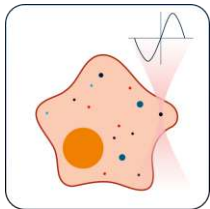
Plasma Membrane Mechanics

Use our automatized membrane tether pulling routine to study membrane flow, membrane tension propagation, membrane-to-cortex attachment and mechanotransduction through transmembrane proteins.



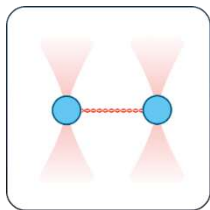
Subcellular Mechanics

Deform and probe intracellular organelles such as the cell nucleus or cytoplasm and measure their mechanical properties using internalized beads or directly trapping endogenous structures like lipid droplets in creep-compliance and stress relaxation tests.



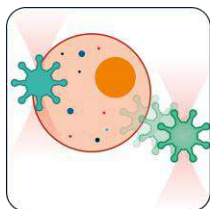
Micro-Rheology of Cells and Biomolecular Condensates

Use our automatized TimSOM Active micro-Rheology routine to get the full complex viscoelastic properties of different subcellular compartments or biomolecular condensates undergoing Liquid-Liquid Phase Separation (LLPS) to monitor ageing signature.



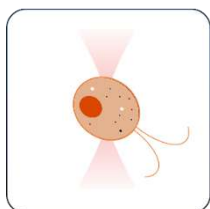
Single-Molecule Biophysics

Study DNA interactions, the activity of motor proteins or characterize the mechanical properties of molecules, filaments and microtubules via 1-bead or 2-bead assays.



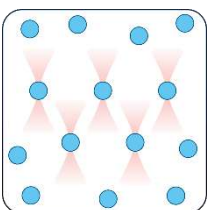
Cell-cell and Cell-ECM Interactions

Measure receptor-binding forces by directly trapping immune and target cells with absolute control on the cells orientation and contact time. Similarly, quantify the focal adhesion strength by attaching functionalized beads on a cell surface.



Bacteria & Microswimmer Force Dynamics

Measure the force dynamics of microswimmers such as bacteria or algae and obtain the system motility energetics by means of stochastic thermodynamics analysis.



Colloidal and Soft Matter Physics

Measure the force-displacement relationship of a colloidal particle in a fluid or gel and use our Active Micro-rheology workflow to obtain the viscoelastic G modulus of complex medium such as gels, active baths or liquid crystals.

Membrane flow and tension propagation

Here we show a **dual membrane tether pulling** experiment to study the spatiotemporal distribution of axonal tension carried out at **M. Krieg's lab (ICFO)** using two trapped beads. Initially, both tethers are separated by a few microns and only one tether is pulled at a time. However, both traps show a characteristic force-relaxation signature indicating **force propagation across the axon** (Fig.1 i-iii). For large tether-to-tether (T2T) distances, the force signature only appears on the active trap (Fig.1 iv-vi). The authors studied how force transmission across the membrane decays with distance for different cell types and conditions while modelling membrane and cytoskeletal conditions (Fig. 2). Optical tweezers allow quantifying **membrane tension fluidity and elasticity**. Combined with fluorescence imaging, the influence of these parameters on several biological functions such as calcium signaling can be studied.

Fig.1 Right: A dual tether pulling experiment performed on a neuron. The force-position data is shown for the two tethers when: they are next to each other (i) and they are subsequently being pulled (ii-iii) and; when the two tethers are separated (iv) and the pulling assay is repeated (v-vi). Bottom left: Data of an assay for increasing T2T.

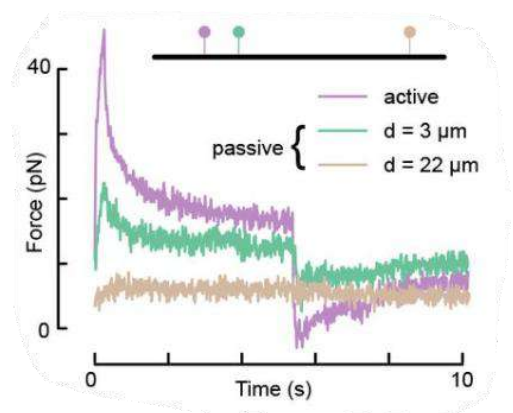
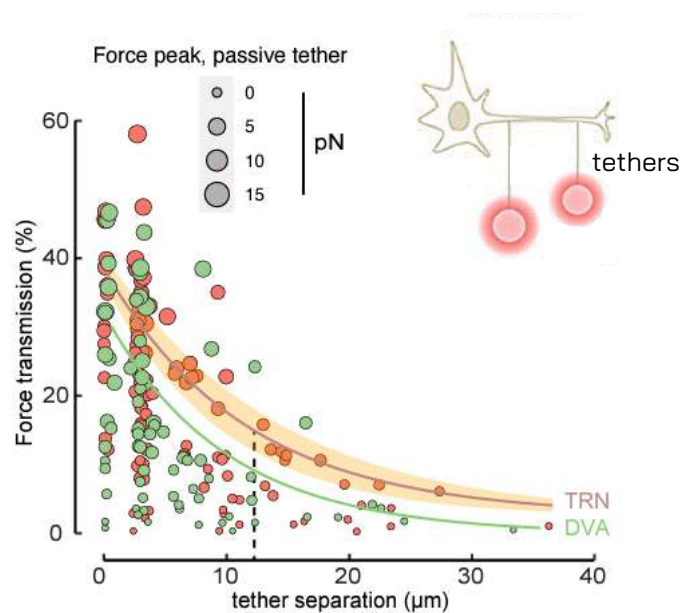


Fig.2 Force transmission decay for increasing T2T distances for Touch Receptor Neurons (TRN) and DVA neurons in *C. elegans*.



Membrane focal adhesions and interactions

The loading rate of force application to single adhesions drives their maturation. In this example (Pere Roca-Cusach lab, IBEC and Timo Betz lab, Göttingen University) a $1\ \mu\text{m}$ fibronectin-coated bead is optically trapped and put in contact with the membrane of a mouse embryonic fibroblast cell. Integrins bind to fibronectin creating focal adhesions while the cell recruits paxillin (Fig.1) that accumulate near the adhesion sites. The bead is subject to periodic oscillations at different frequencies while force and position data is tracked.

