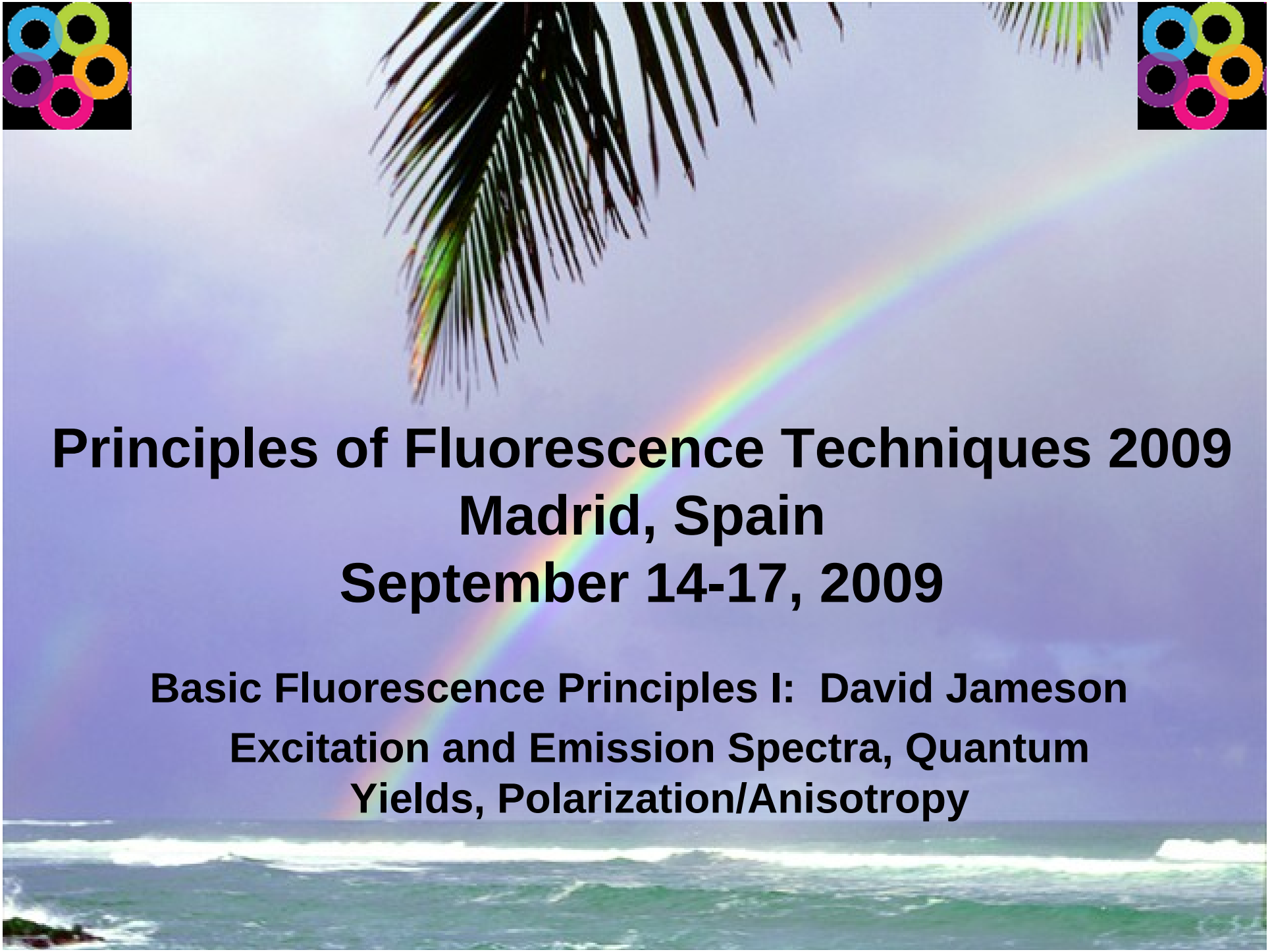


Principles of Fluorescence Techniques 2009
Madrid, Spain
September 14-17, 2009

Basic Fluorescence Principles I: David Jameson

Excitation and Emission Spectra, Quantum Yields, Polarization/Anisotropy

What is fluorescence?

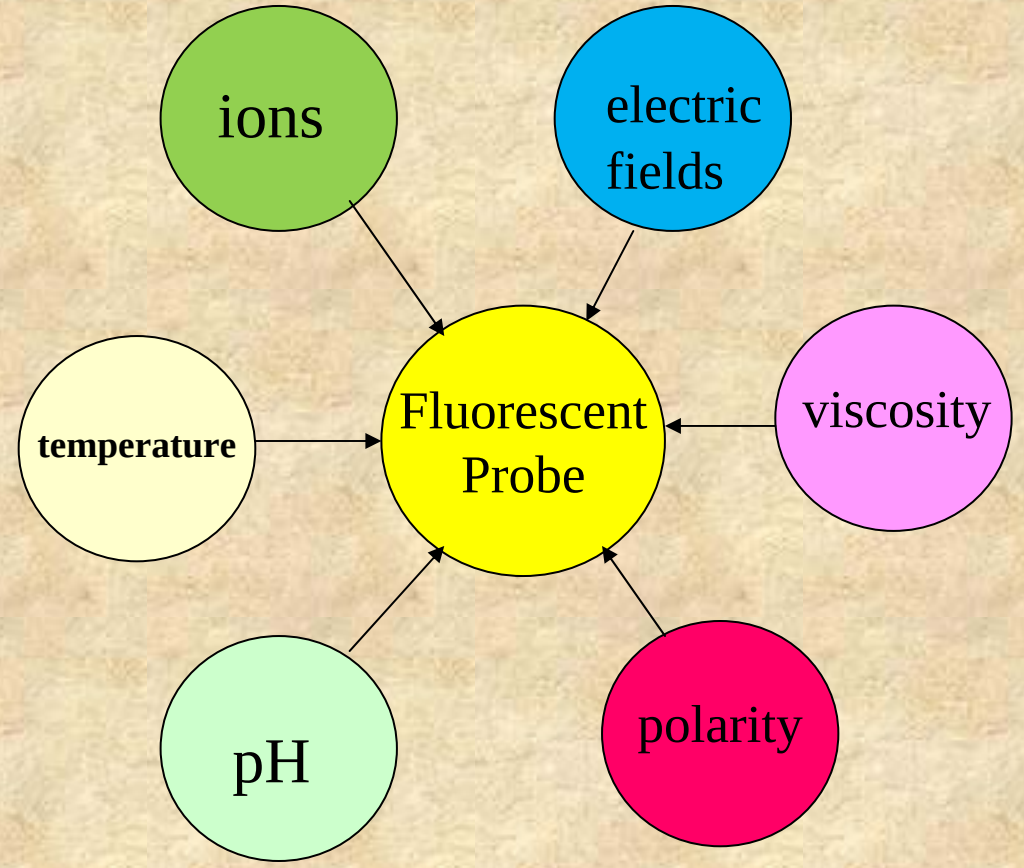
FLUORESCENCE is the light emitted by an atom or molecule after a finite duration subsequent to the absorption of electromagnetic energy.

Specifically, the emitted light arises from the transition of the excited species from its first excited electronic singlet level to its ground electronic level. **(usually)**

The development of highly sophisticated fluorescent probe chemistries, new laser and microcopy approaches and site-directed mutagenesis has led to many novel applications of fluorescence in the chemical, physical and life sciences. Fluorescence methodologies are now widely used in the biochemical and biophysical areas, in clinical chemistry and diagnostics and in cell biology and molecular biology.

Why fluorescence?

- its pretty!
- it provides information on the molecular environment
- it provides information on dynamic processes on the nanosecond timescale



Fluorescence Probes are essentially <u>molecular stopwatches</u> which monitor *dynamic* events which occur during the excited state lifetime – such as movements of proteins or protein domains

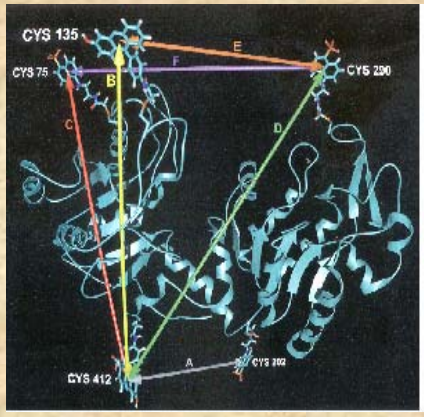
Coupled with modern fluorescence microscopy (confocal, multiphoton, etc) and fluorescent proteins (such as GFP, etc) fluorescence is also providing extremely detailed spatial information in living cells – as well as information on the dynamics of cellular components

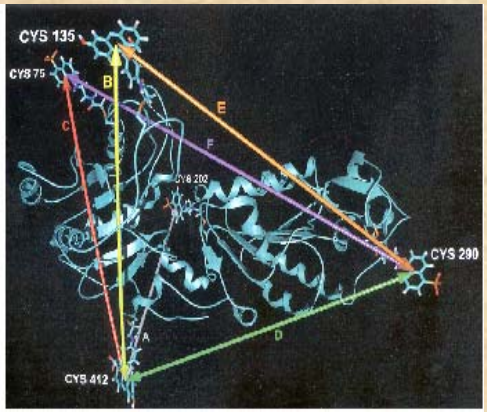
Also fluorescence is very, very, very sensitive!

Work with subnanomolar concentrations is routine while femtomolar and even SINGLE MOLECULE studies are possible with some effort

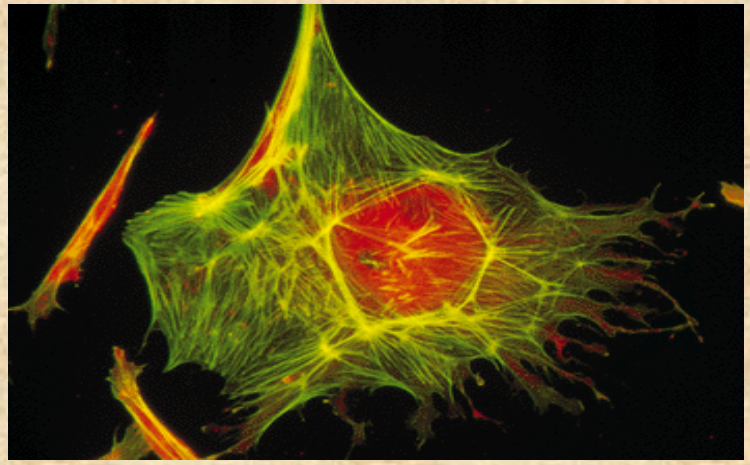
Experimental Systems Accessible to Fluorescence

Molecular structure and dynamics



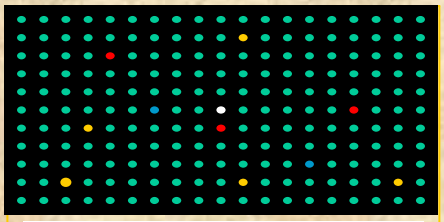


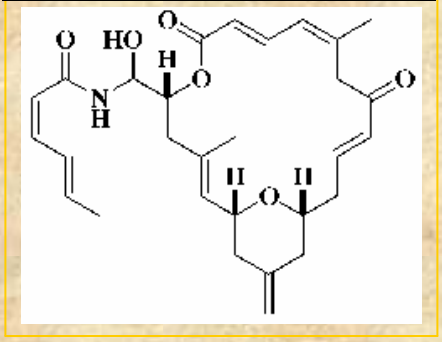
Cell organization and function



Live Animals

Engineered surfaces





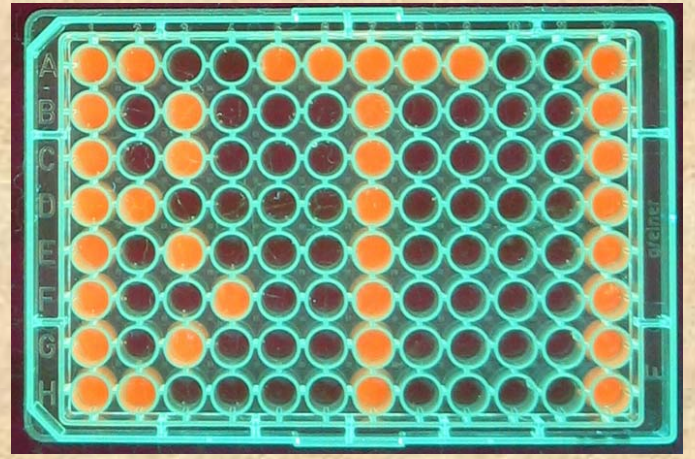
High throughput
Drug discovery

Instrumentation

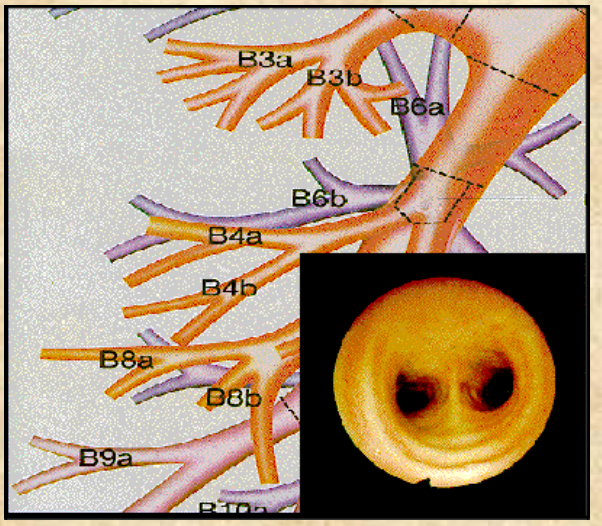
Fluorimeters

Microscopes

High throughput Platereaders



Intravital Imaging



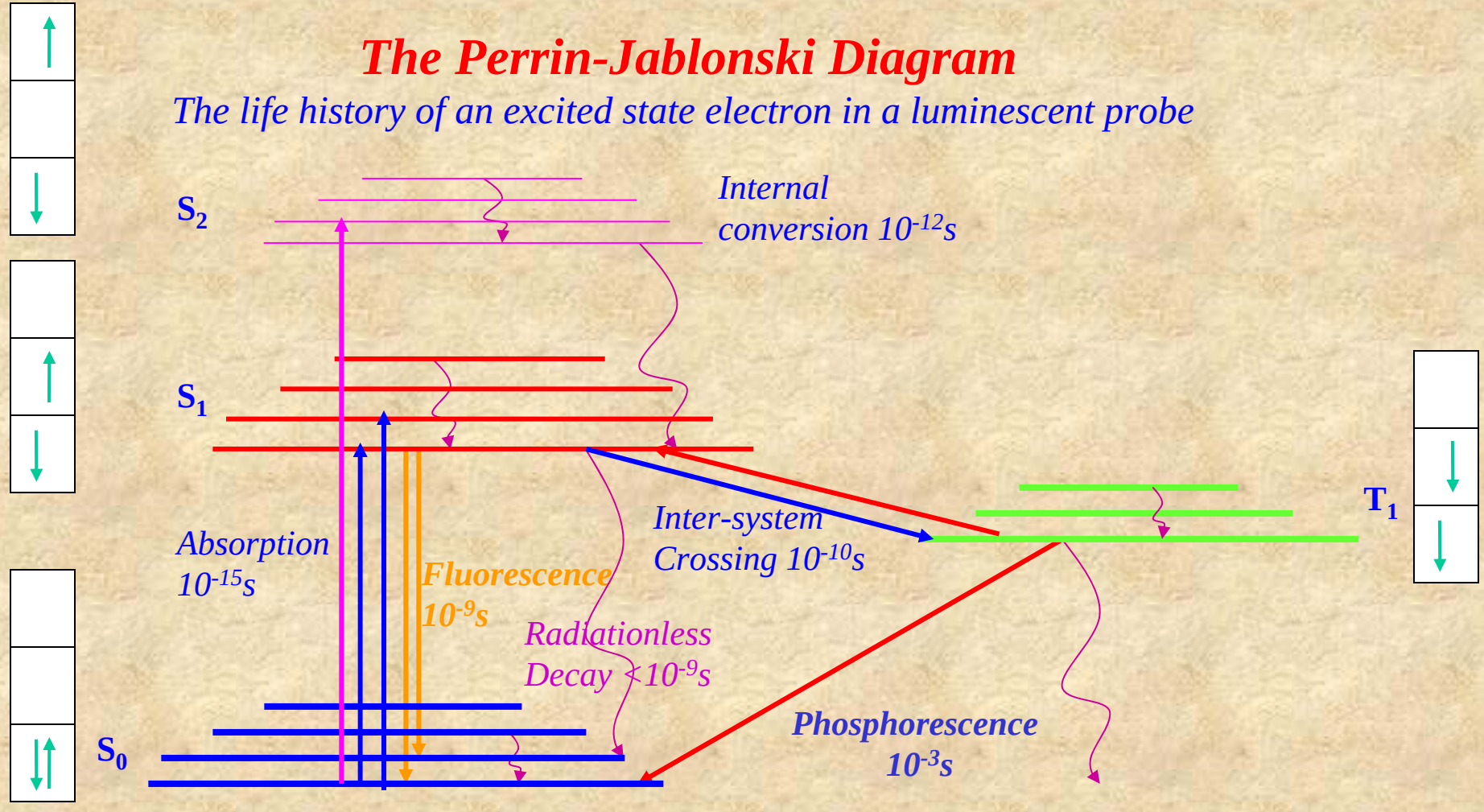
Virtually all fluorescence data required for any research project will fall into one of the following categories.

1. The fluorescence emission spectrum
2. The excitation spectrum of the fluorescence
3. The quantum yield
4. The polarization (anisotropy) of the emission
5. The fluorescence lifetime

In these lectures, we examine each of these categories and briefly discuss historical developments, underlying concepts and practical considerations

The Perrin-Jablonski Diagram

The life history of an excited state electron in a luminescent probe



Key points:

- ✓***Excitation spectra are mirror images of the emission spectra***
- ✓***Emission has lower energy compared to absorption***
- ✓***Triplet emission is lower in energy compared to singlet emission***
- ✓***Most emission/quenching/FRET/chemical reactions occur from the lowest vibrational level of $[S]_1$***

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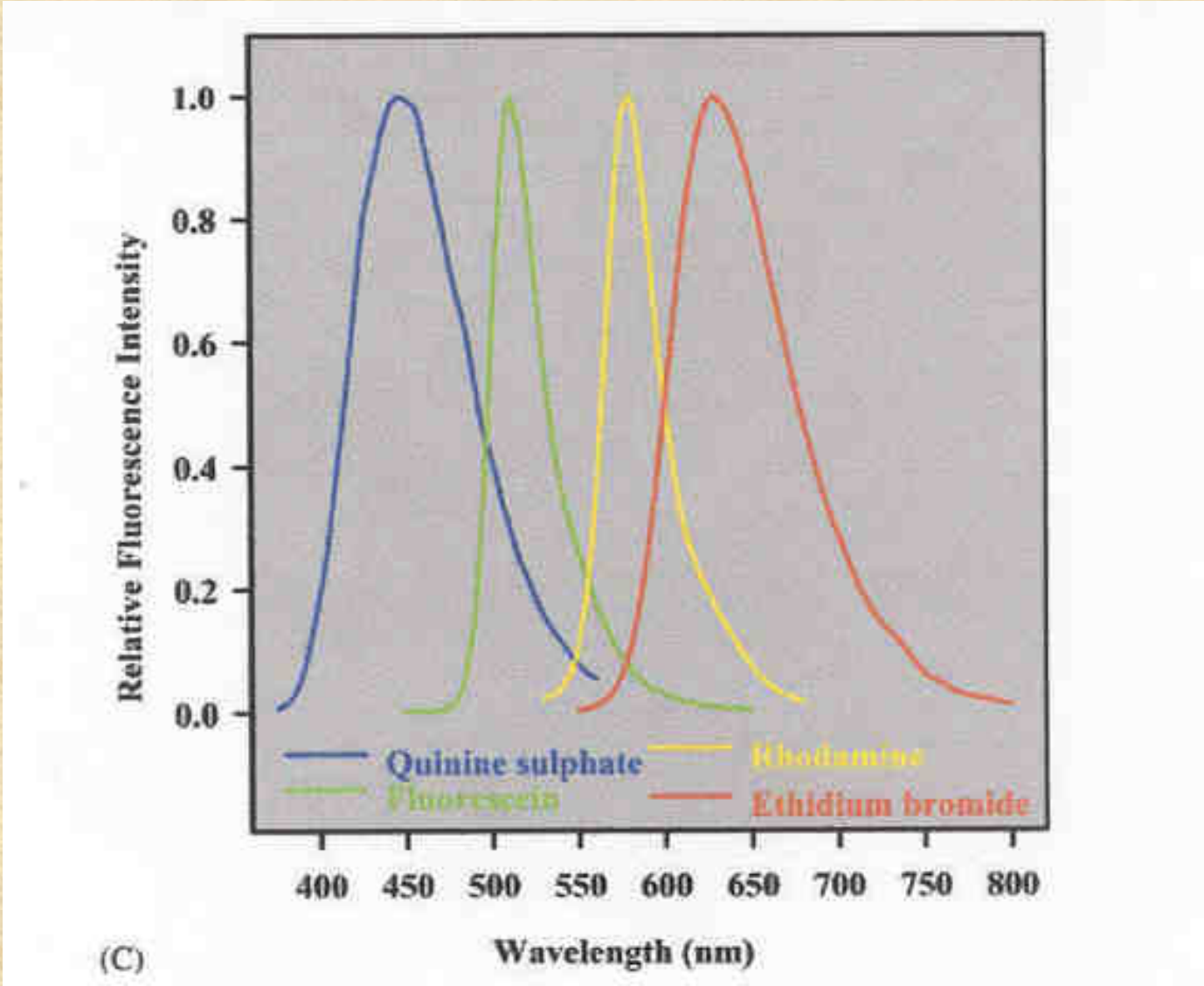
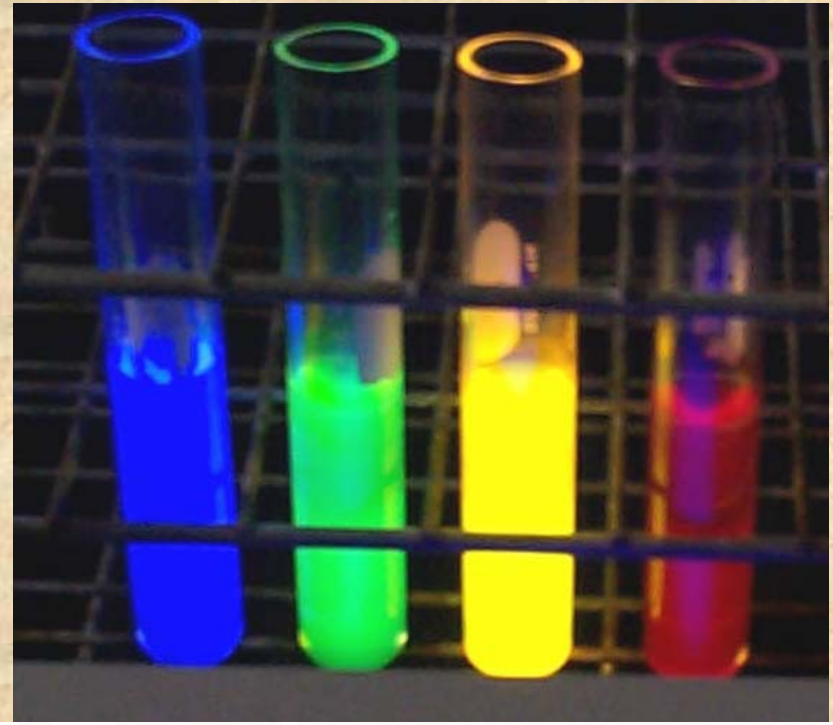
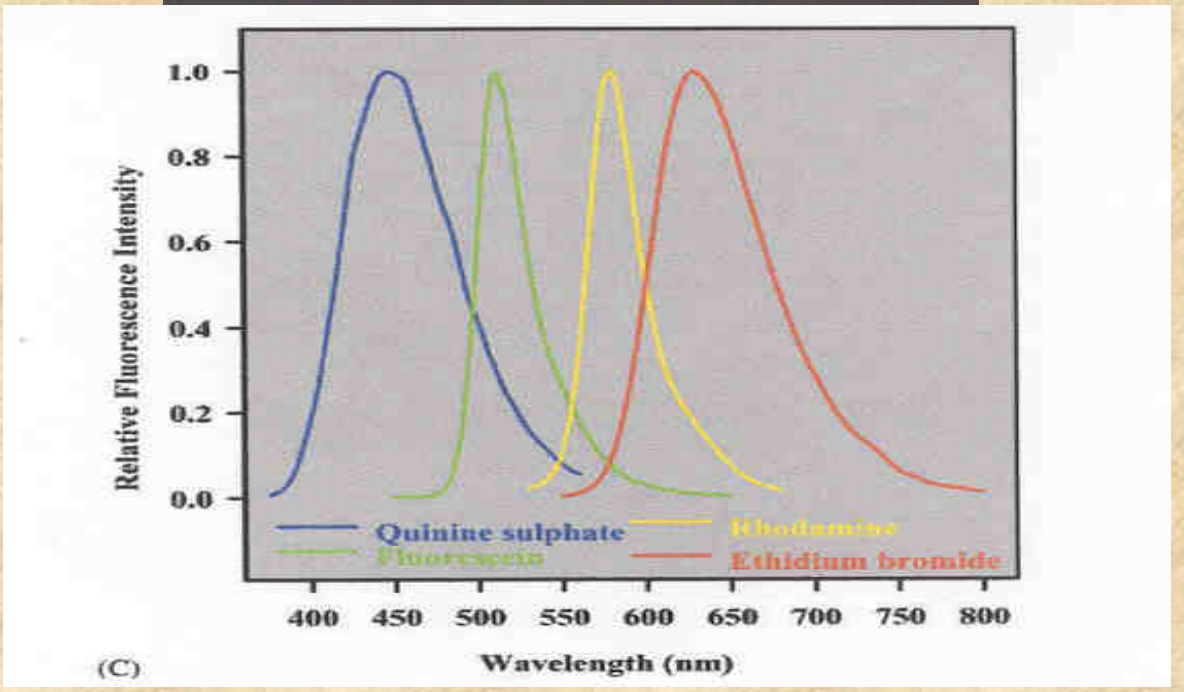
The fluorescence emission spectrum

In a typical emission spectrum, the excitation wavelength is fixed and the fluorescence intensity versus wavelength is obtained






Relative Fluorescence Intensity
1.0
0.8
0.6
0.4
0.2
0.0
Quinine sulphate
Fluorescein
Rhodamine
Ethidium bromide
400
450
500
550
600
650
700
750
800
Wavelength (nm)
(C)

Early examination of a large number of emission spectra resulted in the formulation of certain general rules:

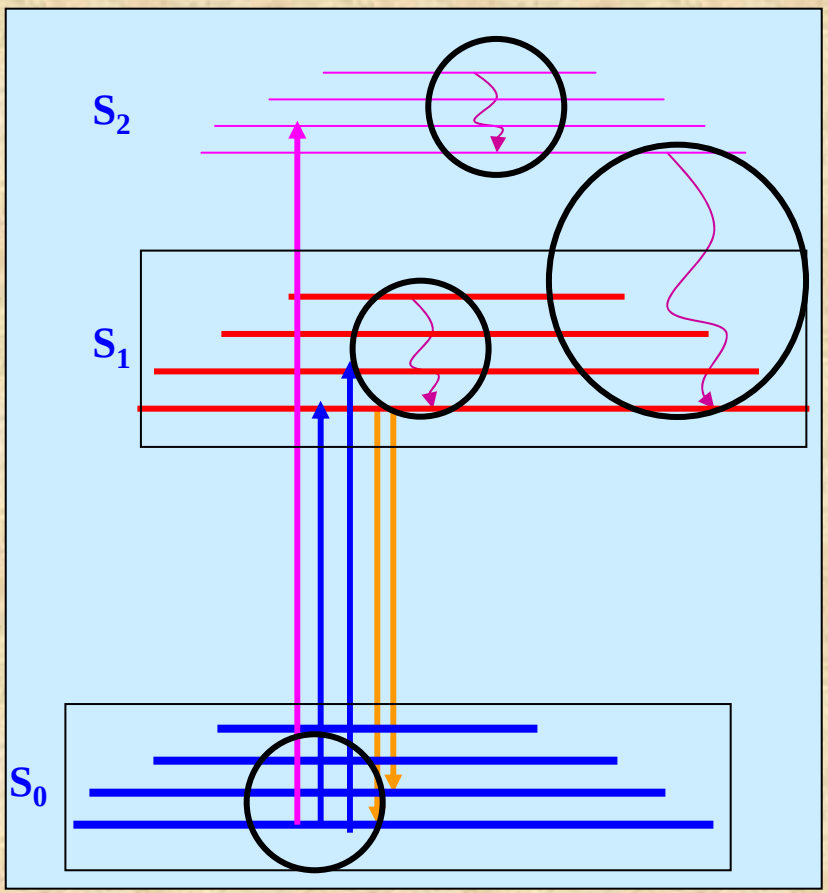
1) In a pure substance existing in solution in a unique form, the fluorescence spectrum is invariant, remaining the same independent of the excitation wavelength

2) The fluorescence spectrum lies at longer wavelengths than the absorption

3) The fluorescence spectrum is, to a good approximation, a mirror image of the absorption band of least frequency

These general observations follow from consideration of the Perrin-Jabłoński diagram shown earlier

Specifically, although the fluorophore may be excited into different singlet state energy levels (e.g., S_1, S_2, etc) rapid thermalization invariably occurs and emission takes place from the lowest vibrational level of the first excited electronic state (S_1). This fact accounts for the independence of the emission spectrum from the excitation wavelength.



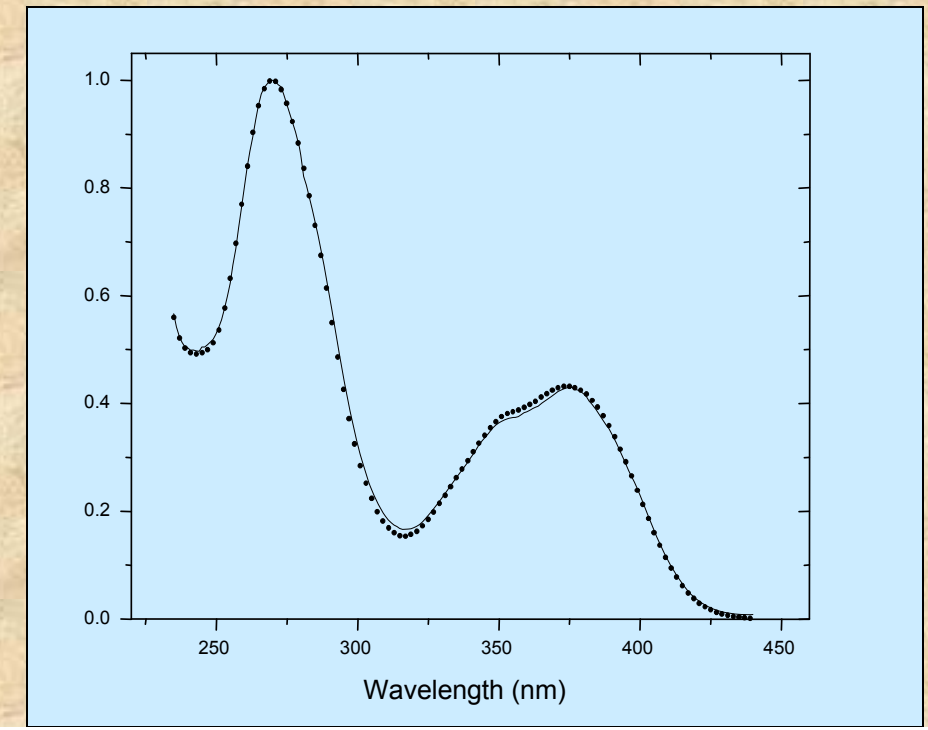
The fact that ground state fluorophores, at room temperature, are predominantly in the lowest vibrational level of the ground electronic state (as required from Boltzmann's distribution law) accounts for the Stokes shift.

Finally, the fact that the spacings of the energy levels in the vibrational manifolds of the ground state and first excited electronic states are usually similar accounts for the fact that the emission and absorption spectra (plotted in energy units such as reciprocal wavenumbers) are approximately mirror images

The fluorescence excitation spectrum

The relative efficiencies of different wavelengths of incident light to excite fluorophores is determined as the excitation spectrum. In this case, the excitation monochromator is varied while the emission wavelength is kept constant if a monochromator is utilized - or the emitted light can be observed through a filter.

If the system is “well-behaved”, i.e., if the three general rules outlined above hold, one would expect that the excitation spectrum will match the absorption spectrum. In this case, however, as in the case of the emission spectrum, corrections for instrumentation factors are required.



Overlay of Absorption Spectrum and Corrected Excitation Spectrum for ANS in ethanol

Quantum Yield

The quantum yield of fluorescence (QY) is dependent on the ***rate of the emission process*** divided by the sum of the ***rates of all other deactivation processes***

$$QY = k_f / k_f + k_i + k_x$$

k_f is the rate of fluorescence, k_i is the rate of radiationless decay and k_x is the rate of intersystem crossing.

Another way to think about QY is:

QY = Number of emitted photons / Number of absorbed photons

If the rates of the deactivation processes are slow compared to k_f then the **QY** *is high*

However, if the rates of these other processes are fast compared to k_f then ***QY*** is low

GM

List of quantum yields from “Molecular Fluorescence” by Bernard Valeur

Tab. 6.1. Standards for the determination of fluorescence quantum yields

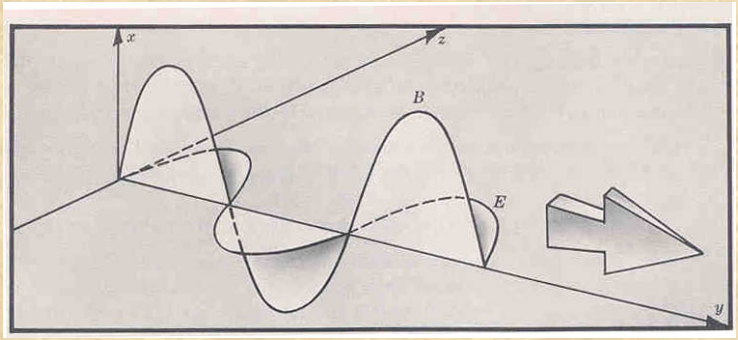
Range	*Compound*	*Temp. (°C)*	*Solvent*	Φ_F	*Ref.*
270–300 nm	Benzene	20	Cyclohexane	0.05 ± 0.02	1
300–380 nm	Tryptophan	25	H_2O (pH 7.2)	0.14 ± 0.02	2
300–400 nm	Naphthalene	20	Cyclohexane	0.23 ± 0.02	3
315–480 nm	2-Aminopyridine	20	0.1 mol L^{-1} H_2SO_4	0.60 ± 0.05	4
360–480 nm	Anthracene	20	Ethanol	0.27 ± 0.03	1, 5
400–500 nm	9,10-diphenylanthracene	20	Cyclohexane	0.90 ± 0.02	6, 7
400–600 nm	Quinine sulfate dihydrate	20	0.5 mol L^{-1} H_2SO_4	0.546	5, 7
600–650 nm	Rhodamine 101	20	Ethanol	1.0 ± 0.02	8
				0.92 ± 0.02	9
600–650 nm	Cresyl violet	20	Methanol	0.54 ± 0.03	10

1) Dawson W. R. and Windsor M. W. (1968) *J. Phys. Chem.* 72, 3251.
2) Kirby E. P. and Steiner R. F. (1970) *J. Phys. Chem.* 74, 4480.
3) Berlman I. B. (1965) *Handbook of Fluorescence Spectra of Aromatic Molecules*, Academic Press, London.
4) Rusakowicz R. and Testa A. C. (1968) *J. Phys. Chem.* 72, 2680.
5) Melhuish W. H. (1961) *J. Phys. Chem.* 65, 229.
6) Hamai S. and Hirayama F. (1983) *J. Phys. Chem.* 87, 83.
7) Meech S. R. and Phillips D. (1983) *J. Photochem.* 23, 193.
8) Karstens T. and Kobs K. (1980) *J. Phys. Chem.* 84, 1871.
9) Arden-Jacob J., Marx N. J. and Drexhage K. H. (1997) *J. Fluorescence* 7(Suppl.), 91S.
10) Magde D., Brannon J. H., Cramers T. L. and Olmsted J. III (1979) *J. Phys. Chem.* 83, 696.

But you should be aware that quantum yields are notoriously difficult to measure. For example, “reliable” literature values for quinine sulfate range from ~0.50 – 0.65!

Polarization

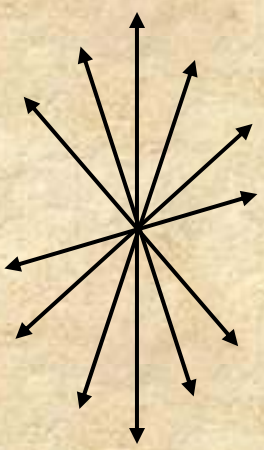
Light can be considered as oscillations of an electromagnetic field – characterized by electric and magnetic components - perpendicular to the direction of light propagation.



In these lectures we shall be concerned only with the electric component.

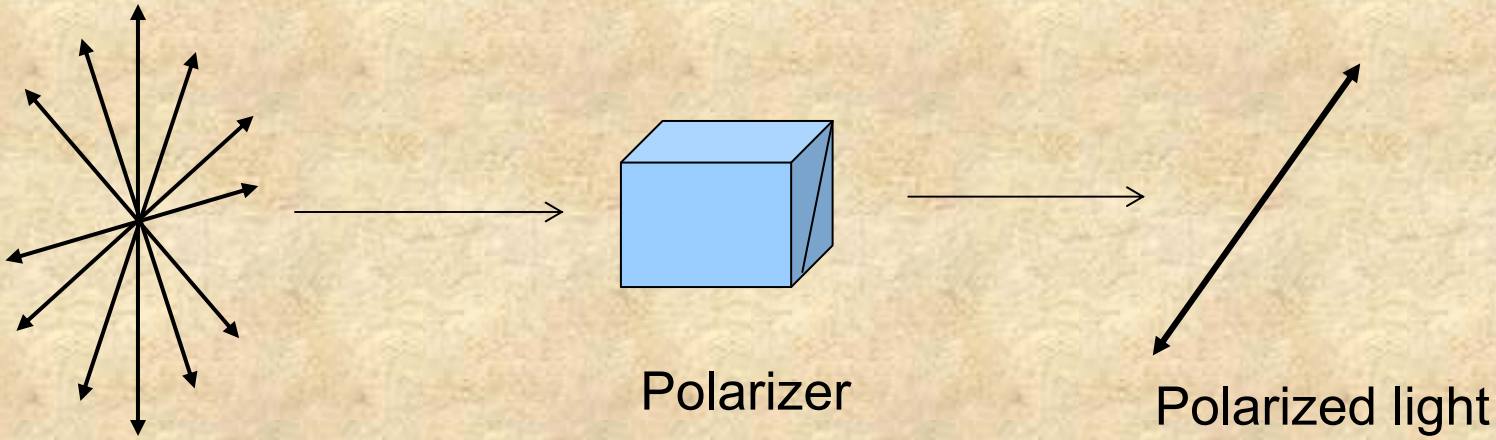
In natural light the electric field vector can assume any direction of oscillation perpendicular or normal to the light propagation direction.

Unpolarized (natural) light



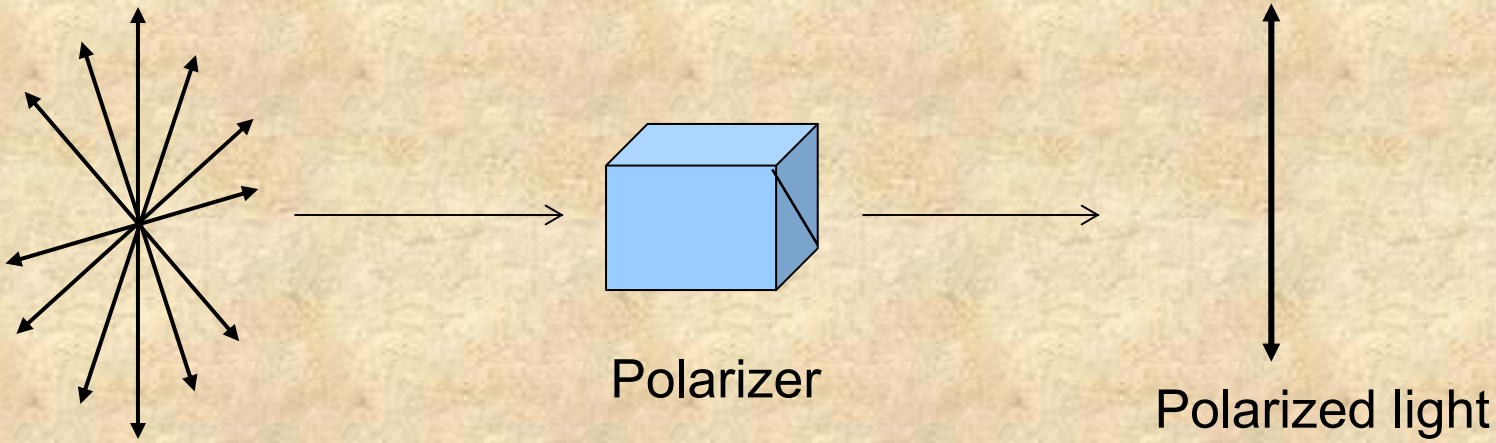
Light Propagation Direction

Polarizers are optically active devices that can isolate one direction of the electric vector.



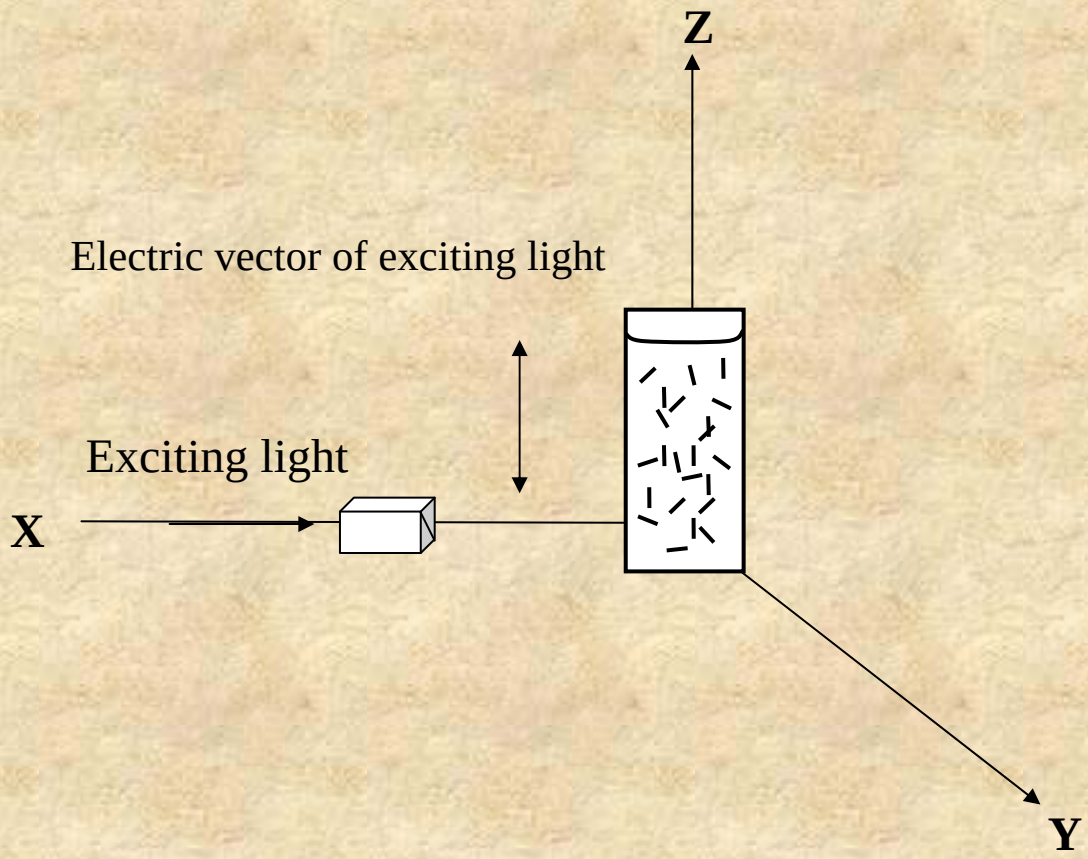
Unpolarized (natural) light

Polarizers are optically active devices that can isolate one direction of the electric vector.