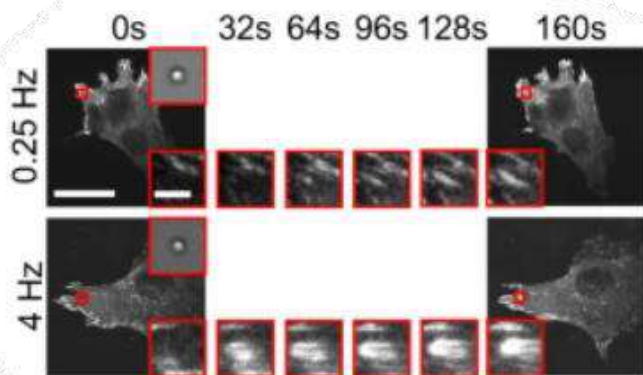
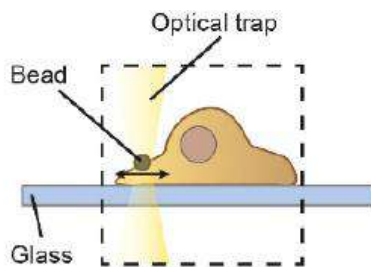


Fig.1 Top: Schematics of a bead-cell adhesion assay. Bottom: Images of cells transfected with GFP-paxillin during force application with triangular signals at 0.25 and 4 Hz, shown as a function of time. The area circled in red indicates the position of the stimulated bead, which is shown magnified at the top-right corner (brightfield image) and bottom-right corner (GFP-paxillin image)

The obtained data shows how the bead displacement decreases with time while force increases when adhesions mature (Fig.2). The force loading rate drives maturation of focal adhesions recruiting paxillin (Fig.3) which connect integrins to actin via vinculin and talin.

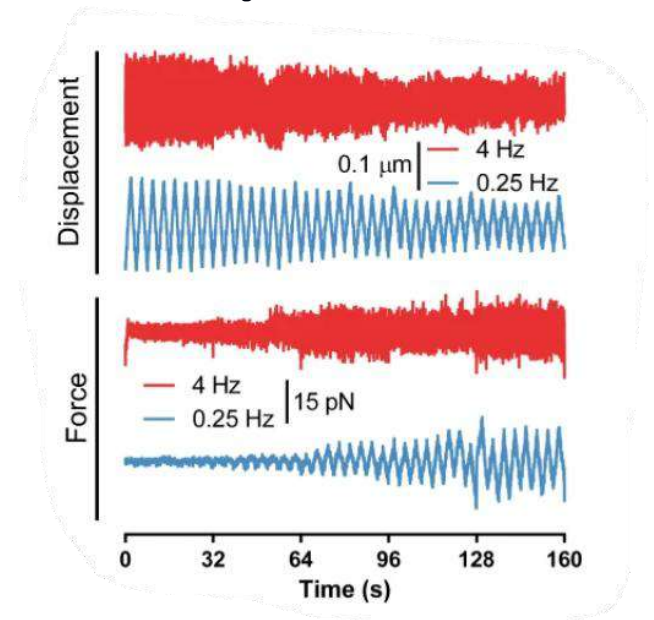


Fig.2 Example traces of displacement and forces traces for beads stimulated at 4 and 0.25 Hz.

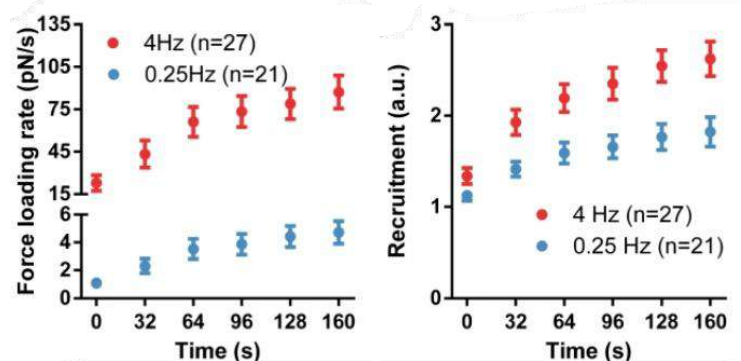


Fig.3 Force loading rate (and recruitment of GFP-paxillin) at beads during maturation of focal adhesions for 4 Hz and 0.25 Hz.



Membrane tension & ion channel activity

Membrane tension gradients are crucial for modulating neuronal activity. Using IMPETUX's SENSOCELL optical tweezers platform, a membrane tether pulling experiment (Michael Krieg's lab, ICFO) was performed by adhering a 1 μm optically trapped fluorescent bead to a neuron. When the adhered bead is pulled away, a lipid filament (tether) is extruded from the cell surface (Fig.1).

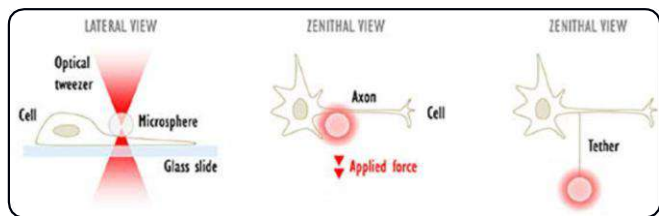
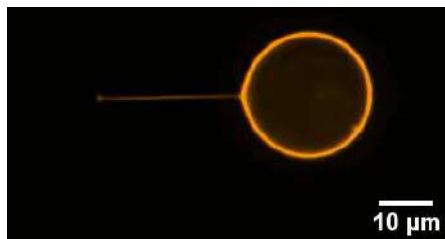


Fig.1 Scheme of a tether pulling experiment performed on a neuron.

The tether is elongated during consecutive pulling steps producing force peaks. After each pulling step, the bead is kept fixed for a certain period of time. During this time, the cell adds material to the membrane tether and the force signal decays. Different pulling rates can be applied at subsequent steps using SENSOCELL's customizable routines. The system monitors the applied force and bead position at real time. The Ca^{2+} fluorescence signal is tracked using confocal imaging.

Tether pulling assays can be performed on a variety of samples, from cells, to GUVs (inset) and embryos.



Tether pulling on a GUV (Confocal imaging) Courtesy of P. Schwile Lab (MPI für Biochemie)

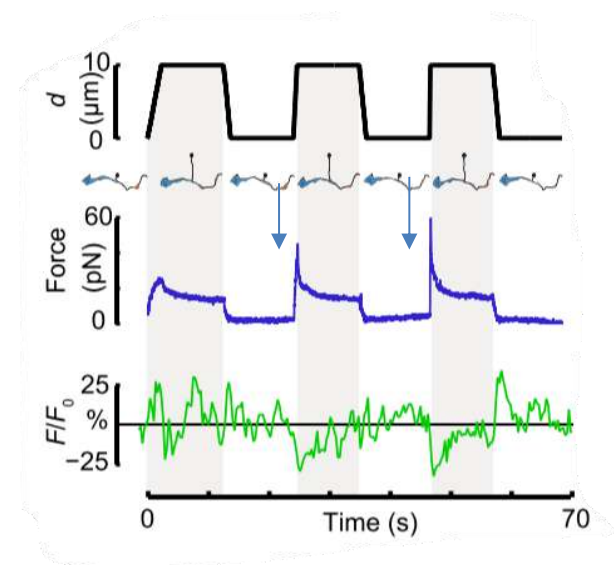
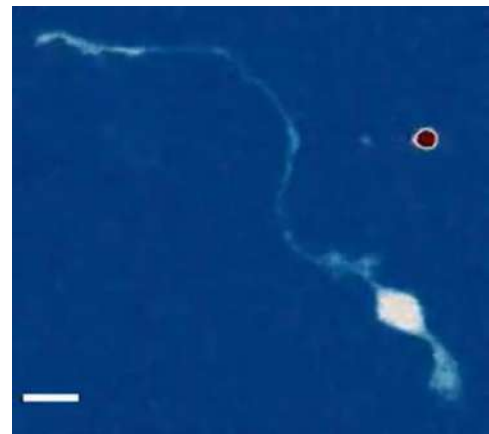


Fig.2 Top: Confocal image during a tether pulling phase (top). Bottom: Force, trap position and Ca^{2+} fluorescence signal during subsequent pulling steps. The pulling rate is increased for each step. The Ca^{2+} fluorescence signal drops during the pulling phase and recovers during the relaxation phase showing negative correlation with membrane tension. Negative correlation between tension and Ca^{2+} activity is observed for *C. elegans* DVA neurons



Membrane-cortex rigidity and permeability

This work shows how mechanical and permeability-related changes induced by interference with the elongase ELOVL6 in T3M4 pancreatic cancer cells can be quantified using SENSOCCELL.