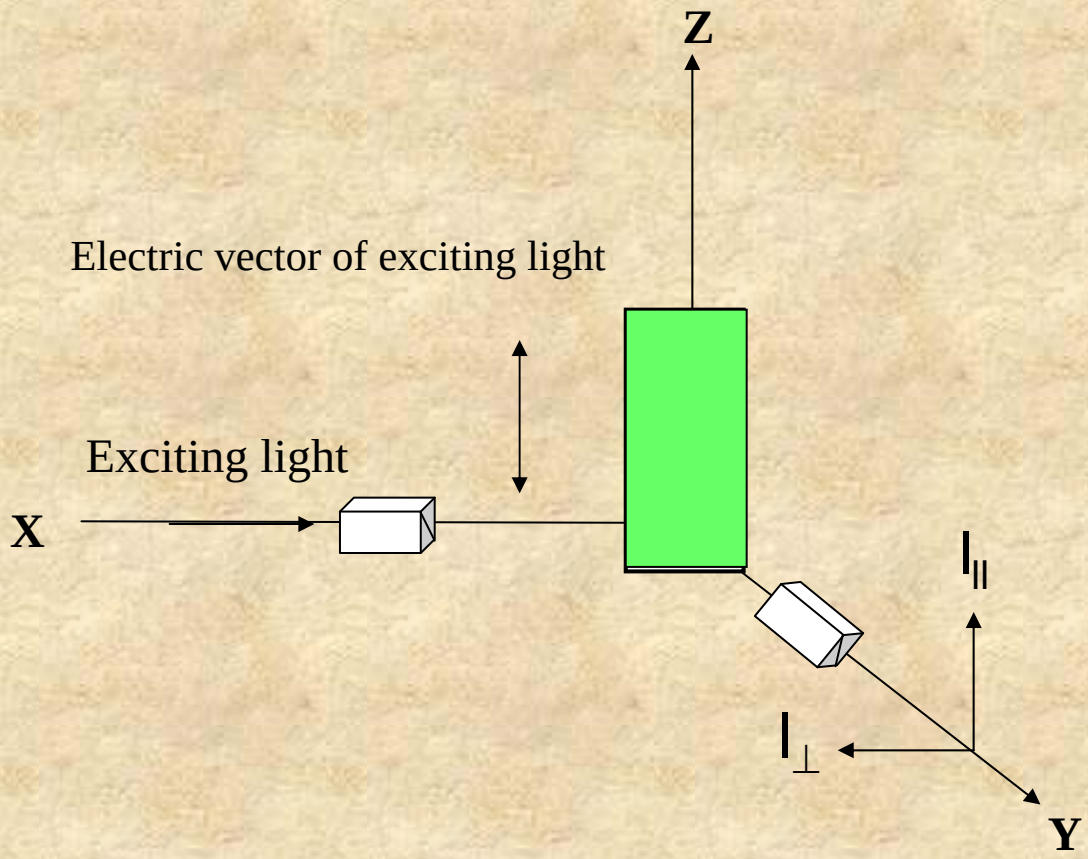
The most common polarizers used today are (1) dichroic devices, which operate by effectively absorbing one plane of polarization (e.g., Polaroid type-H sheets based on stretched polyvinyl alcohol impregnated with iodine) and (2) double refracting calcite ($CaCO_3$) crystal polarizers - which differentially disperse the two planes of polarization (examples of this class of polarizers are Nicol polarizers, Wollaston prisms and Glan-type polarizers such as the Glan-Foucault, Glan-Thompson and Glan-Taylor polarizers)

Consider an XYZ coordinate framework with a fluorescent solution placed at the origin, as shown below, where XZ is in the plane of the page.



In this system, the exciting light is traveling along the X direction. If a polarizer is inserted in the beam, one can isolate a unique direction of the electric vector and obtain light polarized parallel to the Z axis which corresponds to the vertical laboratory axis.

This exciting light will be absorbed by the fluorophore at the origin and give rise to fluorescence which is typically observed at 90^{o} to the excitation direction, i.e., from along the Y axis.



The actual direction of the electric vector of the emission can be determined by viewing the emission through a polarizer which can be oriented alternatively in the parallel or perpendicular direction relative to the Z axis or laboratory vertical direction.

Polarization is then defined as a function of the observed parallel (I_{II}) and perpendicular intensities ($I_{\perp}$) :

$$P = \frac{I_{II} - I_{\perp}}{I_{II} + I_{\perp}}$$

If the emission is completely polarized in the parallel direction, i.e., the electric vector of the exciting light is totally maintained, then:

$$P = \frac{1-0}{1+0} = 1$$

If the emitted light is totally polarized in the perpendicular direction then:

$$P = \frac{0-1}{0+1} = -1$$

The limits of polarization are thus +1 to -1

Another term frequently used in the context of polarized emission is anisotropy (usually designated as either A or r) which is defined as:

$$r = \frac{I_{||} - I_{\perp}}{I_{||} + 2I_{\perp}}$$

By analogy to polarization, the limits of anisotropy are +1 to -0.5.

A comment about the difference between polarization and anisotropy:

Given the definition of polarization and anisotropy, one can show that:

$$r = \frac{2}{3}\left(\frac{1}{P} - \frac{1}{3}\right)^{-1} \quad \text{or} \quad r = \frac{2P}{3 - P}$$

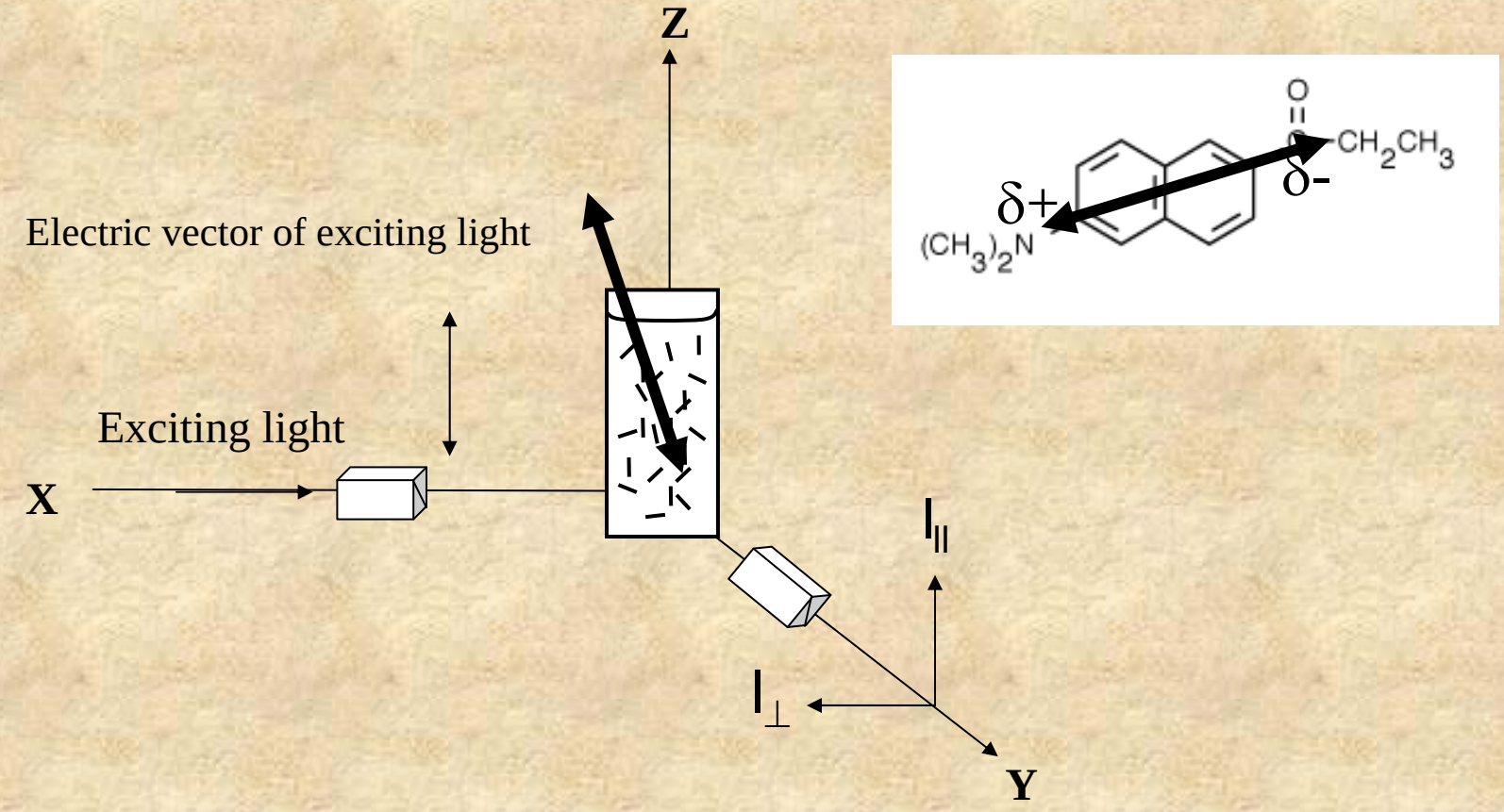
For example:

P	r
0.50	0.40
0.30	0.22
0.10	0.069

Clearly, the information content in the polarization function and the anisotropy function is identical and the use of one term or the other is dictated by practical considerations as will be discussed later.

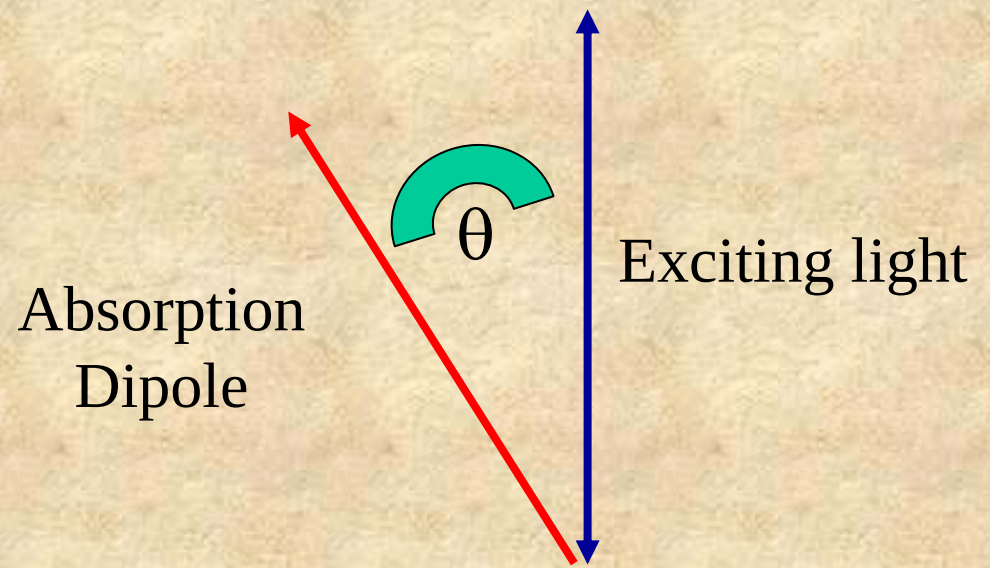
In solution these limits (e.g., +/-1) are not realized. Consider, as shown below, fluorophores at the origin of our coordinate system.

Upon absorption of an exciting photon a dipole moment is created in the fluorophore (usually of different magnitude and direction from the ground state dipole). The orientation of this dipole moment relative to the nuclear framework, and its magnitude, will be determined by the nature of the substituents on the molecule. This excited state dipole moment is also known as the transition dipole or transition moment.

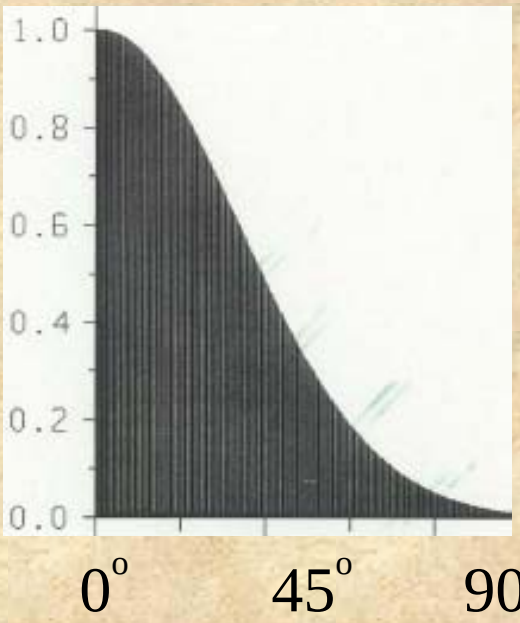


In fact, if light of a particular electric vector orientation (plane polarized light) impinges on a sample, only those molecules which are properly oriented relative to this electric vector can absorb the light.

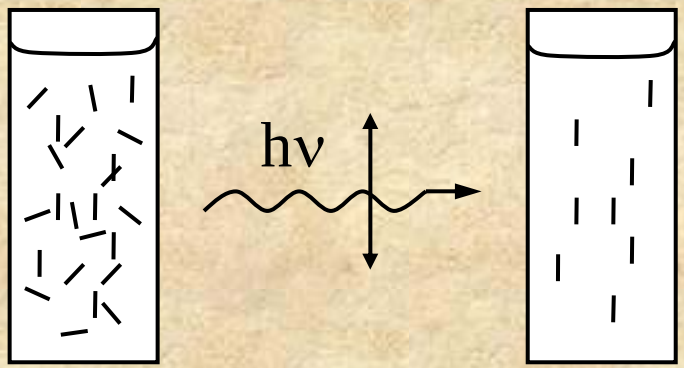
Specifically, the probability of the absorption is proportional to the cosine squared ($\cos^2\theta$) of the angle θ between the exciting light and the transition dipole.

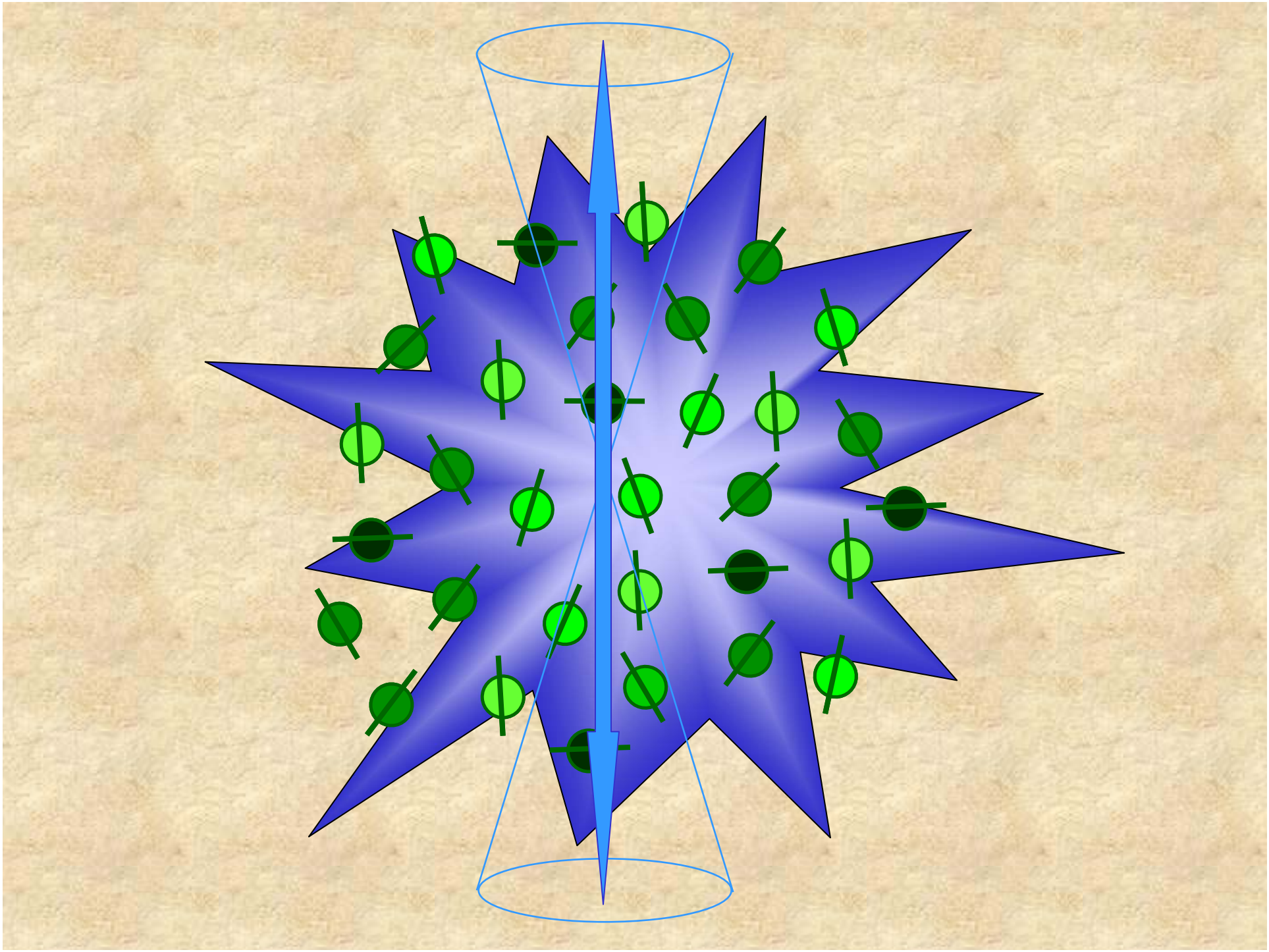


$\cos^2\theta$ function

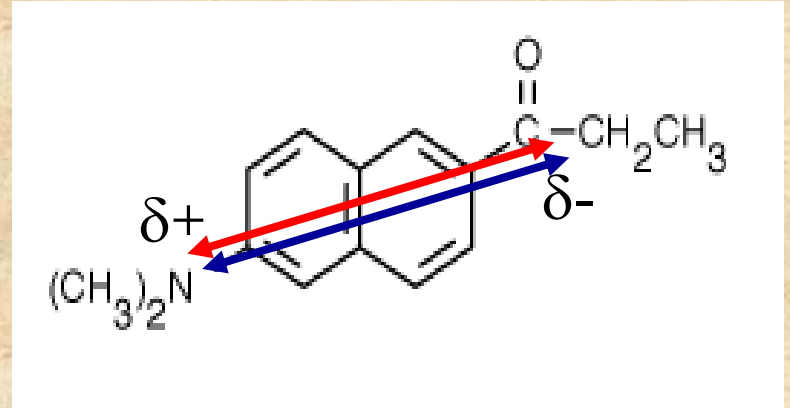


Hence, when we excite an ensemble of randomly oriented fluorophores with plane-polarized light we are performing a *photoselection* process, creating a population of excited molecules which nominally have their excited dipoles lined up with the polarization direction of the excitation. This process is illustrated below:

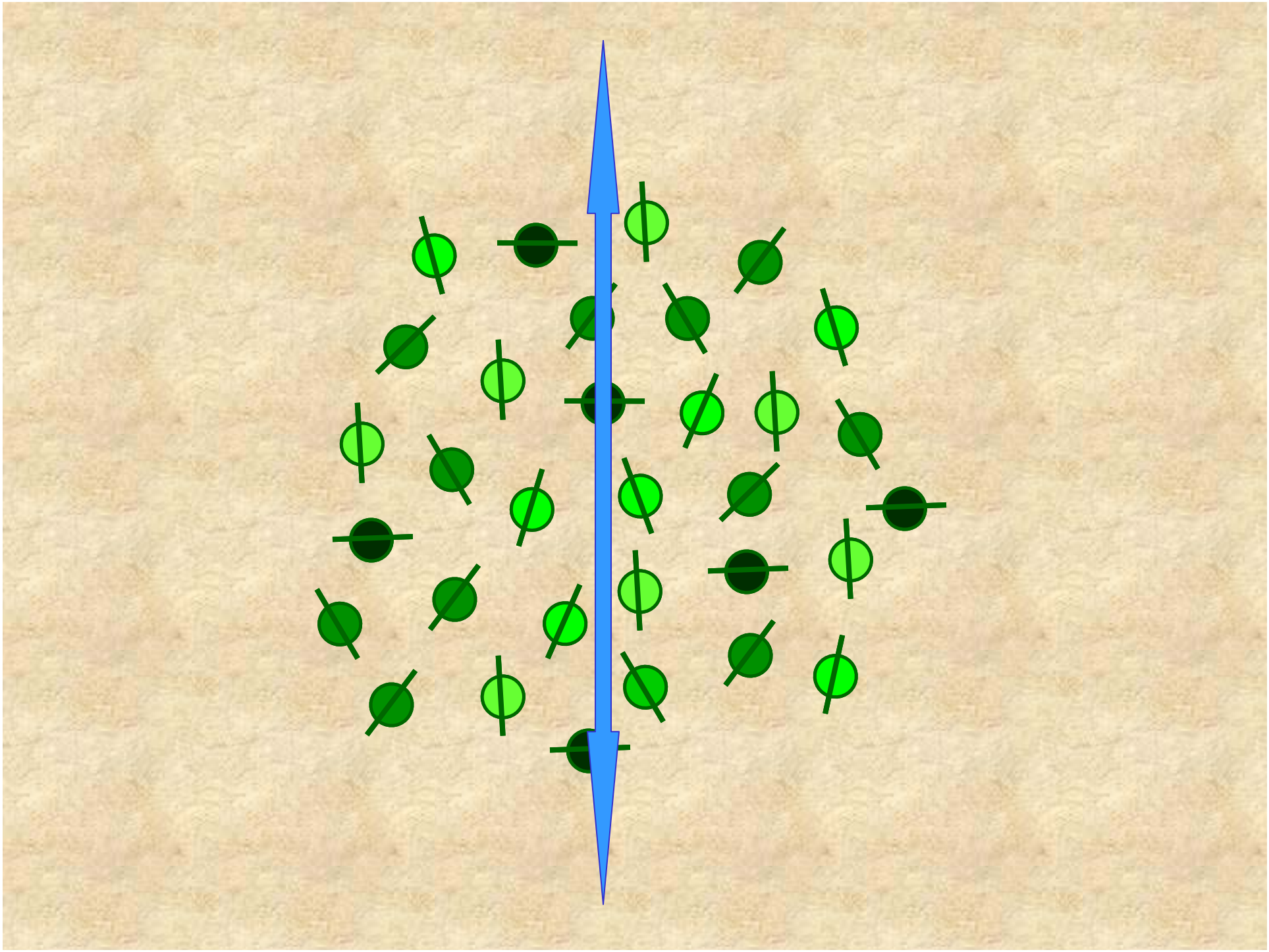




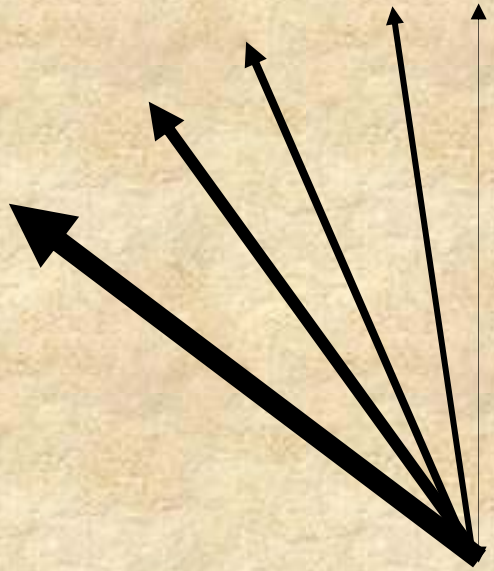
Consider now that the transition dipole corresponding to the emission of light from the excited fluorophore is parallel to the absorption dipole and that the excited fluorophore cannot rotate during the lifetime of the excited state (for example if the fluorophores are embedded in a highly viscous or frozen medium).



If we were to now measure the polarization of the emission it would be less than +1 since some of the dipoles excited will not be exactly parallel to the direction of the exciting light.