By indenting the plasma membrane with a spherical bead and measuring force relaxation over time, two key mechanical parameters, Young's modulus (E_0), a measure of cortical stiffness and the Diffusion coefficient (D_0), related to membrane permeability and poroelastic relaxation, were measured (see Fig. 3). Inhibition of ELOVL6 led to a significant softening of the cell cortex and an increase in membrane permeability. Figures and data are courtesy of Javier Sánchez Arévalo Lobo (UFV) and Francisco Monroy (UCM).

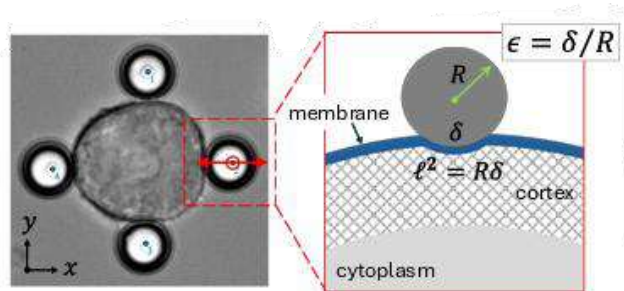


Fig.1 Schematic of the indentation assay where a forcing bead applies lateral strain to a cell immobilized between 3 fixed beads.

The obtained Force-Time traces (see Fig. 2) for the indentation assays depict a characteristic poroelastic relaxation with an initial sharp force overshoot (ΔF) linked to membrane permeability, followed by a slower relaxation to a plateau (F_0) indicating elastic resistance. This approach enables precise determination of cell stiffness (E_0) and diffusivity (D_0), related to membrane permeability and poroelastic relaxation, under small deformations.

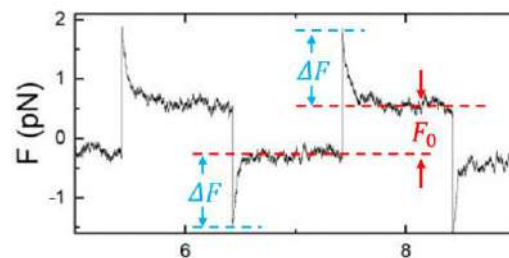


Fig.2 Force-time curves for two subsequent indentation assays performed on a T3M4 cell. The force peak (ΔF) and the final relaxation plateau (F_0) are shown.

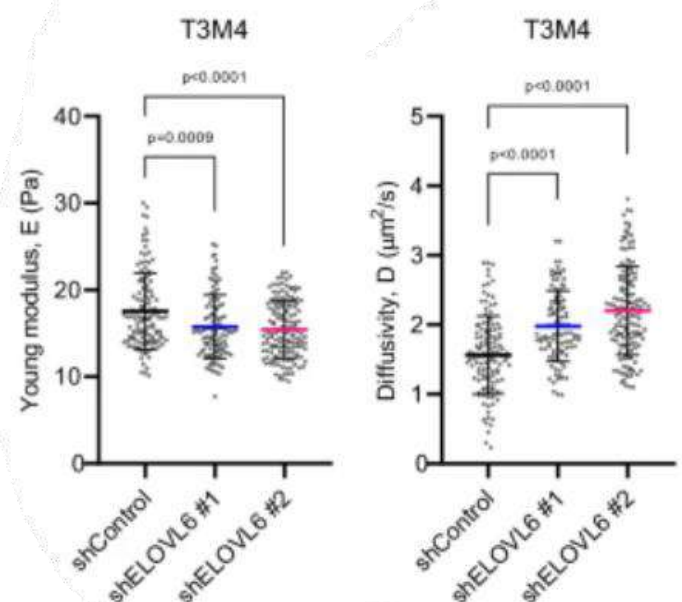


Fig.3 Young's modulus and Diffusivity data of indentation assays in T3M4 cells. Young's modulus decays in ELOVL6 knockdown (shELOVL6) and inhibitor-treated cells, indicating softer cortex mechanics while Diffusivity increases, reflecting greater membrane permeability. These changes were observed without affecting apoptosis, indicating a mechanical and functional, not viability-related, effect.



Cell nucleus stiffness and elasticity

In these examples from Verena Ruprecht lab (UIBK) and Michael Krieg lab (ICFO), SENSOCCELL was used to indent the nucleus of cells in different samples and conditions: from isolated cells (in suspension or confined), to cells inside living Zebrafish embryos (Fig.1) or C. elegans adult worms. Internalized beads or endogenous lipid droplets can be used as optically-trappable force probes. Experiments can be done with multiple traps to deform the nucleus from different sides allowing detecting propagation of forces within the cell. Fig.2 shows a characteristic Force-Time curve for two indentation steps in a suspended zebrafish progenitor stem cell.

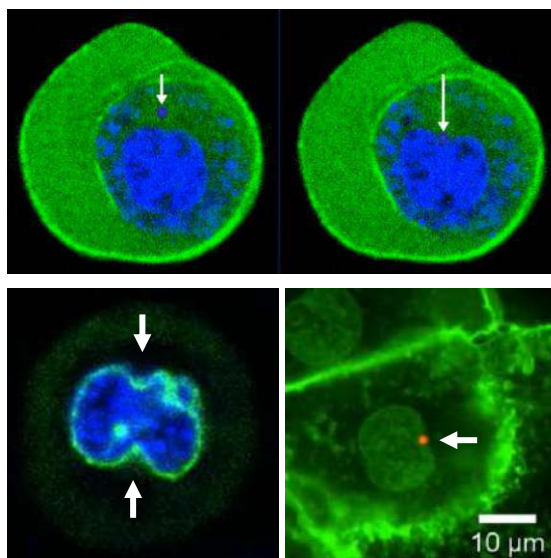


Fig.1 Confocal video snapshots of cell nucleus indentation assays in isolated cells and embryos. Top: 1 bead assay on a stem cell; Bottom left: 2 bead assay on a stem cell. Bottom right: Cell nucleus indentation assay inside a Zebrafish embryo. The arrows point the fluorescent beads used to indent the nucleus. Beads were controlled via an automatized step movement control.

Force and position data are correlated with the saved confocal video frames and can be visualized using our SyncData tool:



Fig.2 Stress-relaxation force signatures (red) for 1 trap during two indentation steps (trap movement in blue). Visualized with SyncData tool.