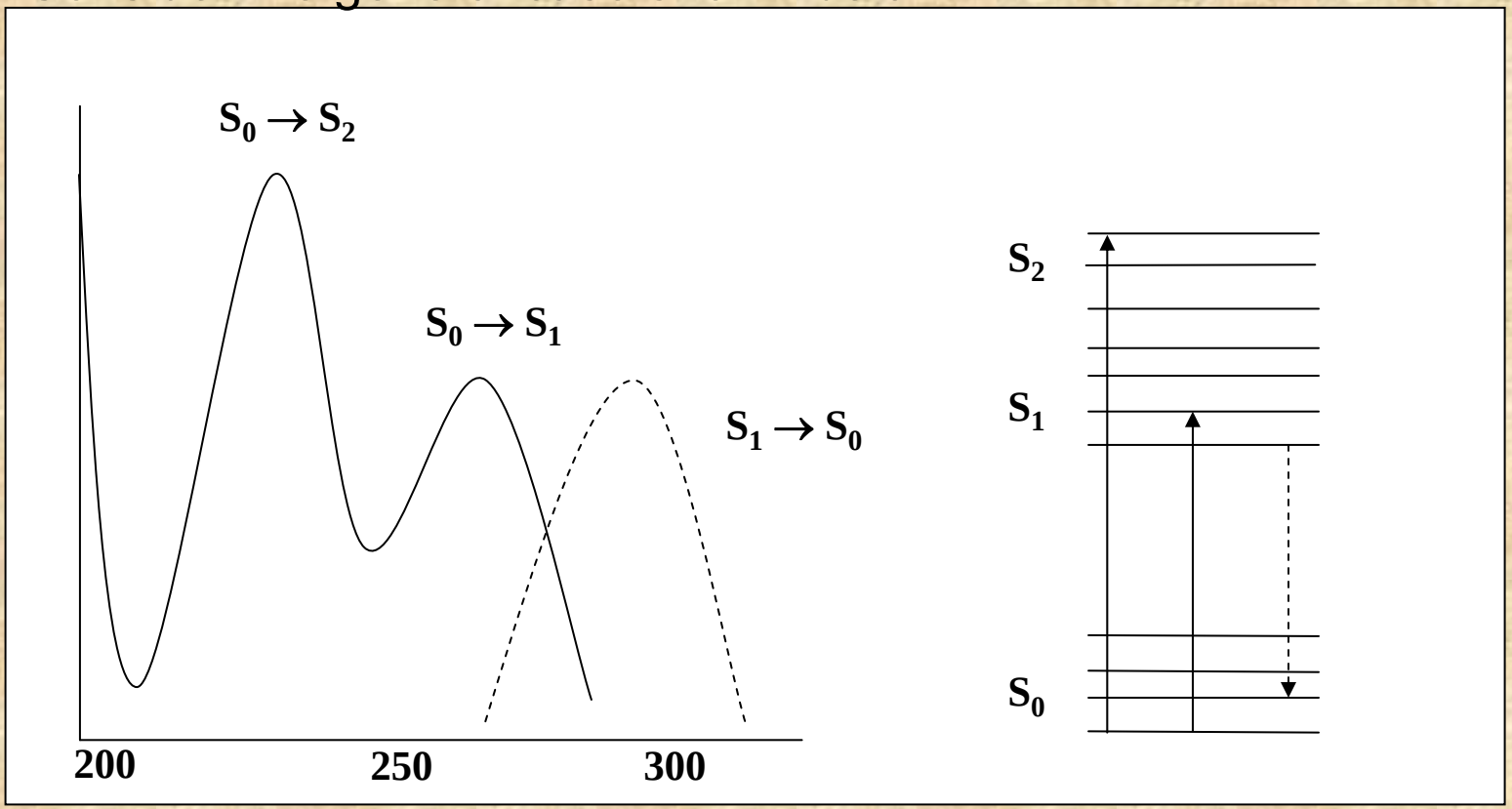
In fact, the number of potential dipoles making an angle θ with the vertical axis will be proportional to sin θ.



We can then calculate that the upper polarization limit for such a randomly oriented (but rigidly fixed, i.e., non-rotating) ensemble - with co-linear excitation and emission dipole - will be +1/2

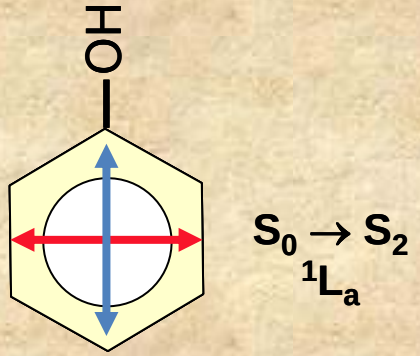
This case, however, assumes that the emission dipole is parallel (co-linear) to the absorption dipole.

Consider the general case shown below:



Here are depicted two principle absorption bands for phenol along with and the emission band. The energy level diagram corresponding to this system is also depicted.

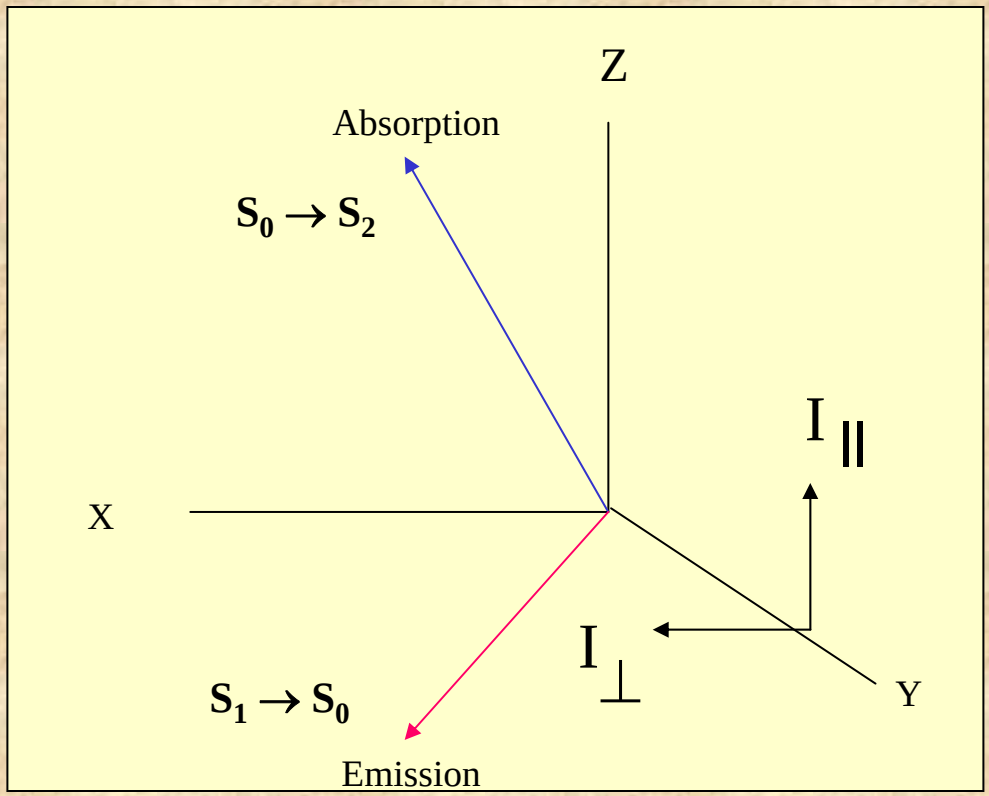
The directions of the absorption dipoles – relative to the nuclear framework – may differ greatly for the two transitions as illustrated on the right.



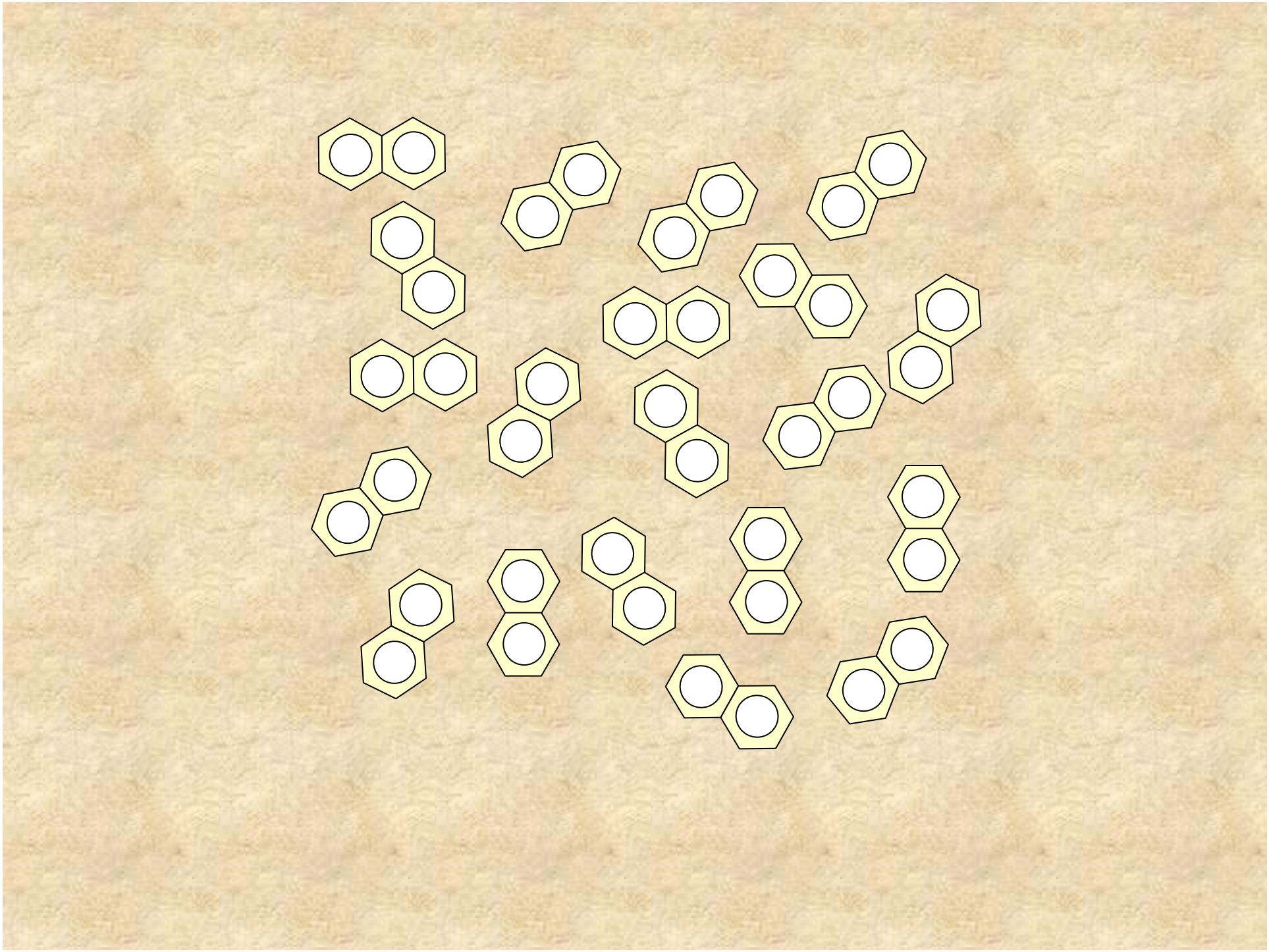
So we see that the two excited dipoles corresponding to the $S_0 \rightarrow S_1$ and the $S_0 \rightarrow S_2$ transitions may be oriented at an arbitrary angle - in the extreme case this angle could be 90^o.

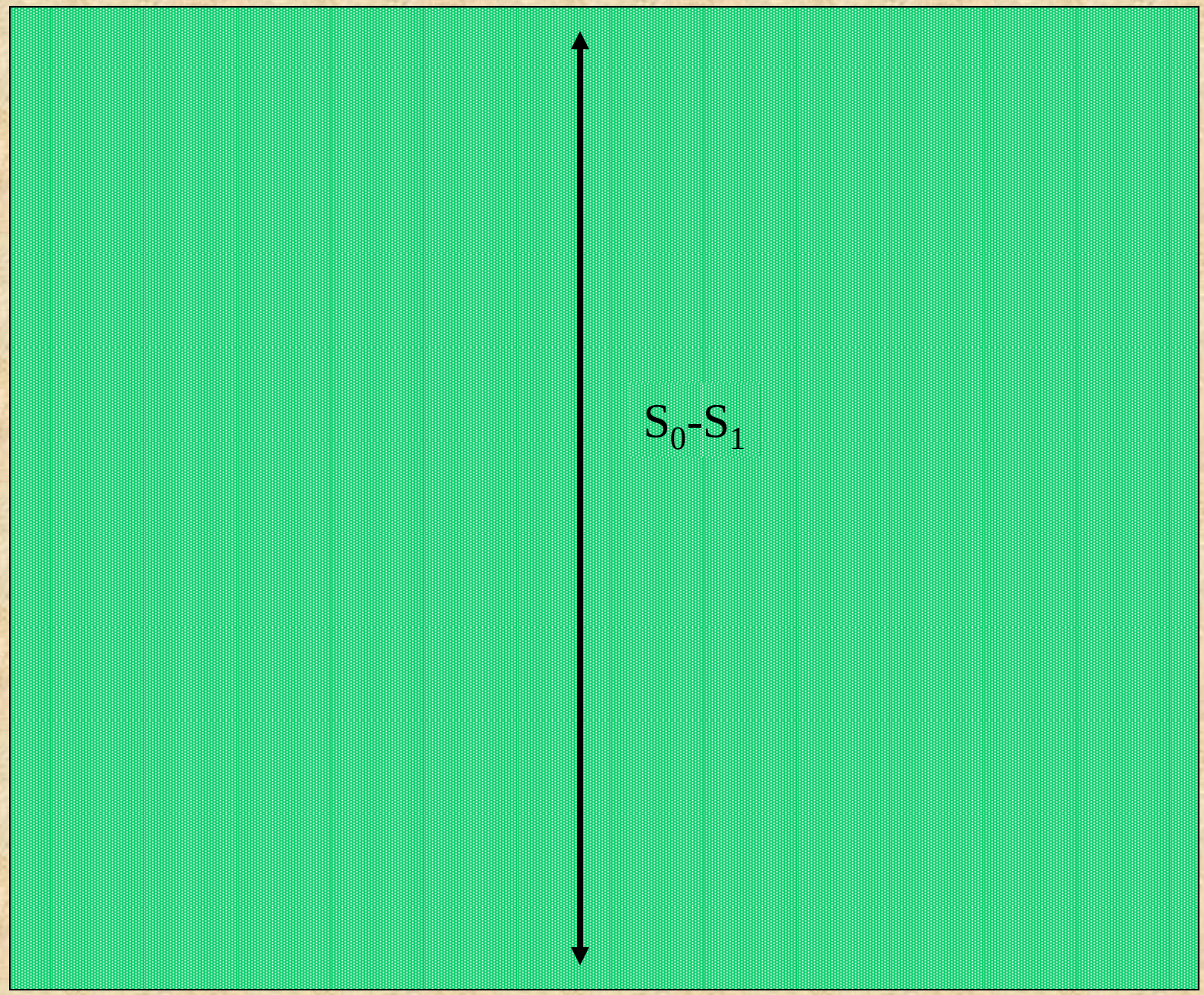
After the excitation process, however, regardless of whether the absorption process corresponded to the S0 $\rightarrow$ S1 or the S0 $\rightarrow$ S2 transition, rapid thermalization leaves the excited fluorophore in the S1 level.

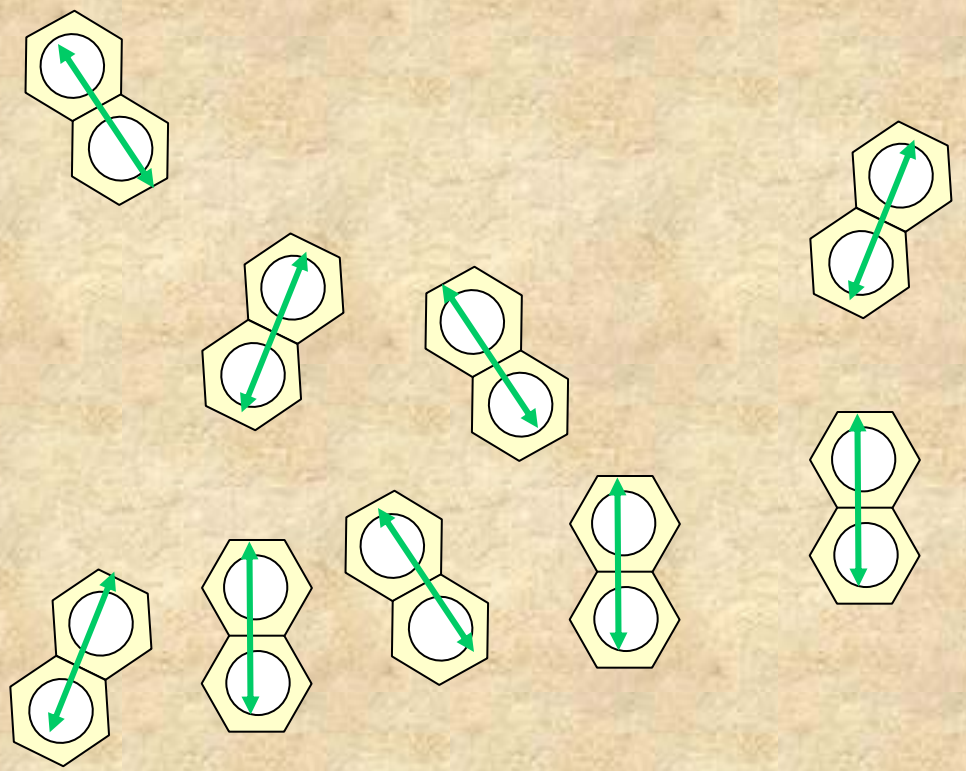
This situation is depicted below:

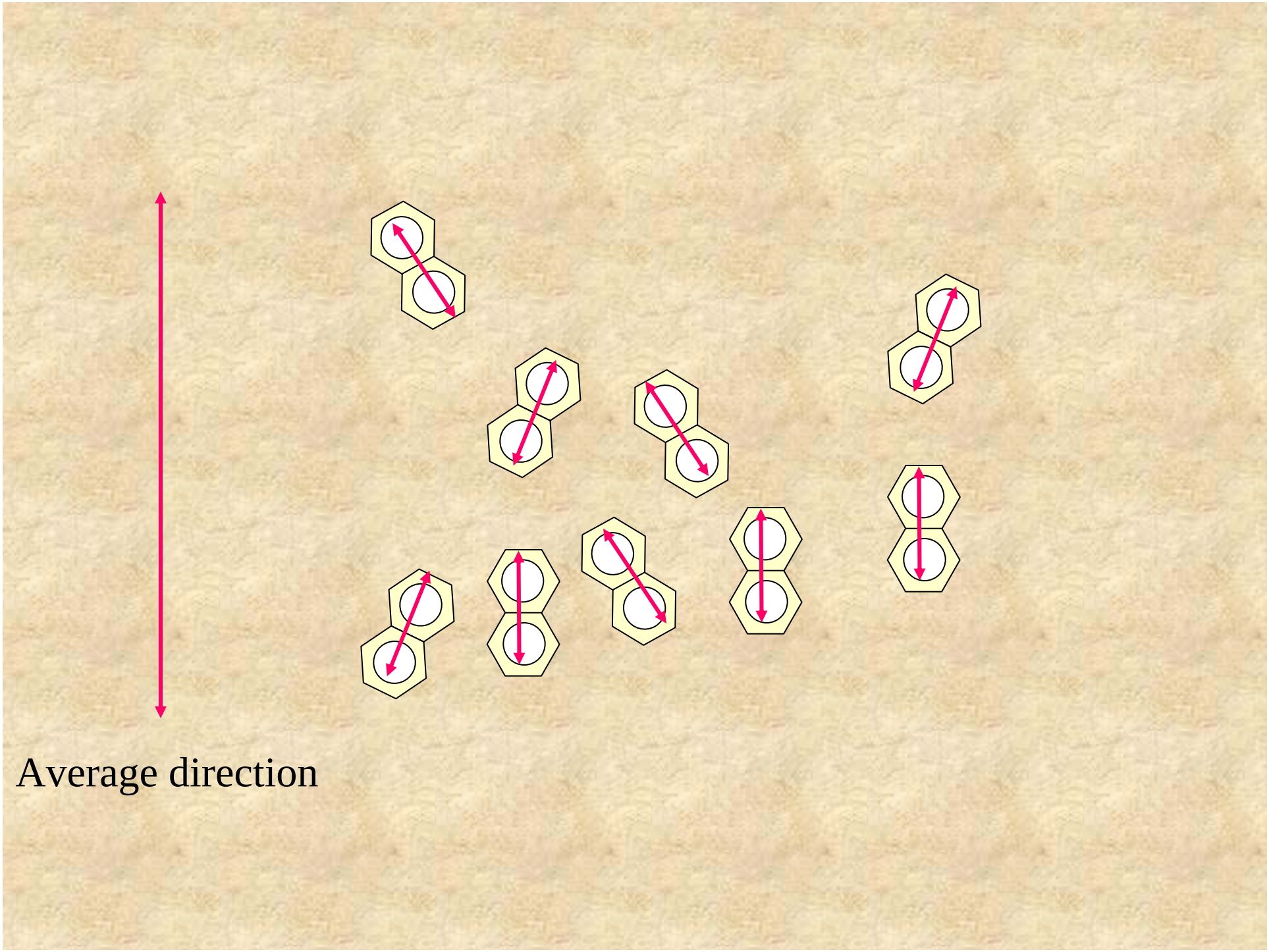


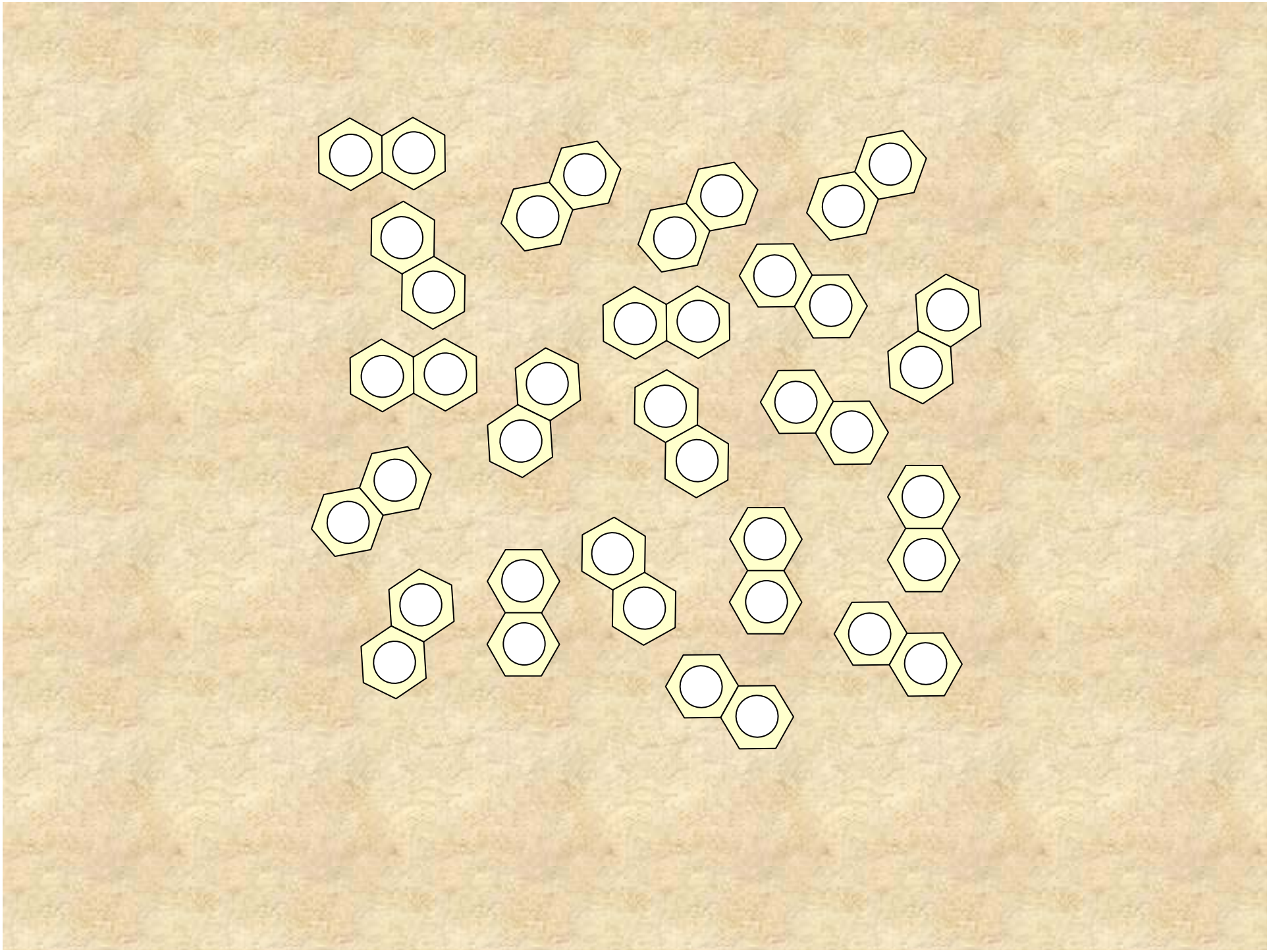
The orientation of the excited dipoles will thus now possess a different average orientation than the absorption dipoles originally photoselected by the exciting light.