Results obtained from indentation assays for isolated and confined cells (Fig.3) confirm that the nucleus is predominantly elastic and that it conserves its mechanical properties under confinement so that deformation is reversible. This allows the cell nucleus to behave as an elastic mechanosensor.

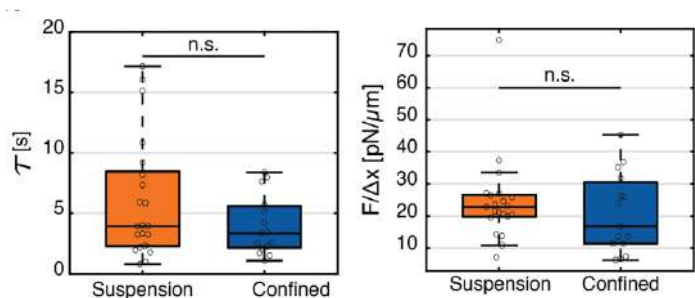


Fig.3 Exponential time decay and stationary force achieved during the relaxation phases of several nucleus indentation assays for suspended (N=21) and confined (N=15) cells.



Direct Force Measurements of Subcellular Mechanics in Confinement using Optical Tweezers.
Frederic Català-Castro et al. *JOVE* (2021)



The nucleus measures shape changes for cellular proprioception to control dynamic cell behavior. Valeria Venturini et al. *Science* (2020)

Cytoplasmic viscoelasticity during cell division

By combining custom optical tweezers with our direct force detector, this study from the [Timo Betz lab \(Göttingen University\)](#) demonstrates how intracellular mechanics evolve throughout cell division. Internalized $1\mu\text{m}$ beads were oscillated inside living cells to perform [active microrheology of the cytoplasm](#). From these measurements, the complex shear modulus G^* , with G' representing elasticity and G'' viscosity, was obtained over a wide frequency range. This revealed that the cytoplasm softens significantly, reaching its [lowest stiffness at the metaphase](#) stage of mitosis. The stiffness prefactor α (derived from G^*) quantifies this effect (Fig. 3), showing a clear drop at metaphase, followed by a gradual recovery as cells progress to telophase. These findings demonstrate the ability of our technology to capture subtle, reversible changes in the viscoelastic landscape of cells.

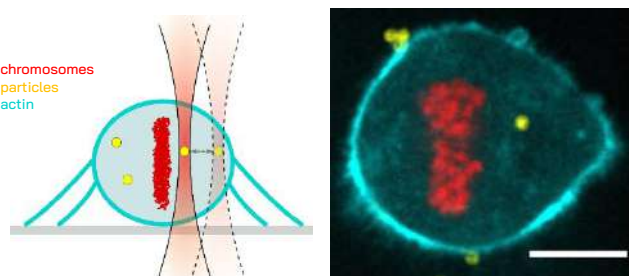


Fig.1 Schematic and confocal image of a mitotic cell with an internalized probe (yellow dot) oscillated by optical tweezers to measure local cytoplasmic mechanics.

While custom setups usually require a secondary laser to finely track the trapped particle's position, [SENSOCELL's integrated Time- Shared Optical Tweezers Microrheology \(TimSOM\)](#) routine uses a single laser for both trapping and tracking. This [simplified hardware](#) avoids complex alignment

steps while the [automatized workflow](#) automatically computes G' and G'' and offers data fitting options with several models used in Soft Matter Physics.

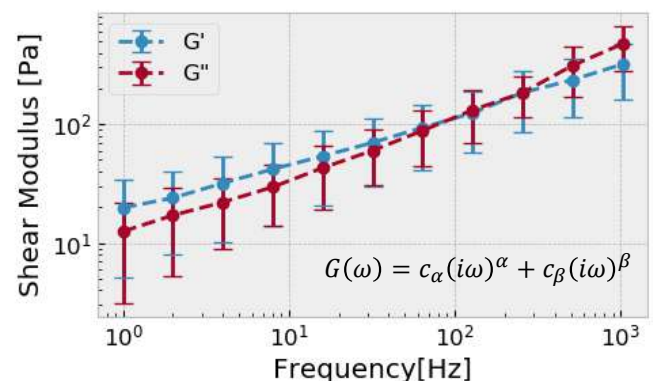


Fig.2 Complex shear modulus $G^*(\omega)$ showing both elastic (G') and viscous (G'') components of the cytoplasm across four frequency decades.

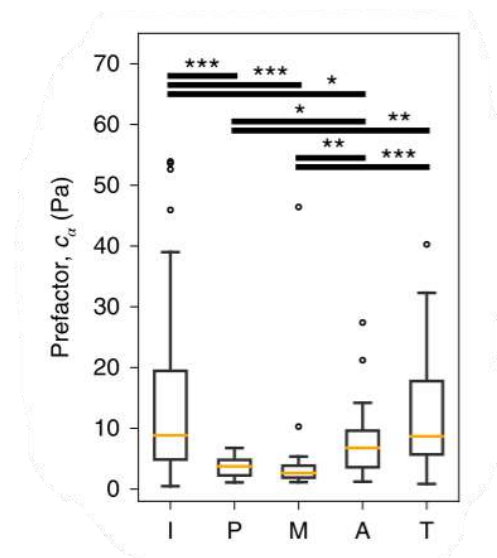


Fig.3 Boxplot of stiffness prefactor C_α across different mitotic phases: Interphase (I), Prophase (P), Metaphase (M), Anaphase (A), Telophase (T). Minimum stiffness is observed at metaphase, reflecting maximal cytoplasmic fluidity.



Viscoelastic ageing (LLPS) of protein condensates

Biomolecular condensates (BMCs) undergo time-dependent physical transitions that reflect changes in their biological state. In this study, optical tweezers microrheology is used to monitor the [mechanical aging of MEC-2 protein droplets](#) over 24 hours. Two complementary approaches are applied: a dual-trap assay, where the droplet is compressed between two optically trapped microspheres, probing interfacial and surface tension; and the TimSOM method, using a single probe bead to assess the bulk rheology of the condensate interior. Data is courtesy of M. Krieg Lab at ICFO. Both approaches reveal a [transition from a liquid-like to a more solid-like state](#) over time. As the droplets age Elastic modulus (E) and viscosity (η) increase while crossover frequency (ω_a) decreases, indicating [slower molecular relaxation](#).

Relaxation time ($\tau = \eta/E$) increases, revealing a gradual arrest in droplet dynamics.

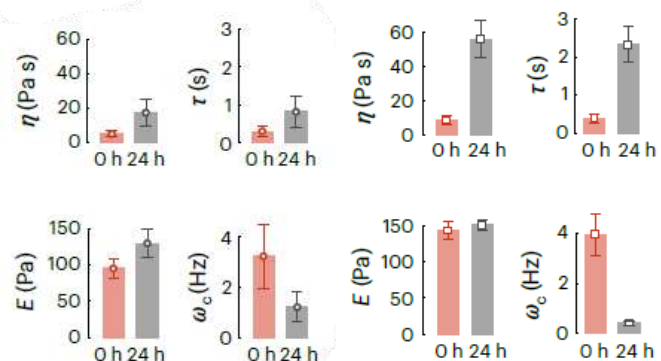


Fig.2 Evolution of elastic modulus (E), viscosity (η), crossover frequency (ω_a), and relaxation time (τ) for the dual trap (left) and single trap (right) assays.