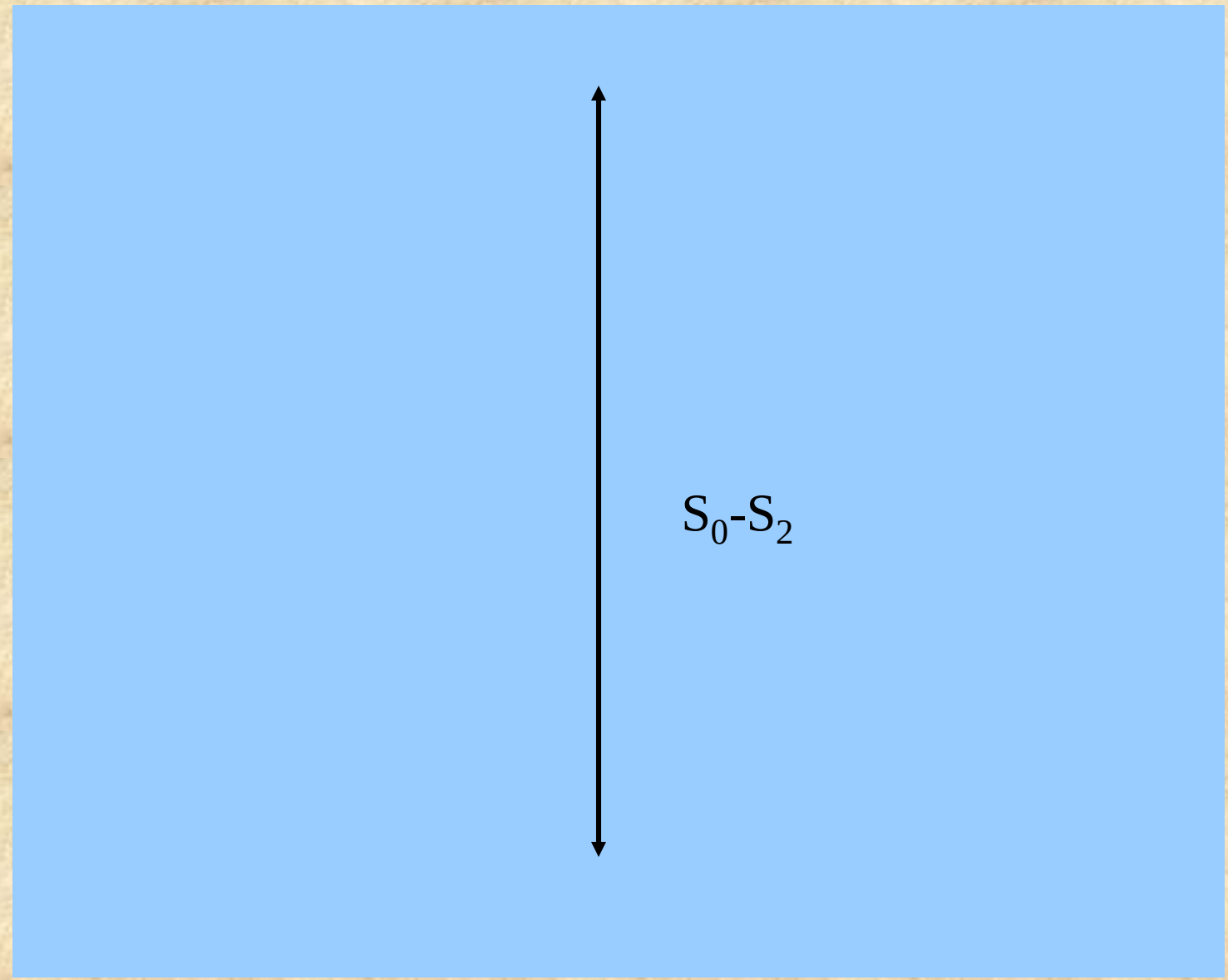


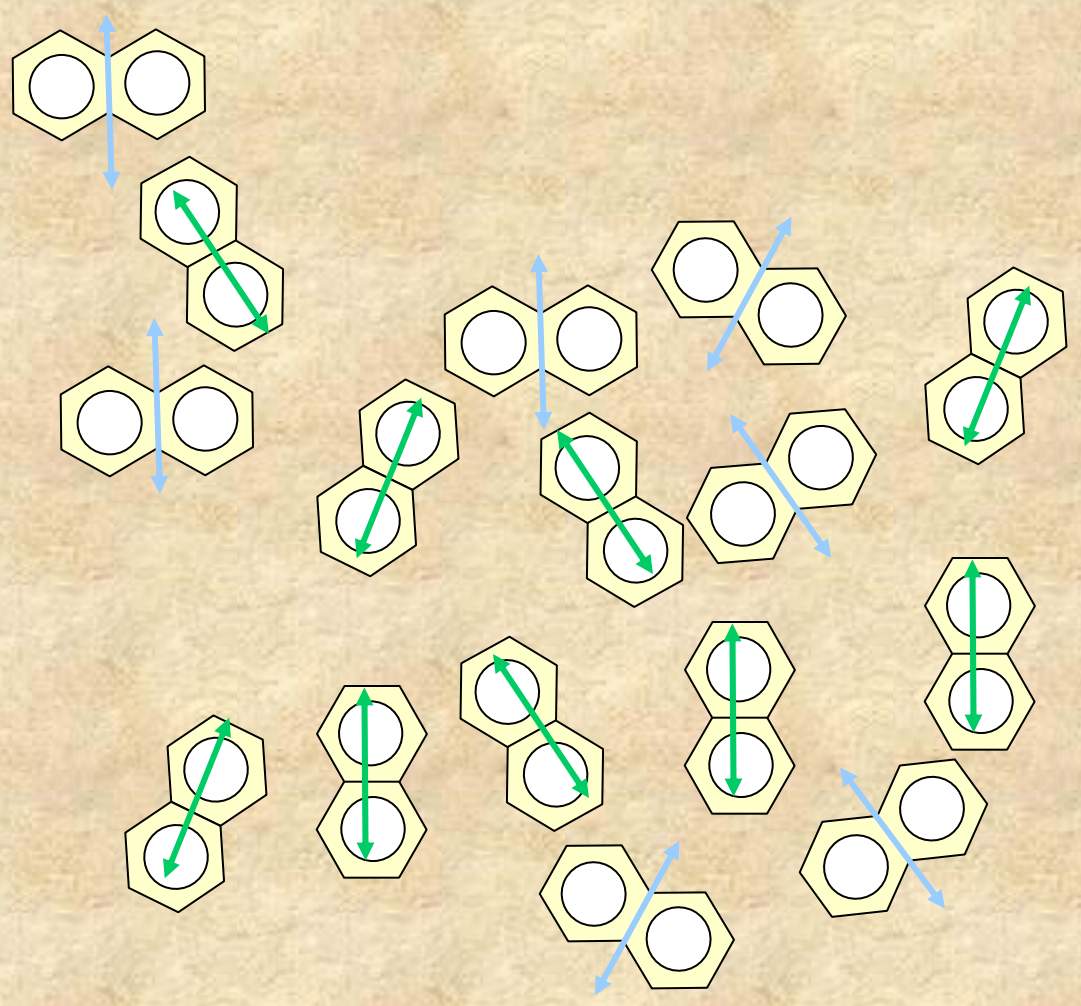

S_0-S_1

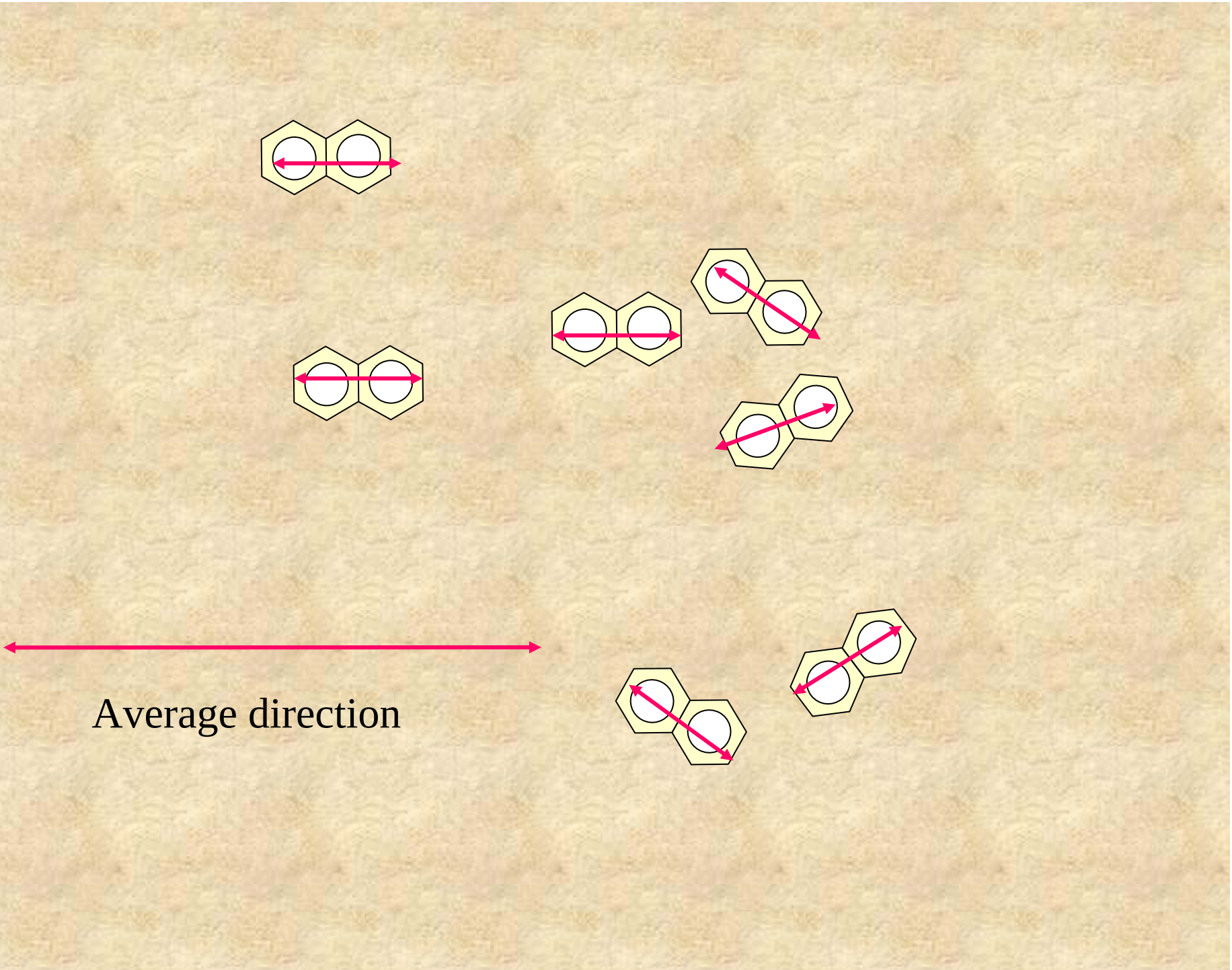



Average direction




S_0-S_2




Average direction

Hence we will observe more emission in the perpendicular direction than in the parallel direction and the resulting polarization will be negative. Considering the same $\cos^2\theta$ photoselection rule and the $\sin\theta$ population distribution as before we can show that, if the absorption and emission dipoles are at 90^o to each other, then P = -1/3.

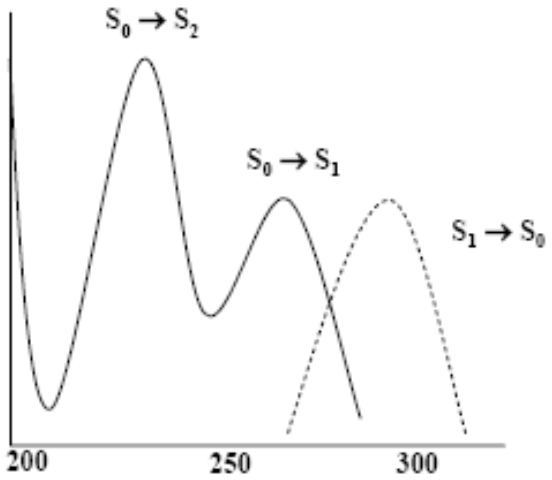
These polarization values, in the absence of rotation, are termed limiting or intrinsic polarizations and are denoted as P_o. In general:

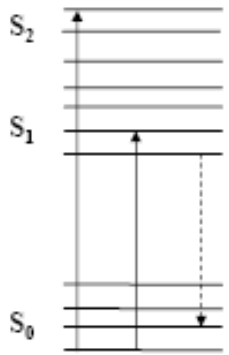
$$\frac{1}{P_o} - \frac{1}{3} = \frac{5}{3}\left(\frac{2}{3\cos^2\phi - 1}\right)$$

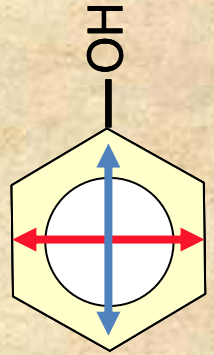
Where ϕ is the angle between absorption and emission dipoles.

We can then understand that the limiting polarization of a fluorophore will depend upon the excitation wavelength.

Consider the excitation polarization spectrum for phenol (in glycerol at - 70 C).







$S_0 \rightarrow S_2$
^1L_a

$S_0 \rightarrow S_1$
^1L_b

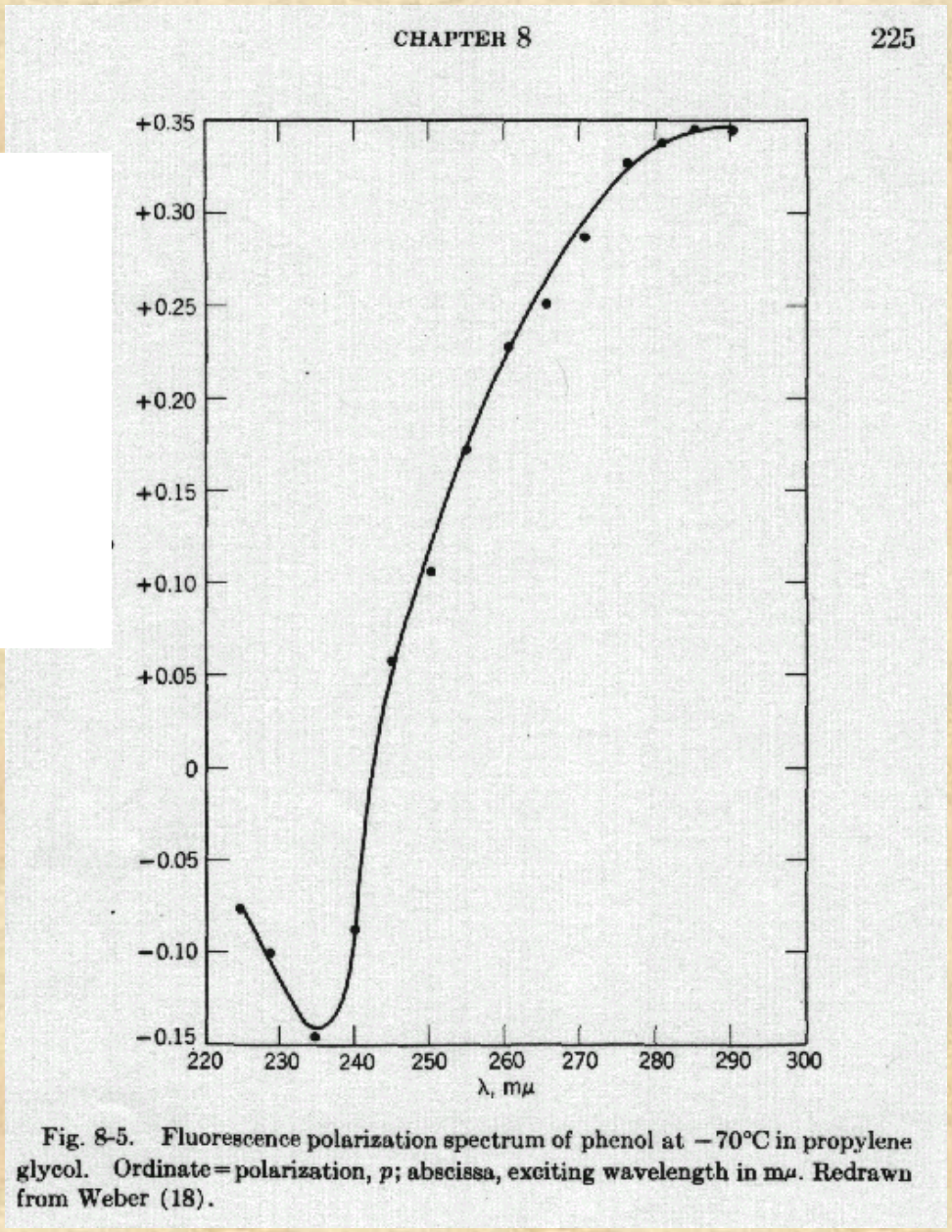


Fig. 8-5. Fluorescence polarization spectrum of phenol at −70°C in propylene glycol. Ordinate = polarization, p; abscissa, exciting wavelength in mμ. Redrawn from Weber (18).

In cases where there are multiple overlapping absorption bands at various angles, the excitation polarization spectrum can be somewhat complex as shown below for indole.