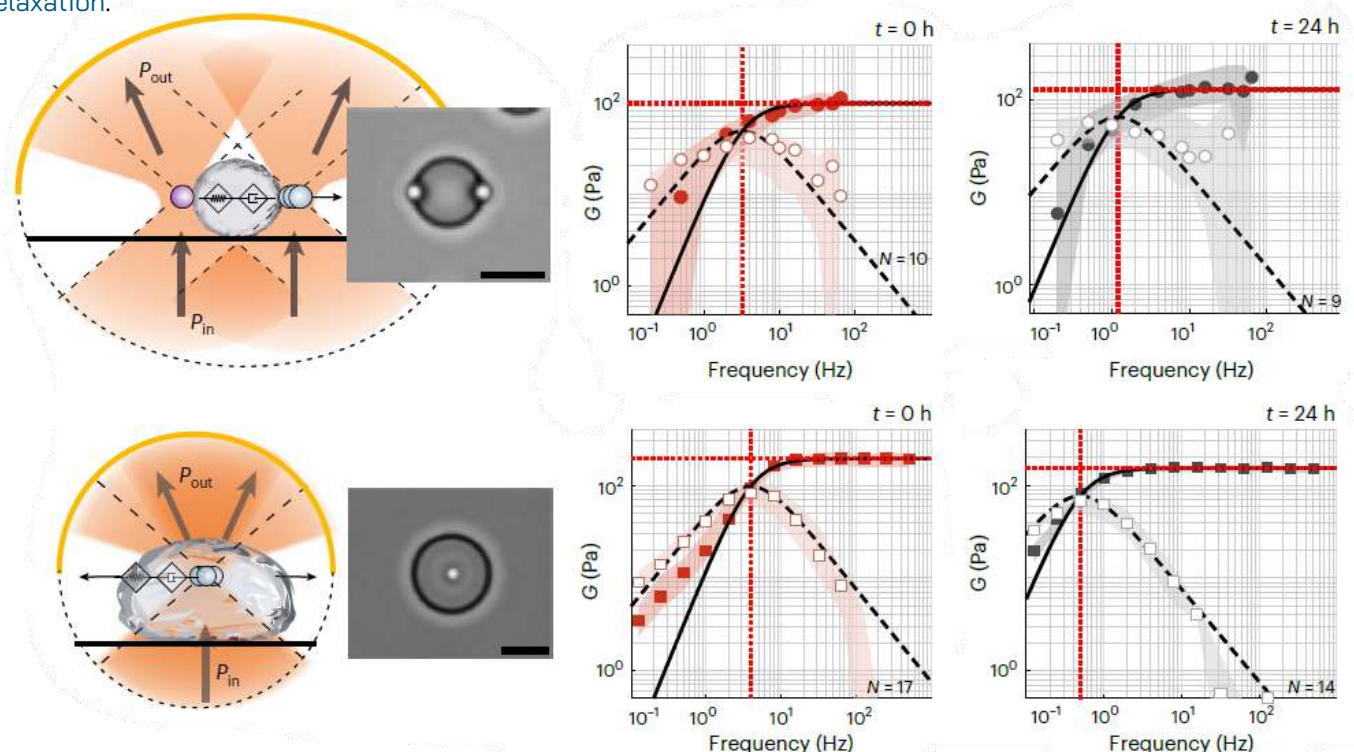


Fig.1 Left: Schematic of two complementary optical tweezer assays: a dual-trap setup probing the external interface of a condensate (top), and the TimSOM setup with an embedded probe measuring internal viscoelasticity (bottom). Right: Frequency-dependent shear moduli (G' , G'') of MEC-2 condensates measured at 0 h and 24 h. Both setups detect a shift toward increased viscosity over time.



Active rheology of cytoplasm & nucleus

SENSOCELL's integrated active microrheology routine (TimSOM) allows measuring the viscoelastic properties of intracellular medium. The routine automatically computes the complex shear modulus G^* , with G' representing elasticity and G'' viscosity, over a frequency range from 0.01 Hz up to 6 kHz.

This study (Michael Krieg lab, ICFO and Verena Ruprecht lab, UIBK) demonstrates how our enables compartment-resolved viscoelastic profiling inside living zebrafish progenitor stem cells. By using a single trapped microsphere sequentially placed in the cytoplasm, at the nuclear interface, and within the nucleoplasm in a single run, the researchers compared the mechanical signatures of these three intracellular compartments under identical conditions.

Using the SENSOCELL system, they measured the complex shear modulus (G^*) across a broad frequency range and fitted the data using a fractional Kelvin–Voigt model. The results show that the nuclear interface is stiffer than both the cytoplasm and nucleoplasm. In the paper it is shown that the cytoplasm softens significantly upon F-actin depolymerization, confirming the role of actin in supporting cellular mechanics. Surprisingly, Lamin A overexpression had little effect on the nuclear interface or nucleoplasm stiffness in microrheology assays. However, when performing larger deformation creep-compliance tests, a slight increase in nucleus rigidity could be observed. The nucleoplasm behaves as a relatively soft compartment with mechanical properties largely unaffected by manipulations.