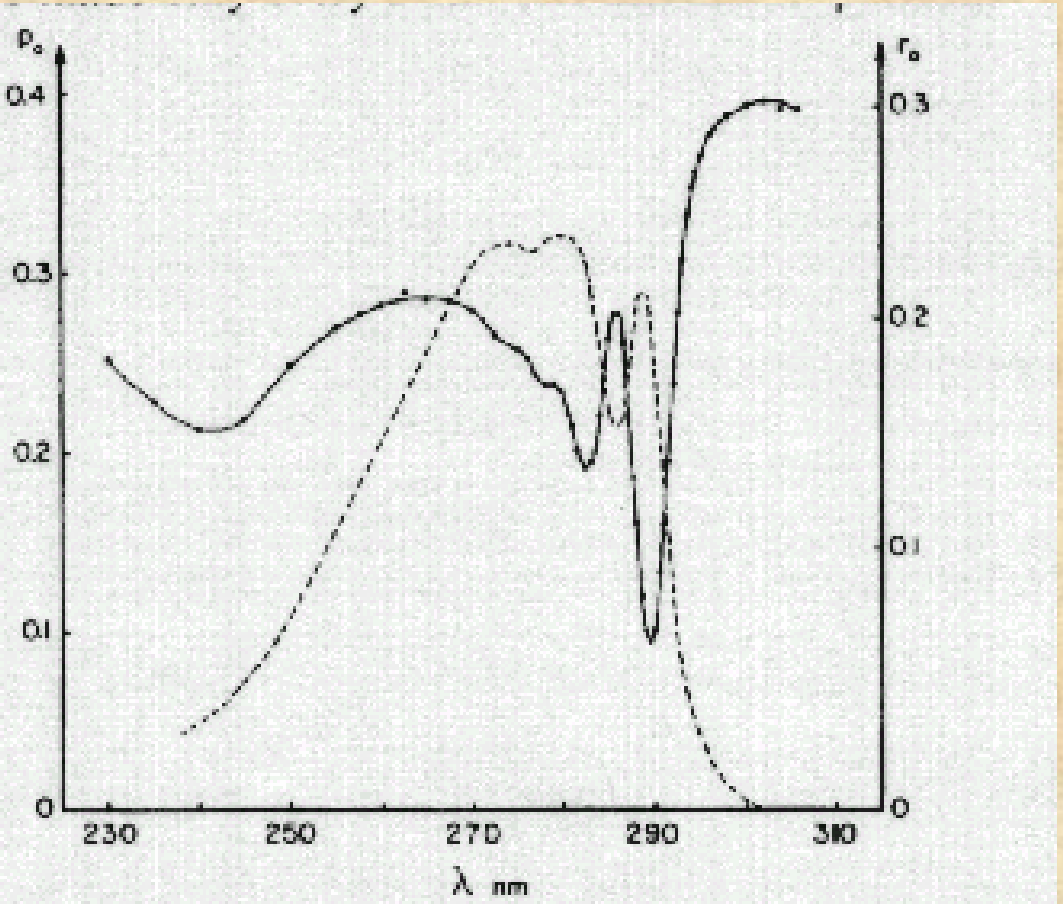
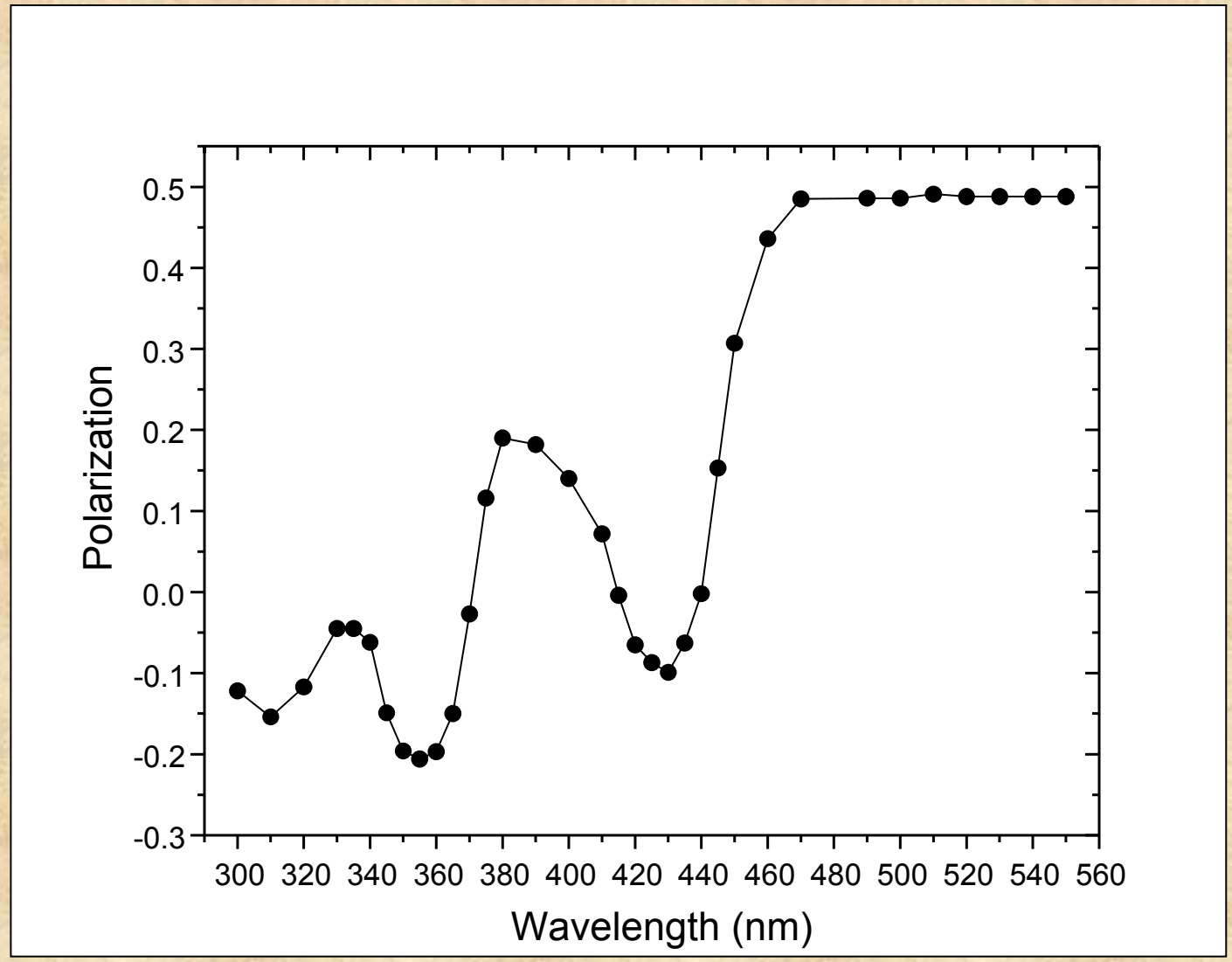
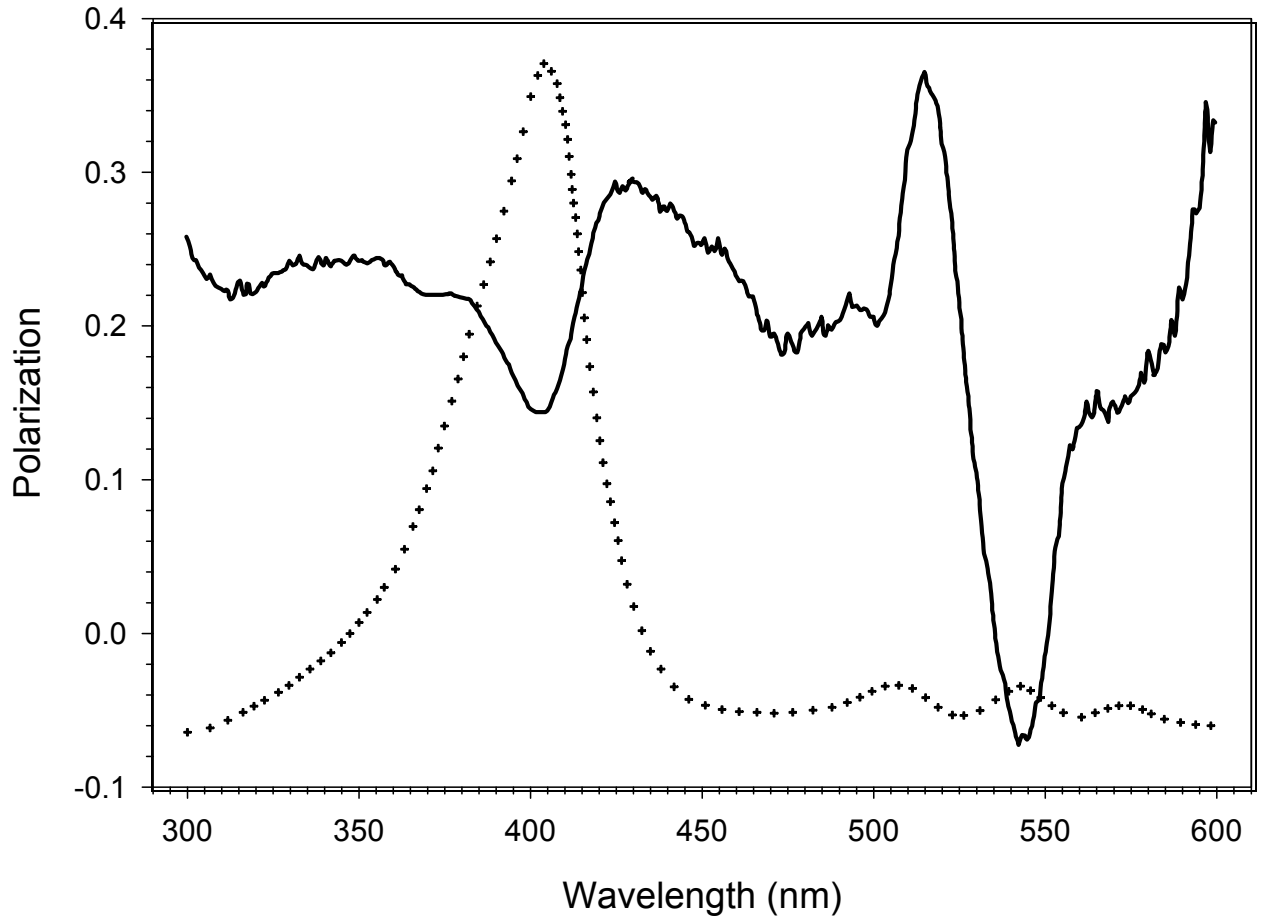


Figure 2. Corrected excitation spectrum (broken line) and excitation polarization spectrum of indole in propylene glycol at −58 . The fluorescence is observed through a Corning 7-39 filter.

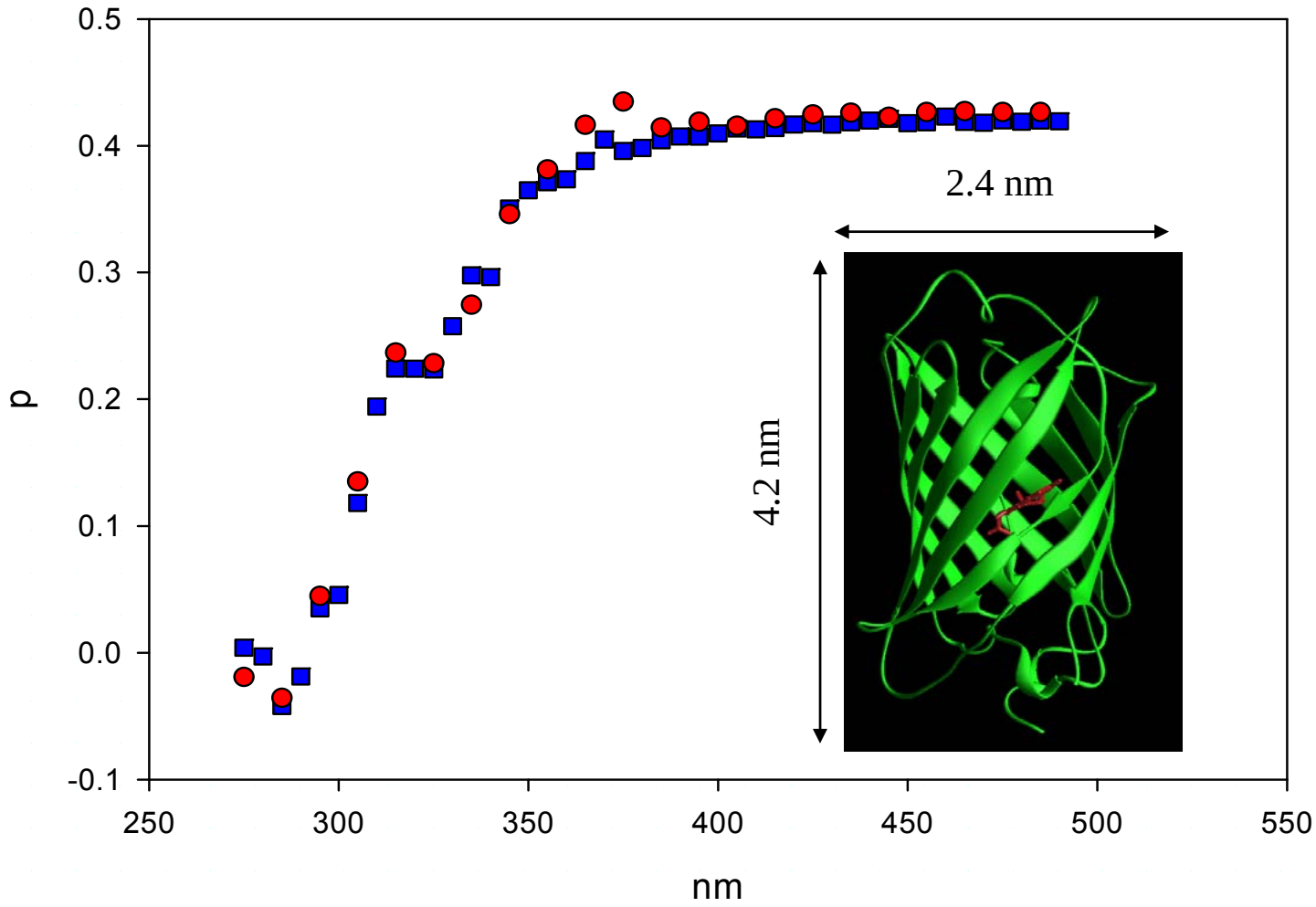
Excitation polarization spectra of rhodamine B embedded in a Lucite matrix at room temperature. Emission was viewed through a cut-on filter passing wavelengths longer than 560nm; slits were ~4nm.



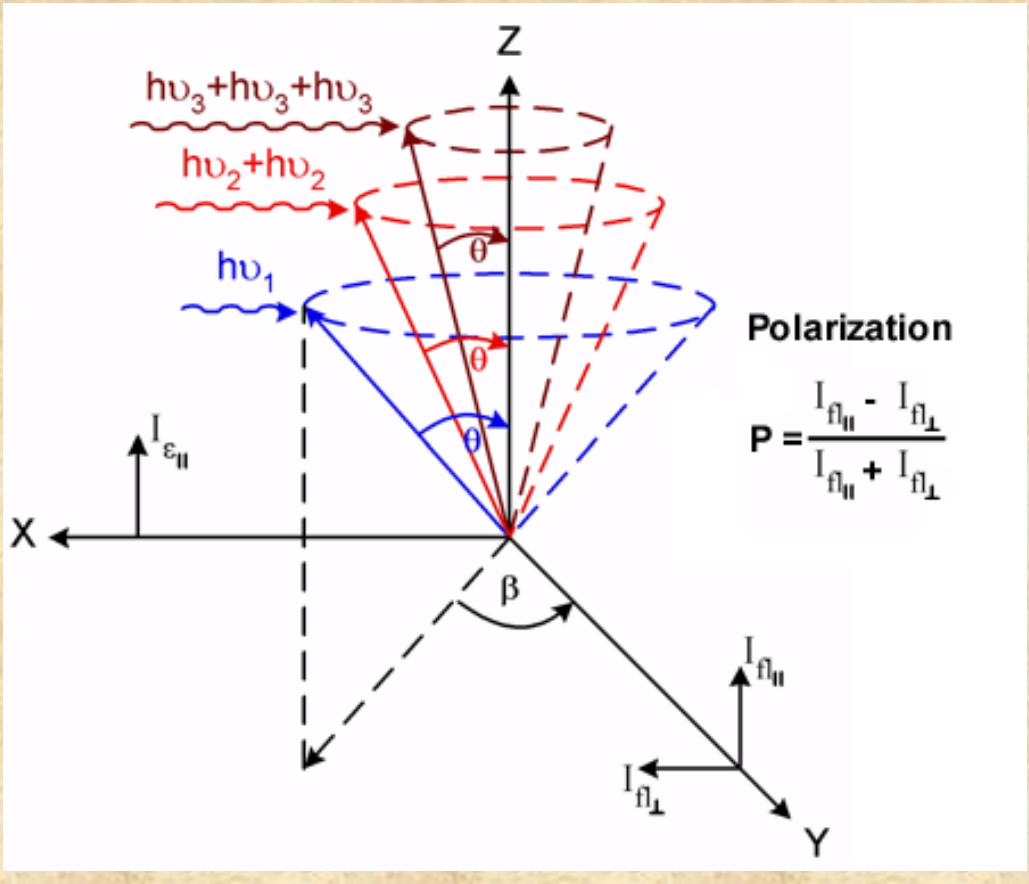
Another example is protoporphyrin IX in glycerol at –20C

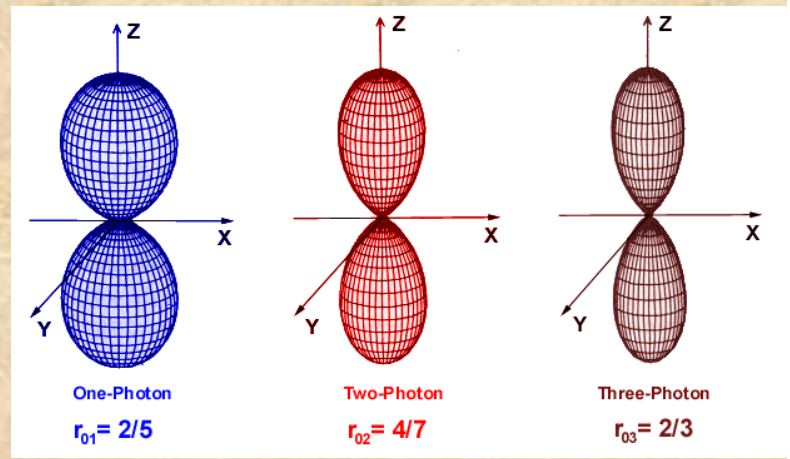


Excitation Polarization Spectrum of GFP
Emission 507nm

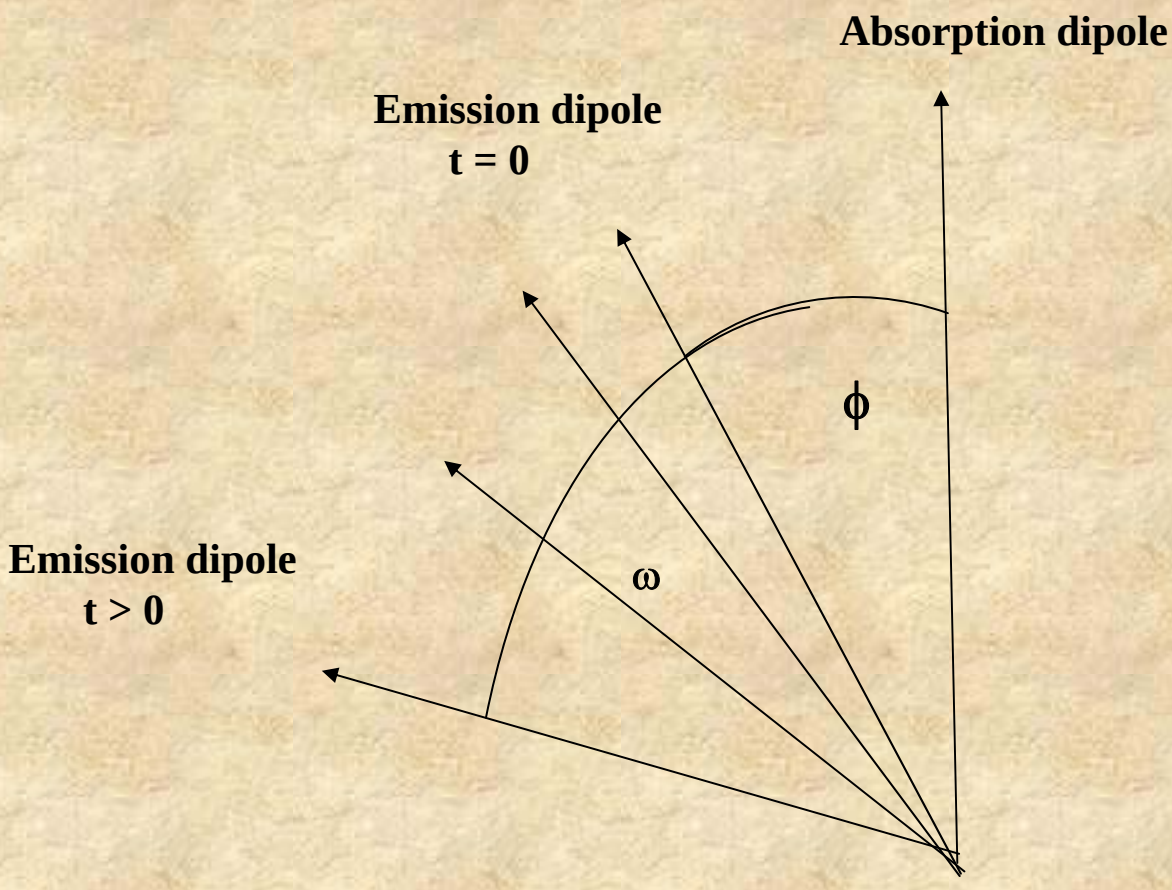


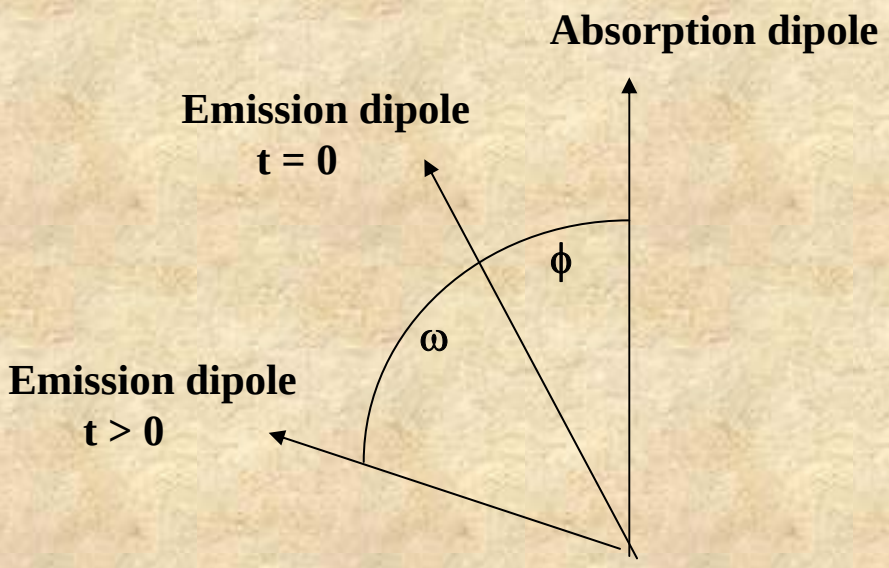
Note: in the case of multi-photon excitation the limits differ





We may now consider the case where the fluorophore is permitted to rotate during the excited state lifetime.



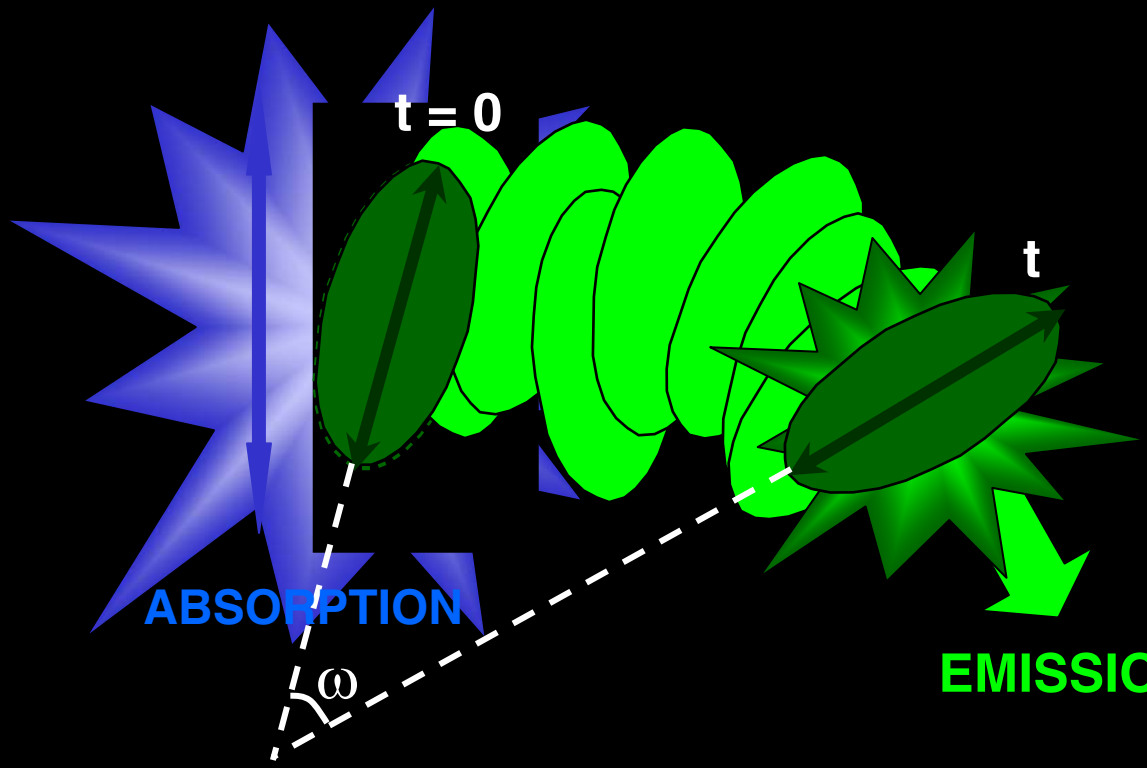


Additional depolarization occurs if the dipole rotates through an angle ω.