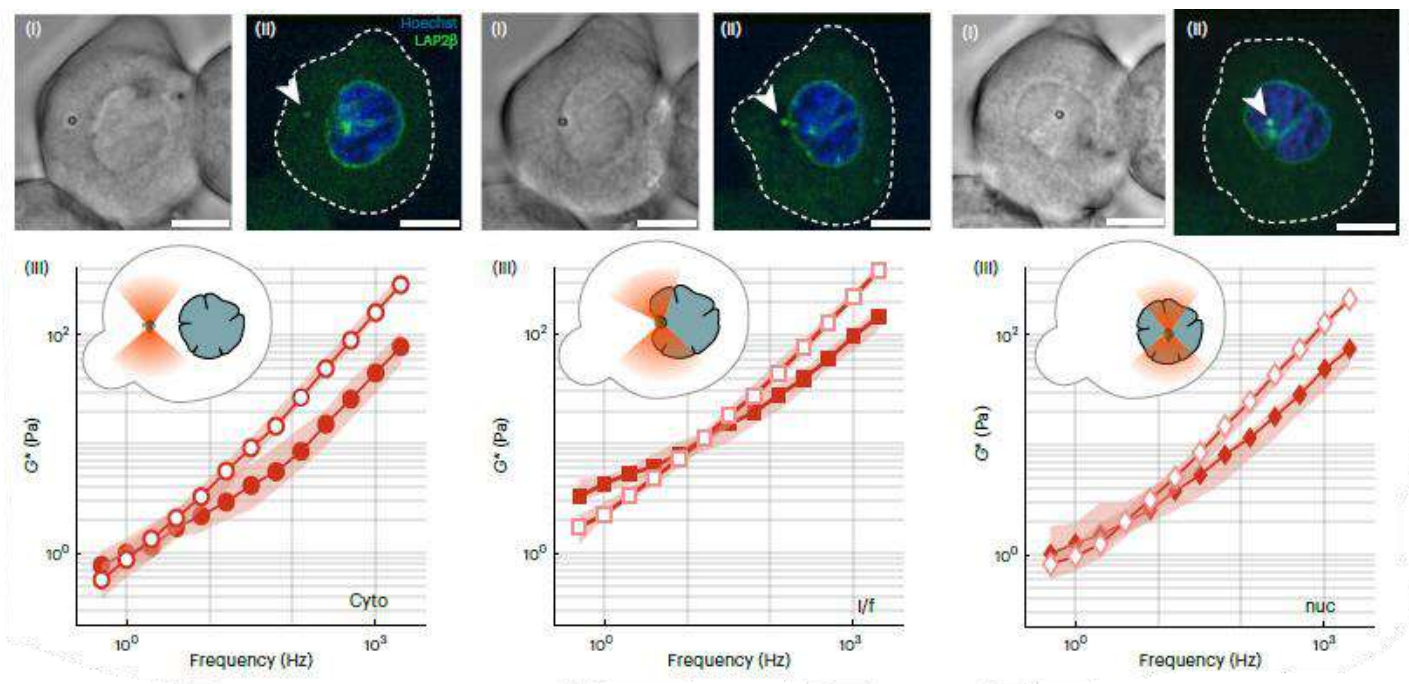


Fig.3 Fig.1 Top: Confocal and bright-field images show bead localization in the cytoplasm (I), nuclear interface (II), and nucleoplasm (III). Bottom: Complex shear modulus spectra showing storage (G') and loss (G'') moduli measured in each compartment.



Immune T-cell and cancer cell interaction forces

Quantifying immune cell-cell adhesion in suspension using optical tweezers:

Understanding how immune cells physically engage with target cells is key in fields such as [immunophysics](#) and [cancer immunotherapy](#). Our optical tweezers platform, SENSOCCELL, enables precise manipulation and [trapping of individual cells in suspension](#), allowing real-time measurement of cell-cell interaction forces under fully controlled conditions.

In this example, we trap a neuroblastoma cancer cell and a T-cell using two independent optical traps. The cancer cell remains in a fixed position, while the T-cell is steered using our “click & drag” mode to make contact. After a defined contact time (e.g., 10 seconds), the T-cell is pulled back. If a molecular interaction has formed, a measurable adhesion force is required to separate the cells. This approach enables users to [quantify immune–target cell adhesion forces](#) with piconewton resolution, with absolute control of contact duration and orientation, allowing testing different target cancer cell types and engineered immune cells, including CAR T-cells.

Adhesion force profiles are acquired in real time. The typical force signal begins at zero (Fig.1 a), increases during separation as tension builds, and drops back to zero once the bond is broken (Fig.1 b). In the example shown, the measured adhesion force was 21pN. Assays were developed in collaboration with [Dr. Carlos Barcia, UAB](#) (neuroblastoma) and [Dr. Manel Juan Otero, Hospital Clínic of Barcelona](#) (lymphoma and engineered immune cells). Statistical studies were also conducted with lymphoma cells and three immune cell types, revealing distinct force distributions depending on receptor type and interaction strength (Fig. 2).

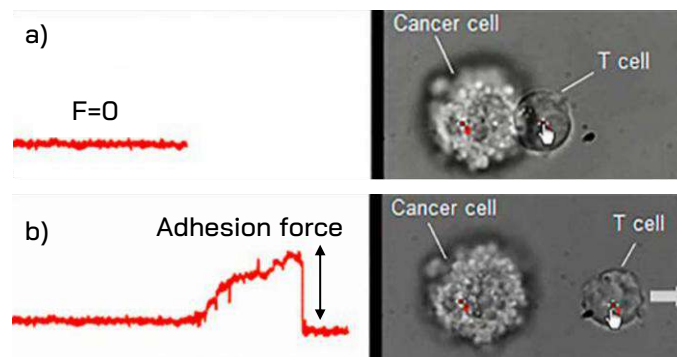


Fig.1 Optical tweezers-based cell-cell interaction assay. The T-cell is brought into contact with a neuroblastoma cancer cell (a). After a few interaction seconds, the T-cell is pulled away while the adhesion force is recorded (b). The force curve shows a characteristic drop when the cell-cell bond breaks during separation corresponding to the cell-cell interaction force.

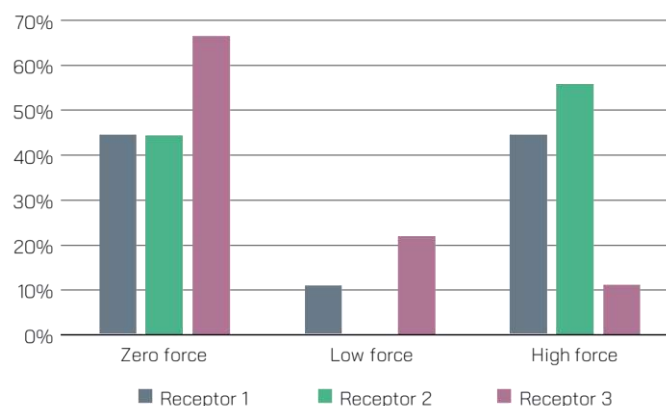


Fig.2 Statistical distribution of adhesion forces measured between lymphoma cancer cells and immune cells expressing three different receptors. Each bar summarizes the outcomes from 9 independent interaction events after 15 s contact time. “Low force” means forces below 60 pN while “high force” means forces over that threshold indicating higher affinity.

Force dynamics of bacteria and microswimmers

Motile microorganisms such as unicellular algae and bacteria continuously expend energy to swim and navigate. With SENSOCCELL, microswimmers can be trapped and their motility forces measured with piconewton sensitivity. Using the principles of stochastic thermodynamics, their **dissipated power and entropy production can be quantified directly from force fluctuations**.

Force dynamics of *Chlamydomonas reinhardtii*

Using a single optical trap, *Chlamydomonas* cells were held in suspension and actively beat their flagella against the restoring force of the trap. The resulting lateral and axial force fluctuations reflect asymmetric strokes, helical swimming behavior and stochastic variation between cells and over time. **Power spectral density analysis reveals clear separation between active and thermal motion**. From this, the average power dissipation was extracted and estimated at ≈ 5 fW. Data courtesy of Wylie Ahmed lab (Toulouse University)

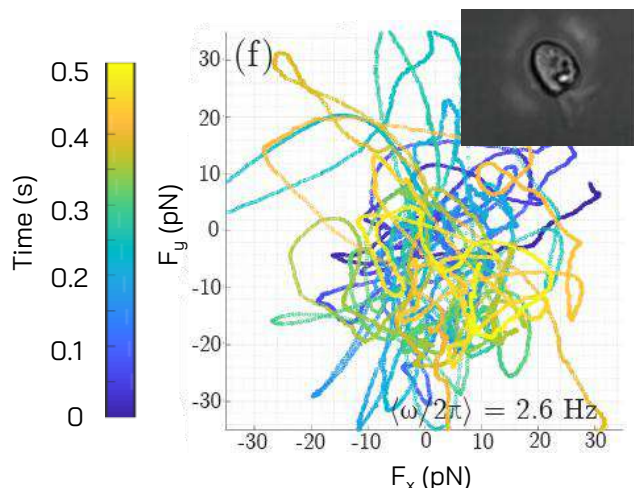


Fig.1 XY Force dynamics of a single *Chlamydomonas* as a function of time showing overall rotational motion of 2.6 Hz.