In fact:

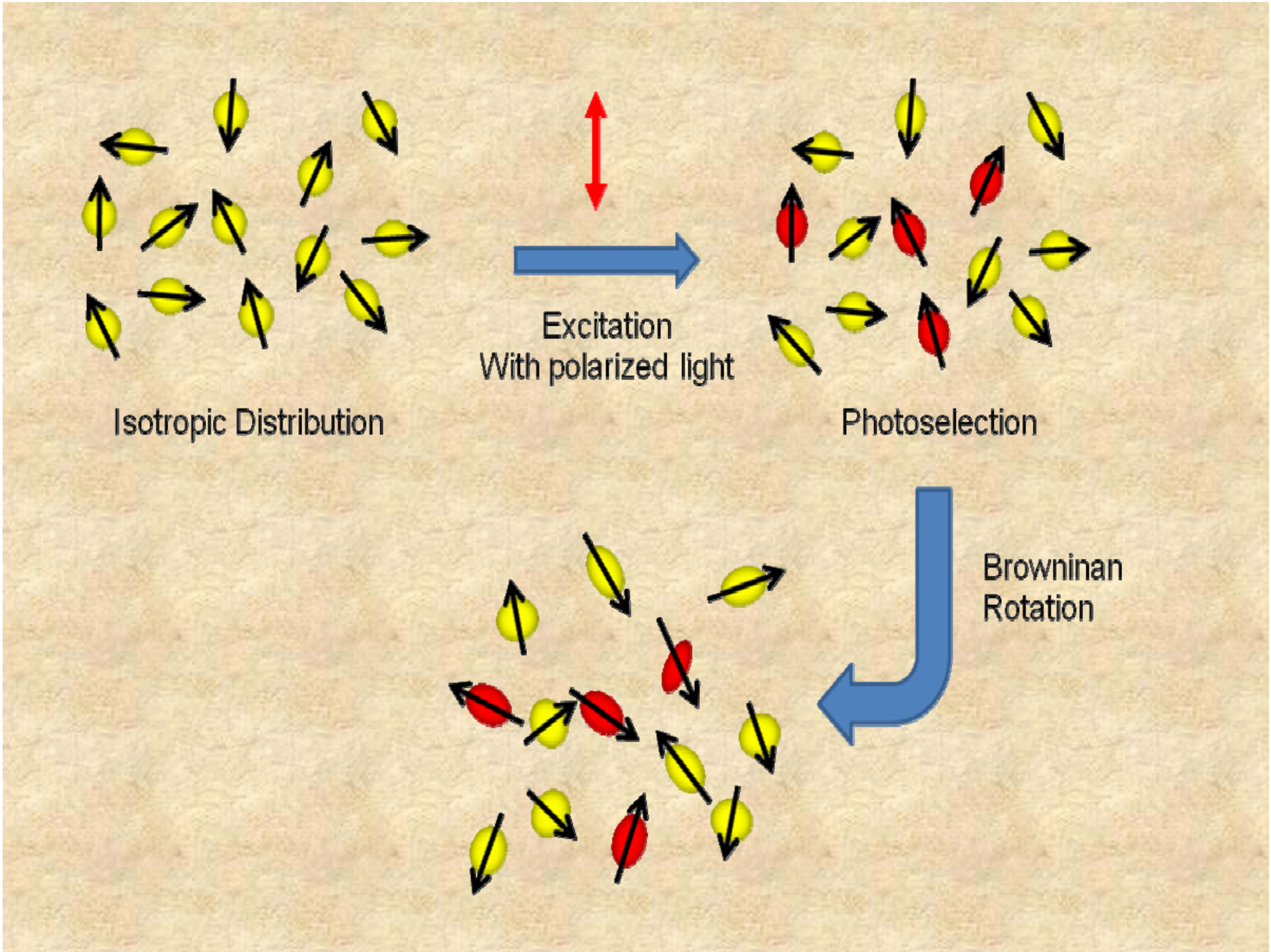
$$\frac{1}{P}-\frac{1}{3}=\left(\frac{1}{P_o}-\frac{1}{3}\right)\left(\frac{2}{3\cos^2\omega-1}\right)$$

where P is the observed polarization. So the total depolarization is determined by an intrinsic factor (P_o) and an extrinsic factor (ω).



- **ORIENTATION AUTOCORRELATION FUNCTION**
  probability that a molecule having a certain orientation at time zero is oriented at angle ω with respect to its initial orientation

$$\frac{\overline{3\cos^2\omega(t)-1}}{2} = \frac{r(t)}{r_0}$$


Isotropic Distribution
Excitation
With polarized light
Photoselection
Browninan
Rotation

F. Perrin related the observed polarization to the excited state lifetime and the rotational diffusion of a fluorophore: *Perrin, F. 1926. Polarisation de la Lumiere de Fluorescence. Vie Moyene des Molecules Fluorescentes. J. Physique. 7:390-401.*

Specifically:

$$\frac{1}{P}-\frac{1}{3}=\left(\frac{1}{P_o}-\frac{1}{3}\right)\left(1+\frac{RT}{\eta V}\tau\right)$$

where V is the molar volume of the rotating unit, R is the universal gas constant, T the absolute temperature, η the viscosity and τ the excited state lifetime.

We can rewrite this equation as:

$$\frac{1}{P}-\frac{1}{3}=\left(\frac{1}{P_o}-\frac{1}{3}\right)\left(1+\frac{3\tau}{\rho}\right)$$

Where ρ is the Debye rotational relaxation time which is the time for a given orientation to rotate through an angle given by the arccos e^{-1} (68.42^o).

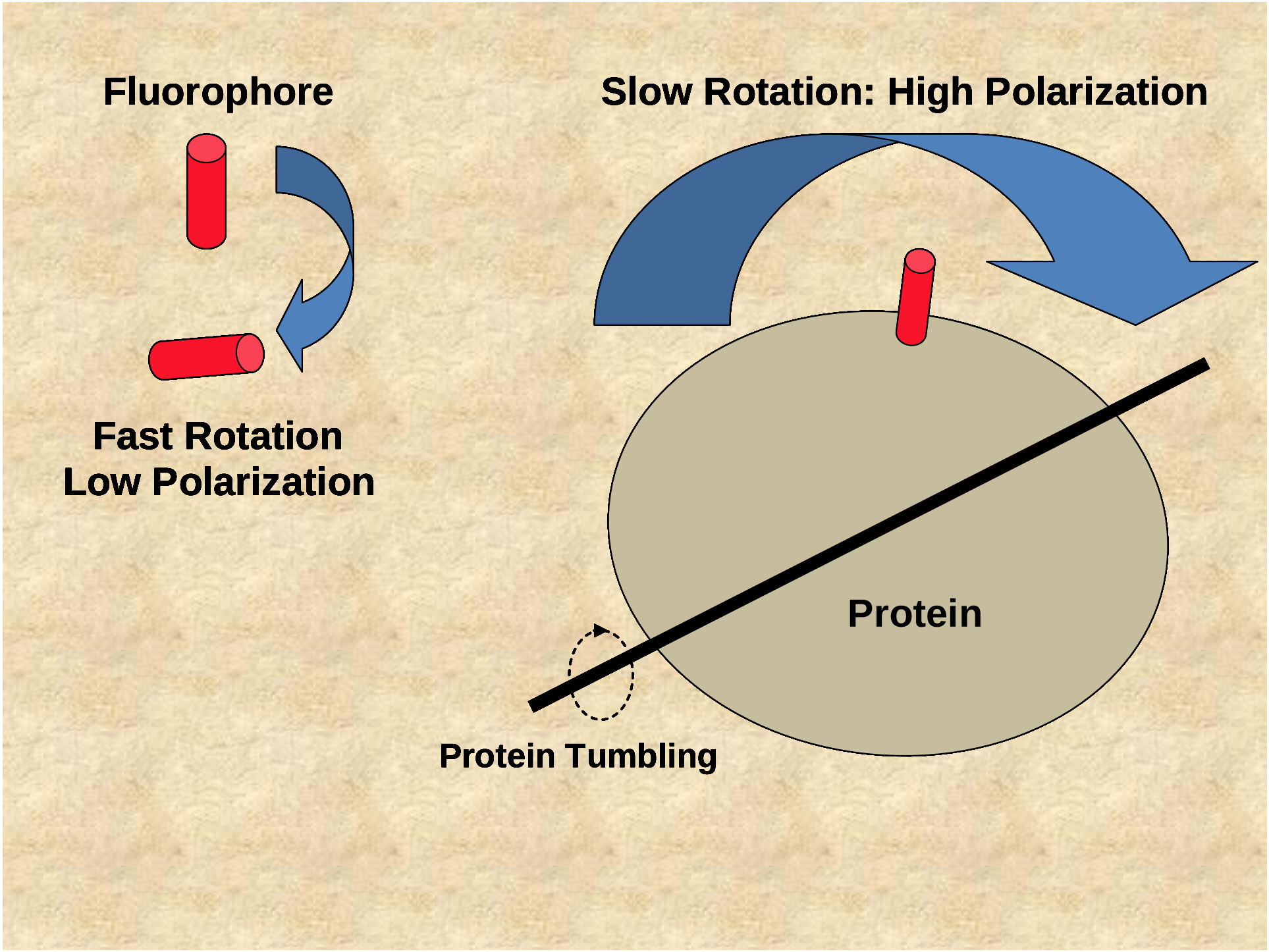
For a spherical molecule:

$$\rho_o = \frac{3\eta V}{RT}$$

For a spherical protein,
it follows that:

$$\rho_o = \frac{3\eta M(\upsilon + h)}{RT}$$

Where M is the molecular weight, ν is the partial specific volume and h the degree of hydration.


Fluorophore
Fast Rotation
Low Polarization
Slow Rotation: High Polarization
Protein
Protein Tumbling

* *Rotational relaxation time* versus *rotational correlation time*.

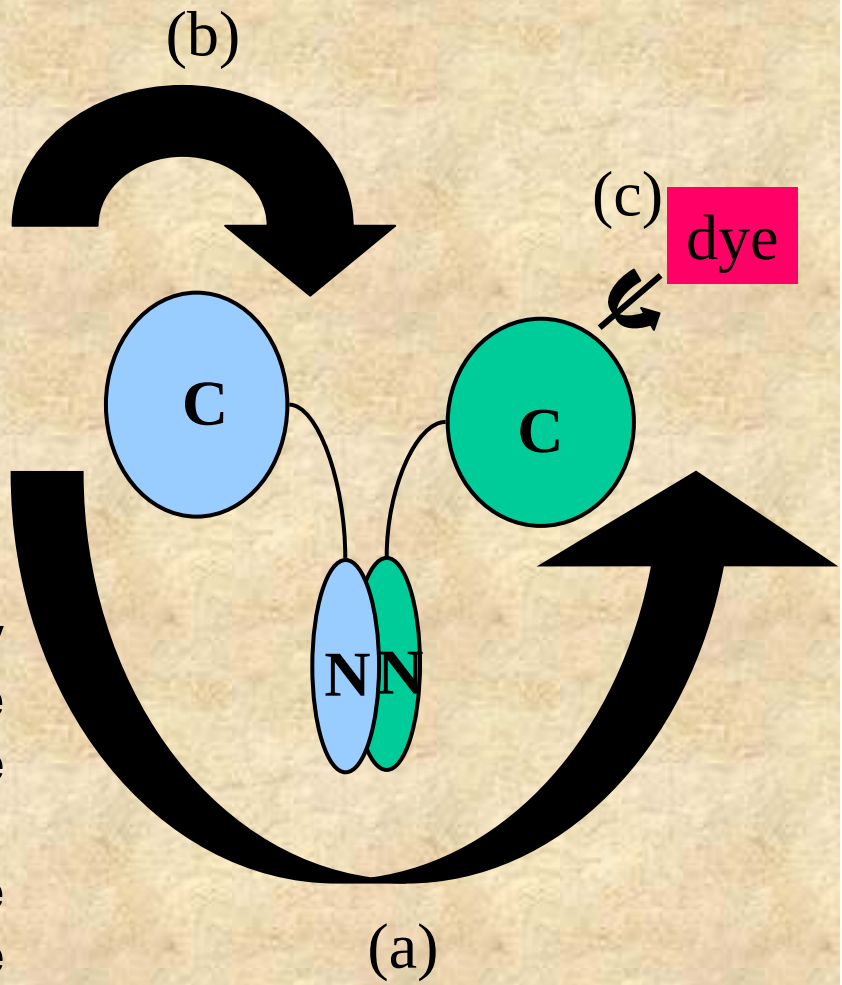
We should note that it is not uncommon to see the term "rotational correlation time", often denoted as τ_c, used in place of the Debye rotational relaxation time. The information content of these terms is similar since $\rho = 3\tau_c$ but we have observed that some people become rather fervently attached to the use of one term or the other.

In the original development of the theories of rotational motion of fluorophores Perrin and others used the rotational relaxation time, as originally defined by Debye in his studies on dielectric phenomena. Only later (in the 1950's) during the development of nuclear magnetic resonance was the term rotational correlation time used by Bloch. It thus seems reasonable for fluorescence practitioners to use ρ but certainly adoption of either term should not lead to confusion. In terms of anisotropy and rotational correlation times, then, the Perrin equation would be:

$$\frac{r_o}{r} = \left(1 + \frac{\tau}{\tau_c}\right)$$

In the case of fluorescence probes associated non-covalently with proteins, (for example porphryins, FAD, NADH or ANS to give but a few systems), the probe is held to the protein matrix by several points of attachment and hence its “local” mobility, that is, its ability to rotate independent of the overall “global” motion of the protein, is very restricted.

In the case of a probe attached covalently to a protein, via a linkage through an amine or sulfhydryl groups for example, or in the case of tryptophan or tyrosine sidechains, considerable “local” motion of the fluorophore can occur. In addition, the protein may consist of flexible domains which can rotate independent of the overall “global” protein rotation. This type of mobility hierarchy is illustrated on the right for the case of a probe covalently attached to a dimeric protein



Rotational Modalities

(a) overall dimer rotation

(b) movement of one C-domain relative to other domains

(c) movement of dye molecule around its point of attachment

In the case of fluorescence probes associated non-covalently with proteins, (for example porphryins, FAD, NADH or ANS to give but a few systems), the probe is held to the protein matrix by several points of attachment and hence its “local” mobility, that is, its ability to rotate independent of the overall “global” motion of the protein, is very restricted.

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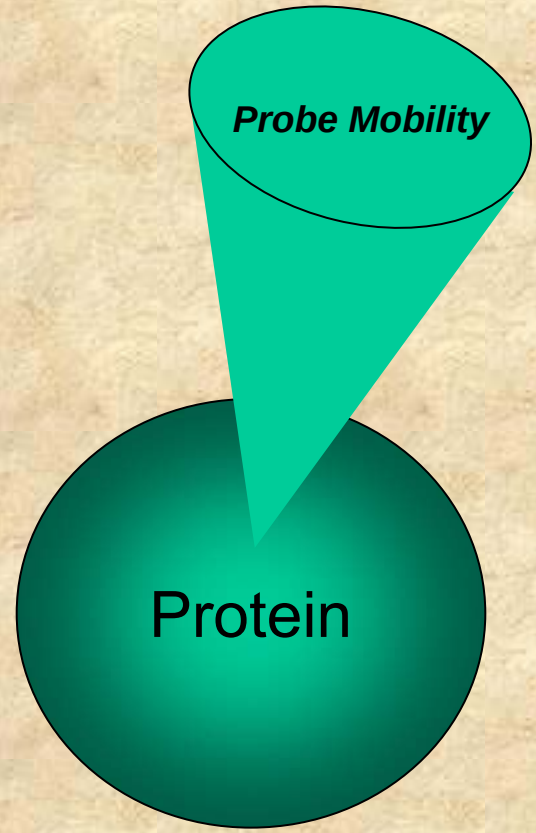
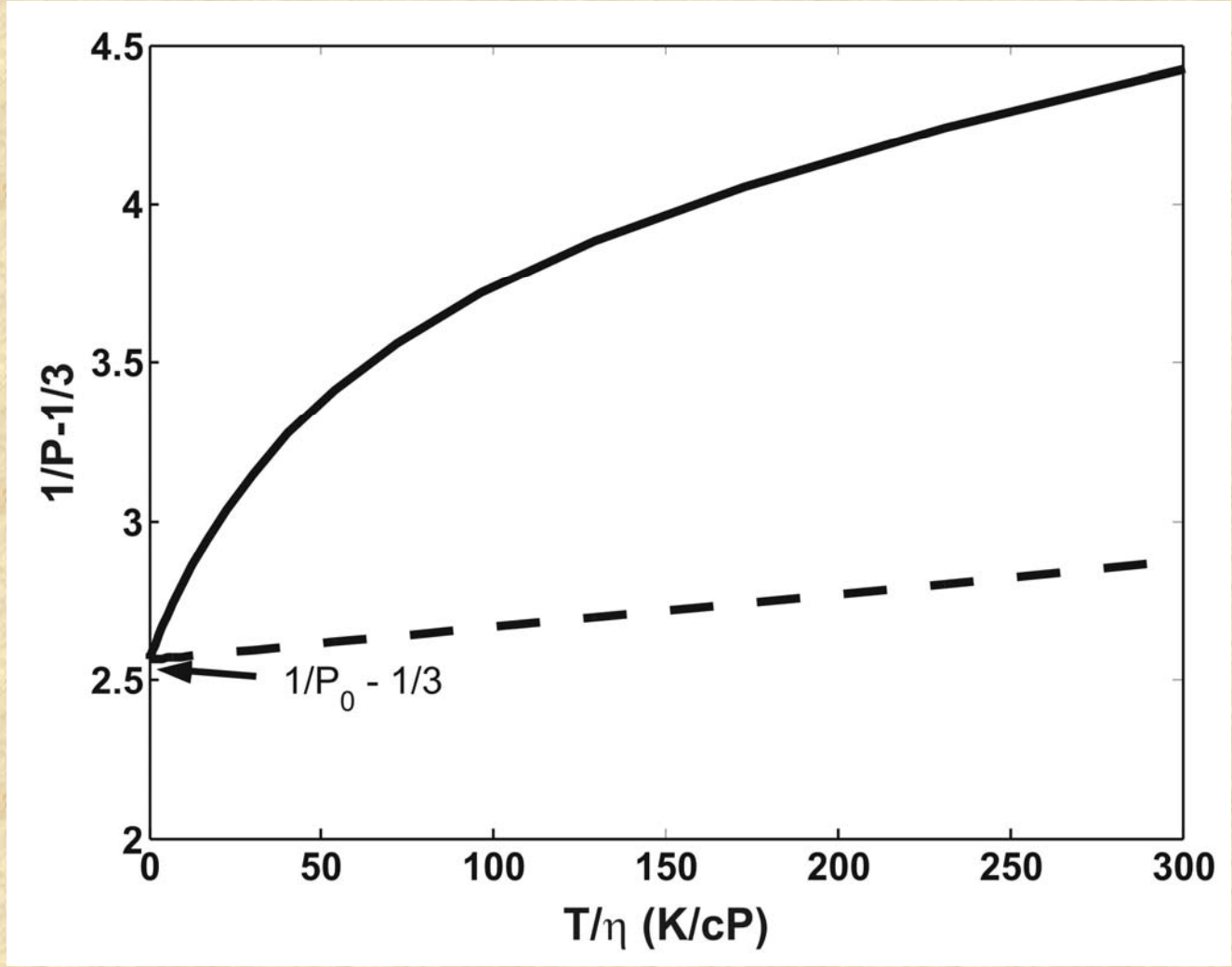
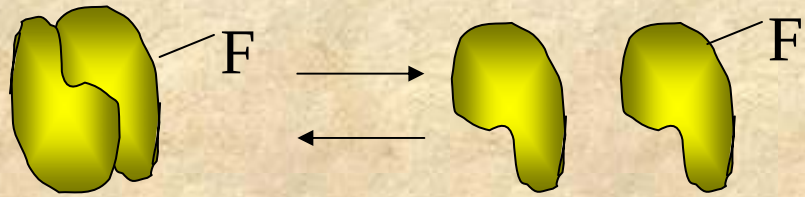


Perrin-Weber Plots can be used to assess the presence of local probe mobility

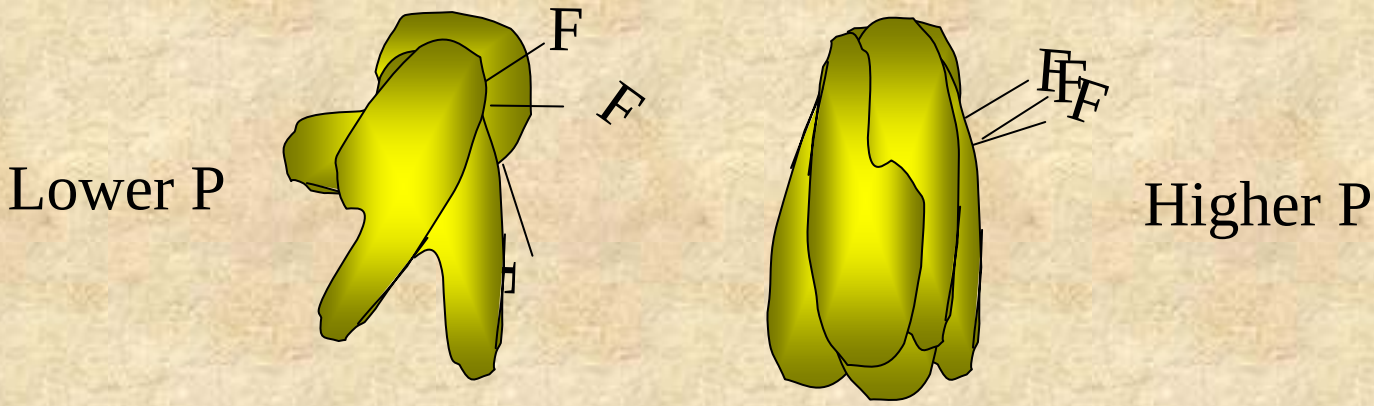


Perrin-Weber plot of a labeled protein (25kDa) with no local motion of the probe (4ns) (dashed line) and where 40% of the depolarization is due to local probe motion (solid line). The P_0 value can be determined from the y-axis intercepts.

Polarization methods are ideally suited to study the aggregation state of a protein. Consider, for example the case of a protein dimer - monomer equilibrium.



Following either intrinsic protein fluorescence (if possible) or by labeling the protein with a suitable probe one would expect the polarization of the system to decrease upon dissociation of the dimer into monomers since the smaller monomers will rotate more rapidly than the dimers (during the excited state lifetime).



Hence for a given probe lifetime the polarization (or anisotropy) of the monomer will be less than that of the dimer

In the concentration range near the dimer/monomer equilibrium constant, one expects to observe a polarization intermediate between that associated with either dimer or monomer. One can relate the observed polarization to the fraction of dimer or monomer using the additivity of polarizations first described by Weber (1952) namely:

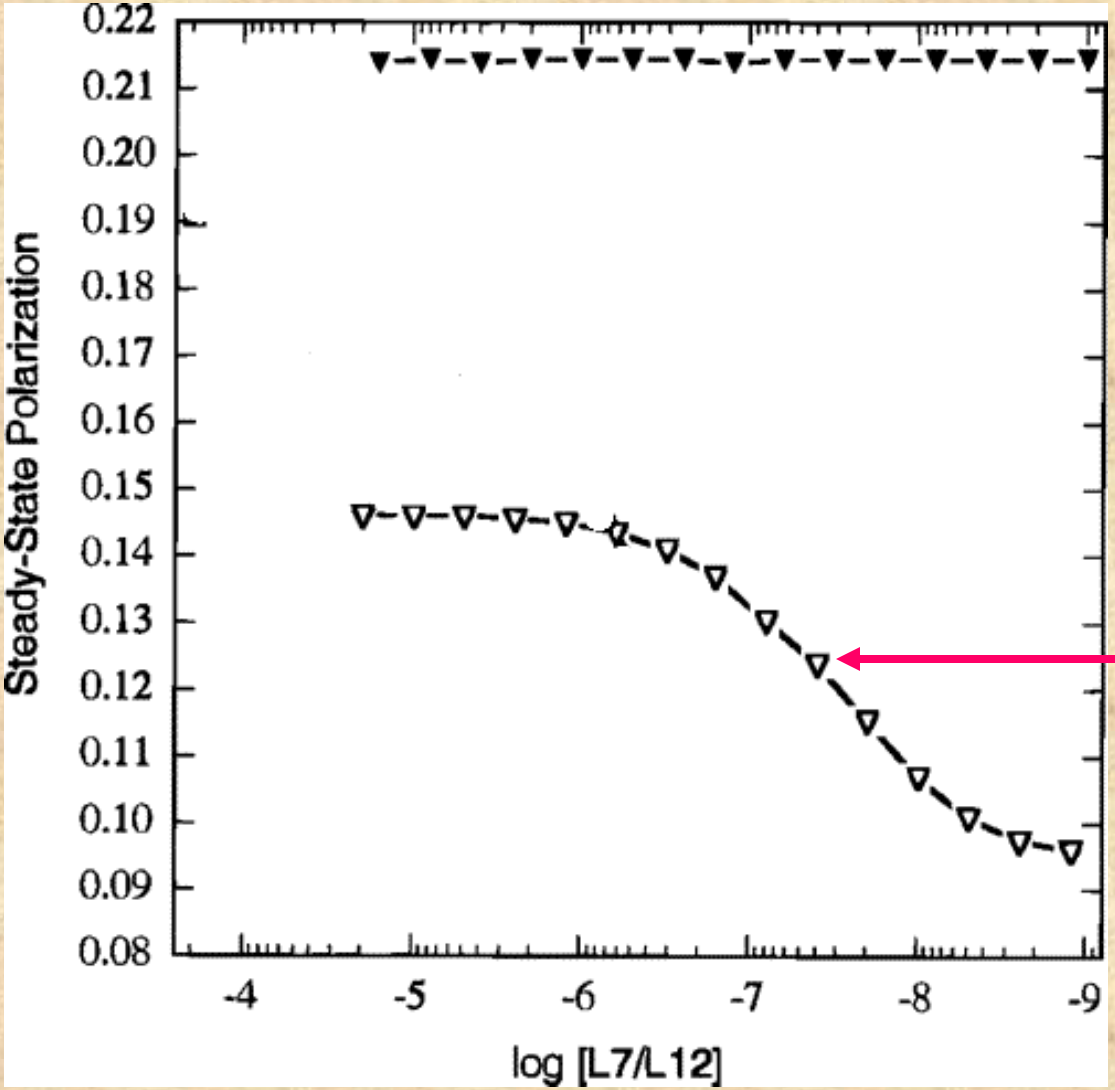
$$\left(\frac{1}{<P>}-\frac{1}{3}\right)^{-1}=\sum f_i\left(\frac{1}{P_i}-\frac{1}{3}\right)^{-1}$$

where <P> is the observed polarization, f_i is the fractional intensity contributed by the ith component and P_i is the polarization of the ith component. One must then relate the fractional intensity contributions to molar quantities which means that one must take into account any change in the quantum yield of the fluorophore associated with either species.

The anisotropy function is directly additive (owing to the fact that the denominator represents the total emitted intensity) and hence:

$$<r>=\sum f_i r_i$$

So to determine the dissociation constant, one can dilute the protein and observe the polarization (or anisotropy) as a function of protein concentration as shown below.



FITC-lysozyme (monomer)

L7/L12 (dimer)

The polarization/anisotropy approach is also very useful to study protein-ligand interactions in general.

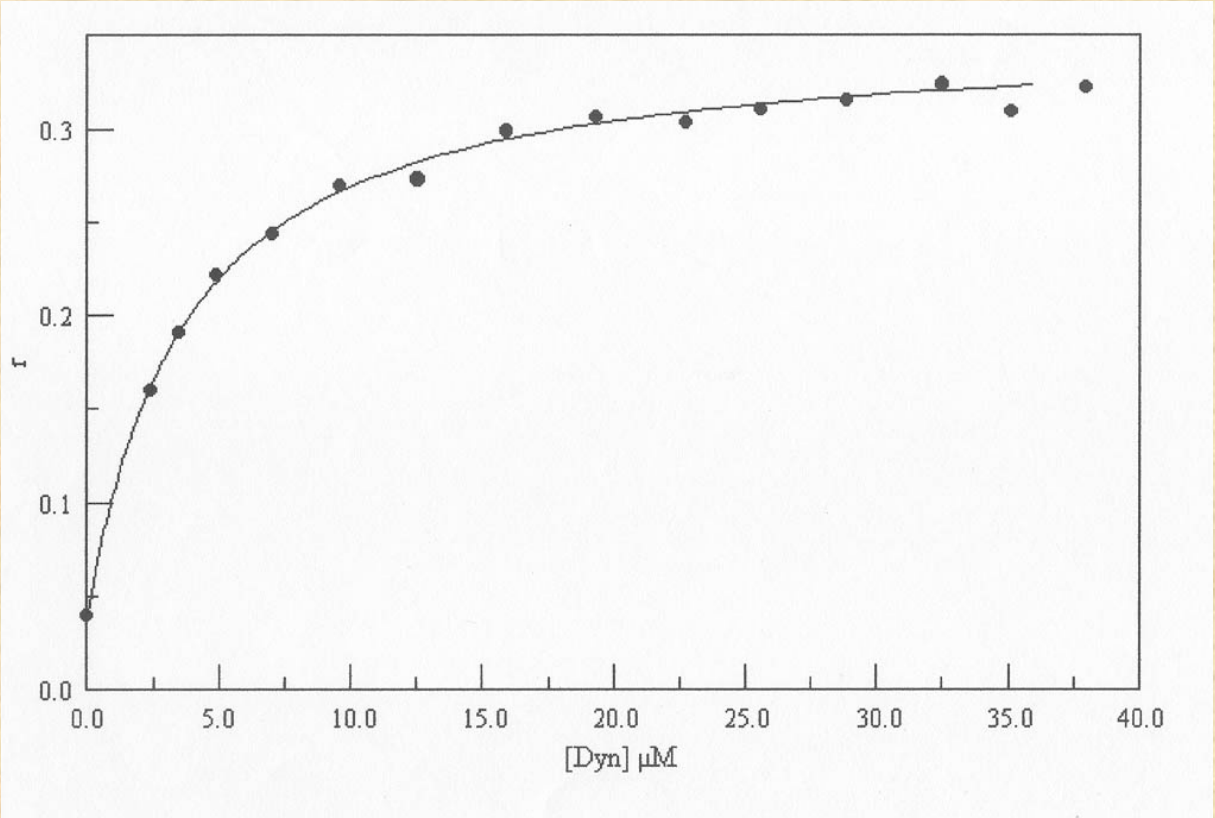
The first application of fluorescence polarization to monitor the binding of small molecules to proteins was carried out by D. Laurence in 1952 using Gregorio Weber's instrumentation in Cambridge. Specifically, Laurence studied the binding of numerous dyes, including fluorescein, eosin, acridine and others, to bovine serum albumin, and used the polarization data to estimate the binding constants.

Although many probes (such as fluorescein) do not significantly alter their quantum yield upon interaction with proteins, one should not take this fact for granted and would be well advised to check. If the quantum yield does in fact change, one can readily correct the fitting equation to take the yield change into account. In terms of anisotropy the correct expression relating observed anisotropy (r) to fraction of bound ligand (x), bound anisotropy (r_b), free anisotropy (r_f), and the quantum yield enhancement factor (g) is:

$$x = \frac{r - r_f}{r_b - r_f + (g-1)(r_b - r)}$$

$$x = \frac{(3 - P_b)(P - P_f)}{(3 - P)(P_b - P_f) + (g-1)(3 - P_f)(P_b - P)}$$

A typical plot of polarization versus ligand/protein ratio is shown below:

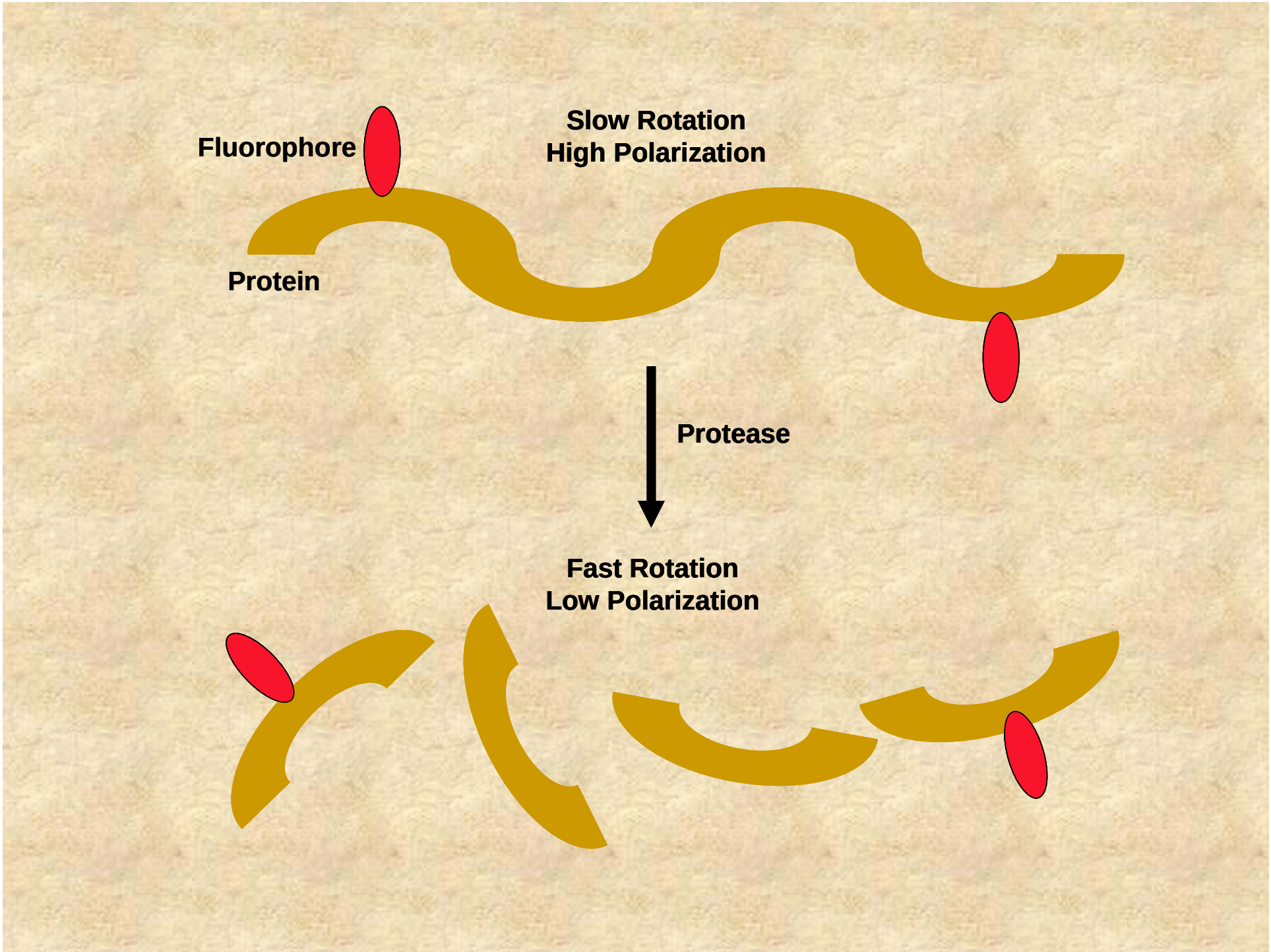


In this experiment, 1 micromolar mant-GTPγS (a fluorescent, non-hydrolyzable GTP analog) was present and the concentration of the GTP-binding protein, dynamin, was varied by starting at high concentrations followed by dilution. The binding curve was fit to the anisotropy equation (in this case the yield of the fluorophore increased about 2 fold upon binding). A K_d of 8.3 micromolar was found

Proteolytic processing, mediated by proteolytic enzymes, or proteases, is critical to many vital biological processes, including post-translational protein processing, blood clotting, digestion, hormone processing, apoptosis, and many others, as well as many deleterious processes, such as those mediated by anthrax and botulinum neurotoxins.

Hence, an evaluation of protease activity is often a requirement for an understanding of a particular pathway or for development of novel therapeutic agents. Protease assays have been around for many decades but more recently the development of rapid and sensitive protease assays suitable for high-throughput screening has attracted considerable attention.

Fluorescence polarization lends itself very well to such assays since the essential aspect of a protease is to cleave a peptide bond which almost always results in smaller molecular weight species. Hence, if the target protein can be labeled with a fluorescence probe one would expect the polarization to decrease after proteolysis since the fluorophore will be able to rotate more rapidly after the protein mass to which it is tethered is reduced in size


Fluorophore
Slow Rotation
High Polarization
Protein
Protease
Fast Rotation
Low Polarization

The first description of a protease assay based on fluorescence polarization was by Richard Spencer and collaborators

Design, Construction, and Two Applications for an Automated Flow-Cell Polarization Fluorometer with Digital Read Out: Enzyme-Inhibitor (Antitrypsin) Assay and Antigen–Antibody (Insulin–Insulin Antiserum) Assay

Richard D. Spencer, Fernando B. Toledo, Ben T. Williams, and Norma L. Yoss

CLIN. CHEM. 19/8, 838–844 (1973)

BODIPY–α-Casein, a pH-Independent Protein Substrate for Protease Assays Using Fluorescence Polarization[1]

Sylvia Z. Schade,[2] Michael E. Jolley,* Brian J. Sarauer, and Lloyd G. Simonson

Naval Dental Research Institute, 2701 Sheridan Road, Great Lakes, Illinois 60088-5259; and
**Jolley Consulting and Research, Inc., 683 East Center Street, Unit H, Grayslake, Illinois 60030*

ANALYTICAL BIOCHEMISTRY **243,** 1–7 (1996)

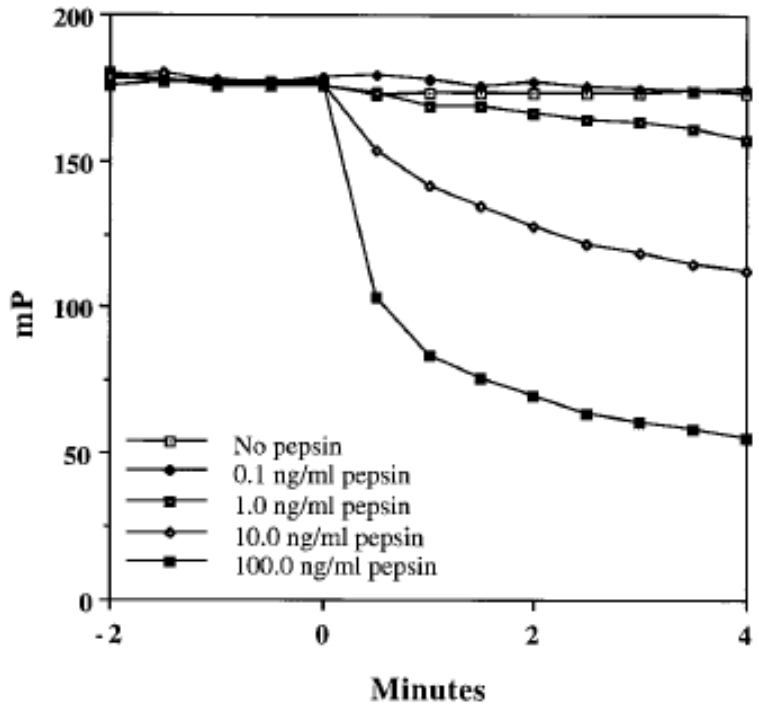


FIG. 1. Protease activity of pepsin at pH 2. Chromatographically purified, crystallized pepsin was assayed at 37°C in 0.01 N HCl using 0.5 μg/ml (25 pmol/ml) BODIPY–α-casein conjugate as a substrate. Proteolytic activity was followed by fluorescence polarization measurements (mP) automatically recorded. One nanogram of pepsin equals 4 mU of enzyme.

FPIA – Fluorescence Polarization ImmunoAssay

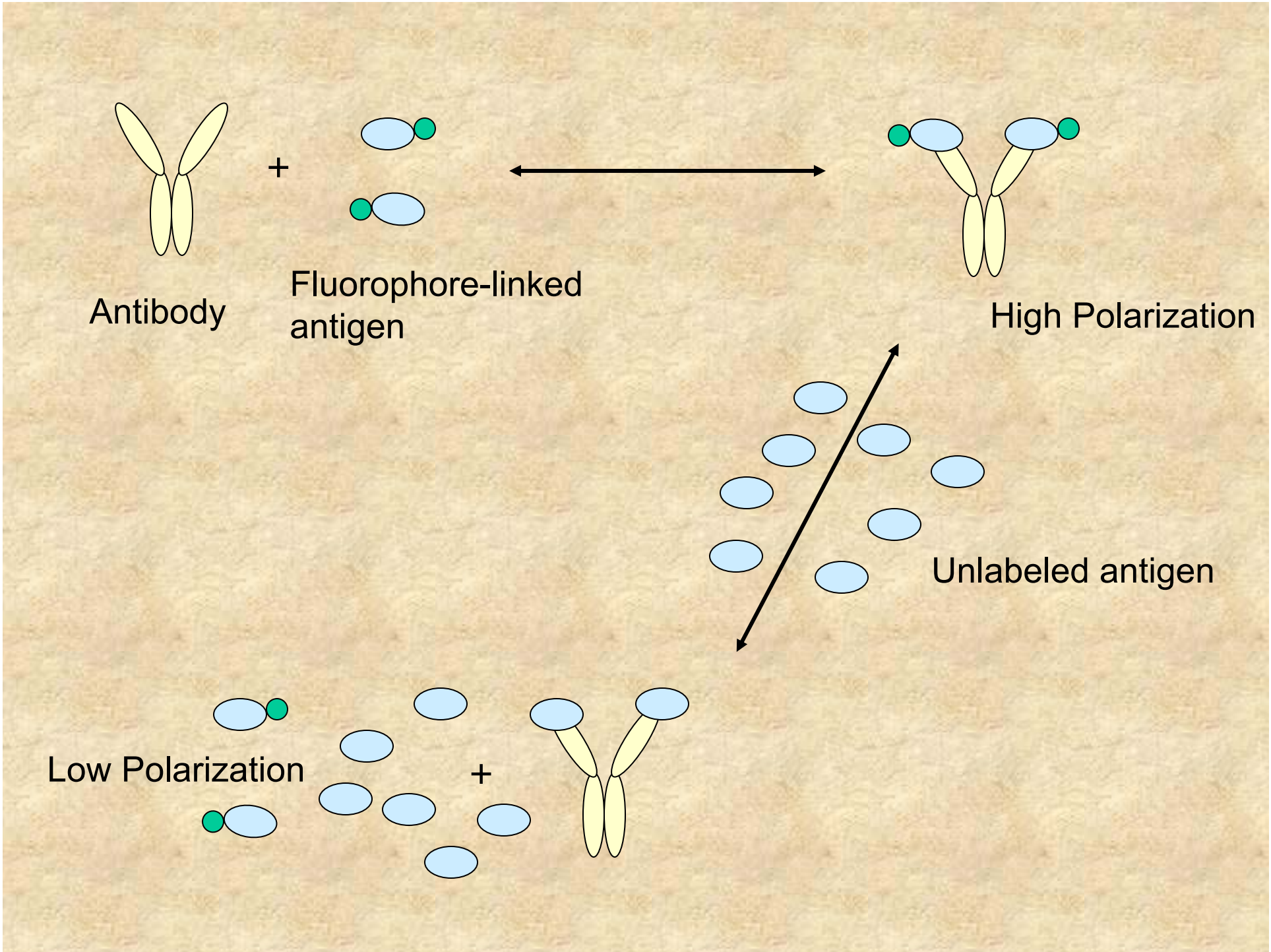
Among the first commercial instruments designed to use a fluorescence polarization immunoassay for clinical diagnostic purposes was the Abbott TDx – introduced in 1981.

The basic principle of a polarization immunoassay is to:

(1) Add a fluorescent analog of a target molecule – e.g., a drug – to a solution containing antibody to the target molecule

(2) Measure the fluorescence polarization, which corresponds to the fluorophore bound to the antibody

(3) Add the appropriate biological fluid, e.g., blood, urine, etc., and measure the decrease in polarization as the target molecules in the sample fluid bind to the antibodies, displacing the fluoroescent analogs.


Antibody
+
Fluorophore-linked antigen
High Polarization
Unlabeled antigen
Low Polarization
+

7. Dandliker, W. B., Kelly, R. J., Dandliker, J., et al., Fluorescence polarization immunoassay. Theory and experimental method. *Immunochemistry* **10**, 219–227 (1973).

CLIN. CHEM. 27/7, 1190–1197 (1981)

Fluorescence Polarization Immunoassay I. Monitoring Aminoglycoside Antibiotics in Serum and Plasma

Michael E. Jolley, Stephen D. Stroupe, Chao-Huei J. Wang, Helen N. Panas, Candace L. Keegan, Robert L. Schmidt, and Kathryn S. Schwenzer

Special Apparatus

A microprocessor-controlled fluorometer was constructed in our laboratories (*19*). The instrument was designed to measure precisely the polarization of fluorescence emitted from the sample contained in a standard 12 × 75 mm disposable culture tube. Each determination was performed in 10 s, to a precision of ±0.001 polarization unit. The instrument determined fluorescence polarization (in arbitrary units) according to the equation (*7*):

$$P = \frac{I_{parallel} - I_{perpendicular}}{I_{parallel} + I_{perpendicular}}$$

where I represents light intensity. For convenience, results are reported in millipolarization units (mP), where 1 mP = 0.001 P.