Energetics of *Proteus mirabilis* swarming behavior

In swarming conditions, *Proteus mirabilis* bacteria exhibit coordinated collective motility. When a single swimmer or a cluster is optically trapped, **lateral force fluctuations arise from both intrinsic activity and swarm-induced mechanical interactions**.

Using stochastic thermodynamics, the dissipated power was computed from force fluctuations and estimated to be 0.5 fW for living swarms. This value is much higher than in dead clusters (≈ 0.01 fW), slightly higher than single bacteria (≈ 0.3 fW), but well below that of uncoordinated groups (≈ 3 fW), indicating that swarming cooperativity reduces net dissipation through mechanical synchronization and lubrication. These results show that **collective bacterial behavior optimizes energetic output** in a way that individual motility cannot. Data courtesy of Francisco Monroy lab (UCM).

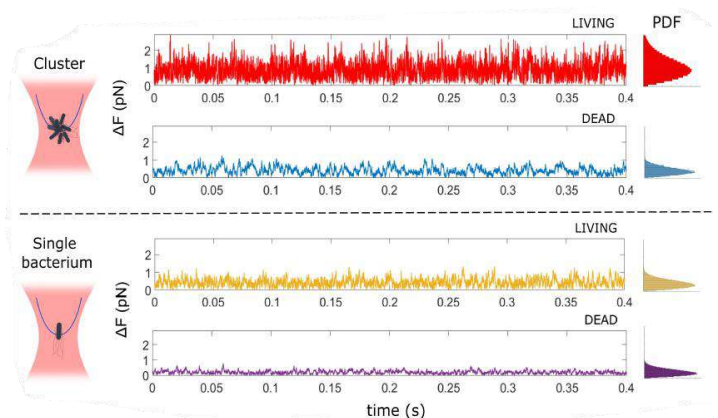


Fig.2 Fluctuating force realizations for living and dead *Proteus mirabilis* clusters (top) and single bacteria (bottom) over time, $\Delta F(t)$. Inserts on the right show force probability distributions following Maxwellian modulus statistics.



Stochastic motility energetics reveals cooperative bacterial swarming in optical tweezers. Clara Luque-Rioja et al. [Arxiv \(2025\)](#)



Stochastic force dynamics of the model microswimmer *Chlamydomonas reinhardtii* : Active forces and energetics. Corbyn Jones et al. [Phys Rev E \(2021\)](#)

Motor protein stall forces measured in living cells

Motor proteins are responsible for driving many essential biological processes inside cells. Among their key functions is the active transport of vesicles, organelles, and other cargos along the cytoskeleton. One of the best-characterized motor proteins is *kinesin*, which walks toward the plus-end of microtubules by hydrolyzing ATP, thereby converting chemical energy into mechanical work. Using SENSOCCELL optical tweezers, we can measure the forces generated by molecular motors not only in *in vitro* conditions but also in their native cellular environment by trapping intracellular targets such as lipid droplets. In Fig.1, we show the stall force of a single kinesin motor transporting a lipid droplet in an A549 cell. The force plateaus at ≈ 7 pN is consistent with known values for single kinesin molecules *in vitro*. In a similar way, we can also reveal how multiple

motors coordinate or compete when attached to the same cargo in a living A549 cell (Fig. 2).

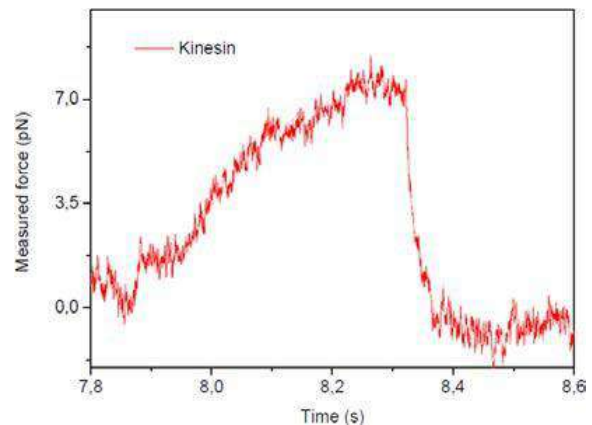


Fig.1 Stall force measurement of a lipid droplet in an A549. The force plateaus at ≈ 7 pN indicates motor stalling consistent with single kinesin mechanics.

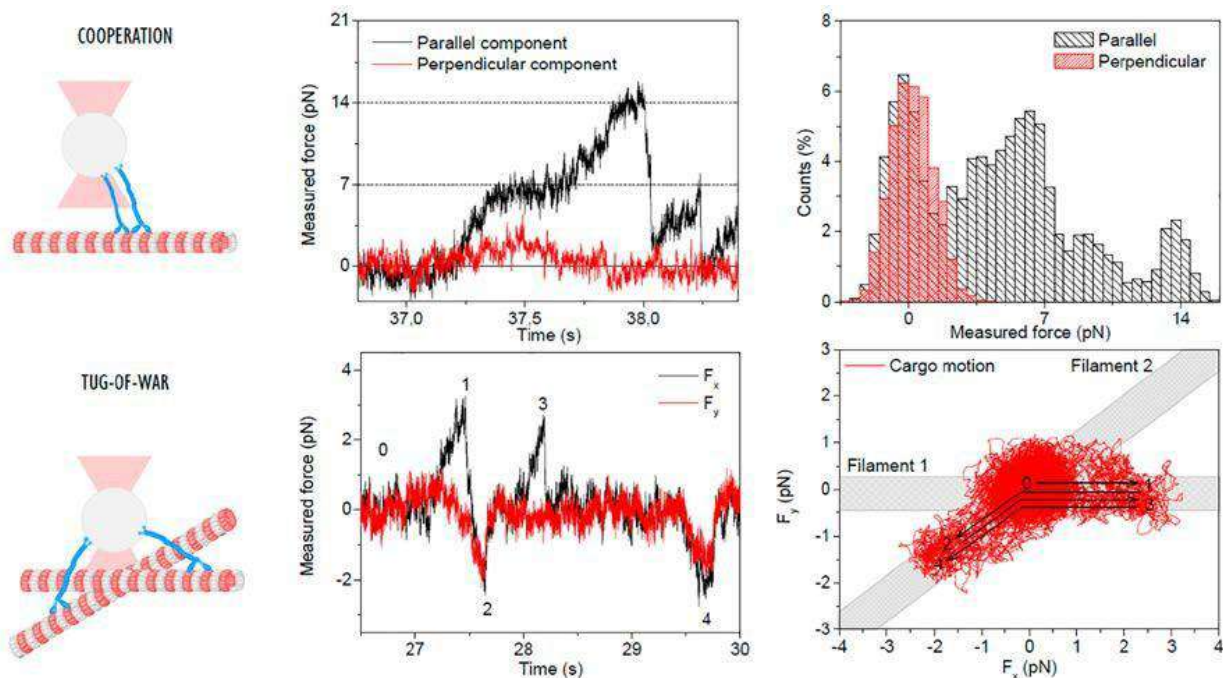


Fig.2 Intracellular force profiles for a cargo transported by multiple kinesin motors. Cooperation: Two motors walk along the same microtubule direction. Competition (tug-of-war): Motors pull the same cargo along different microtubules or directions. Top left: Cooperation scenario. Force rises in discrete steps (≈ 7 and 14 pN), aligned with microtubule direction. Top right: Histogram showing force distribution with peaks at 7 pN (1 motor) and 14 pN (2 motors). Bottom left: Competition (tug-of-war) case. Force fluctuates along both parallel (F_x) and perpendicular (F_y) axes. Bottom right: XY force map shows transitions between force directions, reflecting cargo switching between filament tracks. Data is courtesy of Biopt Lab (UB).

Force dynamics in passive or active colloids

Optical tweezers provide a powerful toolset for investigating colloidal systems by enabling precise manipulation and direct measurement of forces at the pN scale. From surface wetting phenomena to energy dissipation in active suspensions, optical trapping allows researchers to probe the mechanics and dynamics of soft matter with unmatched resolution and control. In a recent study, Squillace et al. (2024) used optical tweezers to explore the wetting interactions between silicone oil droplets and textile fibers in aqueous environments. By measuring the force needed to detach individual droplets from single fibers, the authors quantified adhesion forces and capillary suction effects under varying ionic strength and surfactant concentrations. As shown in Figure 1, the adhesion force and effective contact angle were sensitive to both electrolyte content and surfactant chemistry, revealing how surface interactions can be tuned at the nanoscale. Data is courtesy of Andrew Ward (Central Laser Facility, UK).

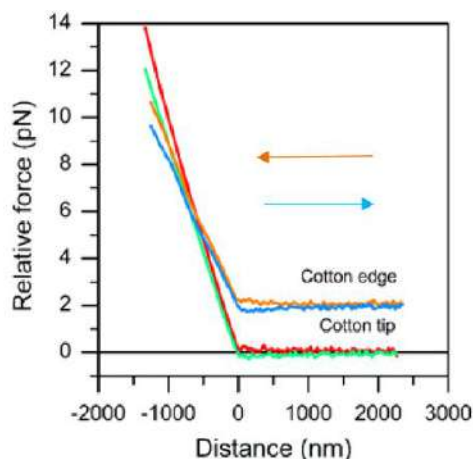


Fig.1 Adhesion forces between silicone oil droplets and textile fibers measured under varying salt and surfactant conditions.

In a complementary application, H. Seyforth et al. (2022) investigated the nonequilibrium force fluctuations experienced by a colloidal bead trapped in a bath of motile *E. coli* bacteria. The active forces generated by swimming cells cause enhanced stochastic motion of the bead as illustrated in Figure 2. Data is courtesy of W. Ahmed Lab (Toulouse University). The team measured heat production, fluctuation-dissipation violations, and energy dissipation rates in active matter. This technique provides a unique quantitative window into non-equilibrium thermodynamics at the microscale.

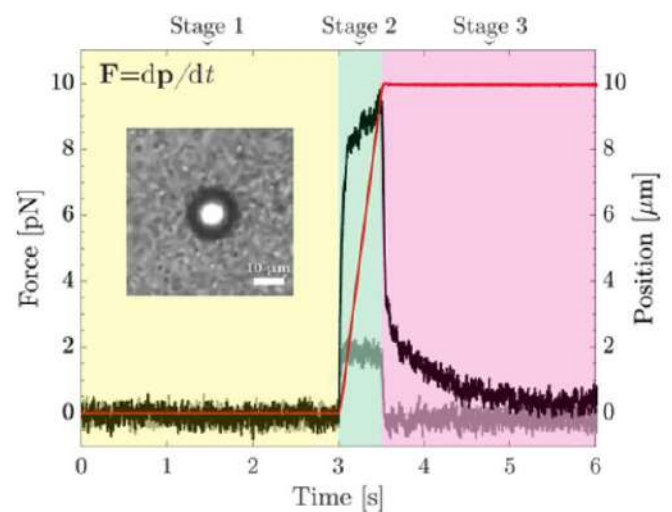


Fig.2 Force fluctuations of a colloidal probe in an active bath (black data) and passive bath (grey data). The optical trap captures active noise signatures and enables quantification of energy dissipation in an out-of-equilibrium system.

Together, these studies highlight the versatility of optical tweezers for measuring colloidal forces in both passive and active systems, offering insights into adhesion, wetting, active noise, and dissipation in complex environments.



Nanoscopic wetting behaviour of single oil droplets on a fibre. *Ophelie Squillace et al. Colloids and Surfaces A: Physicochemical and Engineering Aspects (2024)*



Nonequilibrium fluctuations and nonlinear response of an active bath *Hunter Seyforth et al. Phys. Rev. Res (2022)*

Key Specifications

Laser Unit	1064 nm SM fiber-coupled TEM00 single-frequency laser (3W)
Laser key specs	Linewidth <500kHz. Noise < ± 0.1 % rms (@ 100 Hz – 10 MHz)
Trapping module	AOD based: up to 256 independent optical traps (25 kHz steering)
Manipulation modes	Click and drag, trajectories, oscillations and dynamic trap patterns
Trap stiffness	~ 3 pN/($\mu\text{m}\cdot\text{mW}$) for 1 μm polystyrene bead. Normalized to laser power
Trap resolution	Position resolution < 1 nm. Minimum displacement < 0.1 nm
Trapping area	70 x 70 μm with power homogeneity < ± 2 %
Force detection	Direct force detection based on Light Momentum. Calibration-free
Multiple readings	Simultaneous force measurements on all active traps
Force resolution	50 fN resolution and 25 kHz sampling rate
Detection modes	Force detection - Force clamp mode - Position tracking (< 1 nm res.)
Confocal upgrade	Confocal, Widefield and Brightfield: 1-click instant switch between modes 405 – 473 – 561 – 639 nm laser lines FOV: 23.1 mm 230 fps at full FOV. Camera limited Deep imaging: hundreds of microns penetration capacity Diffraction limited resolution Camera-based detection (sCMOS with QE> 90%) Simultaneous color imaging in sequential mode for up to 4 colors Motorized filter wheel (6 positions) Selectable ROI for illumination
Epi-FL upgrade	Available Epi-Fluorescence imaging modules from Ti2-LAPP system
Incubation cage	SENSOCELL can be upgraded with a custom incubator chamber from OKOLAB with temperature control (3°C above ambient to 42 °C)

Embedded Class 4 IR laser integrated into a research-grade inverted microscope, with controlled beam delivery to the sample and safety interlock. Intended for use by trained users in research laboratory environments.



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